Abc Taxa

A Guide to the Parasites of African Freshwater Fishes

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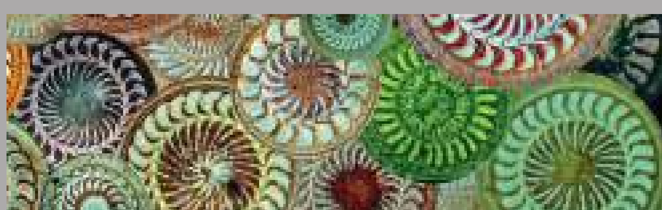

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Cover illustration: collage of trichodinid ciliates. Photograph by L. Basson.

Inner page photograph: *Ergasilus* sp. attached to the gill filament of the blackspotted squeaker, *Synodontis nigromaculatus*, from the Okavango Delta, Botswana. Photograph by J. Van As.

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A Guide to the Parasites of African Freshwater Fishes

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Abstract

The rivers and lakes of Africa contain almost 25% of the world's 13,000 freshwater fish species and are second only to South America in species richness. These fish are parasitised by a wide range of organisms that can be detrimental to both farmed and wild fishes with consequent effects on economic development, and often on human health. Knowledge of these parasites in African freshwater fishes is limited and this book is intended to promote and advance understanding of African fish parasites by providing information on the best techniques for investigating fish and their parasites and keys to parasite identification. The first comprehensive list of all known freshwater fish parasites in Africa is presented here, with information on their known hosts and distribution, keys to all genera and representative illustrations for every genus. This information should facilitate and stimulate the development of fish parasitology on the African continent which has great potential for aquaculture and fishery development.

Dedication

This book is dedicated to the memory of Jo Van As for his extraordinary contribution to fish parasitology in Africa.

Keywords

ichthyoparasitology, fish disease, protists, helminths, parasitic crustaceans

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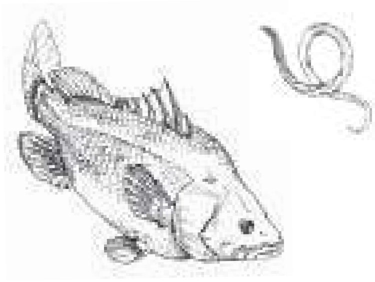
INTRODUCTION



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Freshwater systems in Africa are dominated by 12 large river systems that contribute to the bulk of the surface water including the Nile River, the world's longest river (6,700 km), and the Congo River that transports the second largest annual volume of water of any river in the world. Additionally, on a global scale, Lake Victoria is the third largest natural freshwater lake, Lake Tanganyika the second oldest and deepest and the Okavango Delta is the largest inland delta. Collectively, these freshwater bodies contain almost 25% of the world's 13,000 freshwater fish species, second only in species richness to South America (Lévêque *et al.* 2008; Snoeks *et al*. 2011).

The rich freshwater fish fauna, a high percentage of endemism at the species (almost 100%) and the family (over 40%) level (Lundberg *et al.* 2000), several well-known cases of adaptive radiation and several fish species that are the basis of worldwide aquaculture (*e.g.*, the different species of 'tilapia' and clariid catfishes) contribute to Africa's potential to serve as an important model for ecological and evolutionary studies on fish parasites and their interactions. However, fish parasites are still poorly known, especially when compared with other continents, in particular Europe and North America, which makes it impossible to reliably assess their diversity, interrelations, distribution and potential effects on their fish hosts, whether they are native or invasive species.

Only a small percentage of known African fishes have been examined for parasites and the present knowledge of the parasite fauna of African fishes is fragmentary and represents only the tip of the iceberg. Similarly, data from a majority of African countries are scarce or completely lacking (Khalil & Polling 1997). Therefore, future research should focus on poorly studied fish hosts as well as the regions from where limited or no information is available. Attention should also be paid to potential pathogens of fishes in aquaculture, and the diversity and distribution of invasive parasites. Studies on the life cycles of African fish parasites are almost completely lacking. Since much valuable material of the parasites from Africa is unusable or has been lost due to usage of incorrect methods, the application of adequate methods for collecting, processing and identifying fish parasites is crucial.

Consequently, this book – *A Guide to the Parasites of African Freshwater Fishes* – aims at filling a considerable gap in the present knowledge of an important group of eukaryotic organisms that may have detrimental effect on cultured and wild fishes, but also may negatively influence human health in the case of fish-borne parasites. The focus of the book is on methods necessary for adequately processing fish and their parasites including tools that may help in parasite identification and studies on their life cycles, ecology and pathology. The only comprehensive sources of data on fish parasites in Africa are the checklists of Khalil (1971) and Khalil and Polling (1997), the latter reporting 568 species of helminth parasites of African freshwater fishes, Paperna's (1979) book on the Monogenea of freshwater fishes and his FAO guide to fish parasites (Paperna 1996). However, these books and checklists obviously need an update and they do not provide methodological information. Moreover, recent developments in parasitology, especially molecular tools and biostatistics, applied in identification, elucidating life cycles and phylogenetic and ecological studies, warrant a new comprehensive text on African fish parasites.

During the previous two decades, considerable progress has been made in unravelling the diversity of fish parasites in Africa, their host associations and distribution, to a large extent thanks to the collaborative effort of several research institutions in Europe and Africa, which are well renowned globally and which are able to apply a multidisciplinary approach in research on a wide spectrum of parasite groups (see Fig. below). In addition to the theoretical importance of data on fish parasites, adequate knowledge of causative agents of fish diseases is crucial for decreasing economic losses they may cause, especially in aquaculture, which is rapidly developing in many African countries. In view of currently widely accepted integrative approaches to human, animal and ecosystem health, capacity development in monitoring and identification of pathogens and vectors in the Global South is crucial (Keune *et al.* 2017).

Fig. Research on fish parasites throughout Africa. **A.** Fish collection in Lake Turkana, Kenya; **B.** Fish examination in the Sudan; **C.** Teaching course on fish parasitology at the University of Khartoum, Sudan; **D.** Field laboratory in the Sudan. (Photographs by R. Blažek, A. de Chambrier and T. Scholz)

It is thus timely to document this progress to the professional public in Africa in a manner that will stimulate and facilitate the development of modern fish parasitology in this continent, which has a great, but yet only partly exploited potential for aquaculture and fisheries. The present book provides basic information about methods used to study unicellular and metazoan parasites of freshwater fishes and an updated list of these parasites found in Africa, together with their hosts, keys to all genera and representative illustrations of members of every genus.

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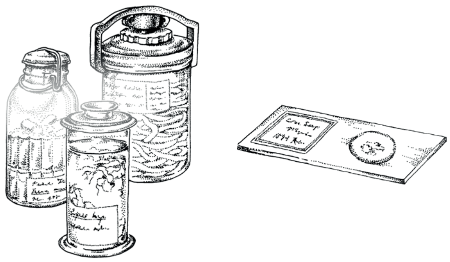
PART 1

HISTORY OF FISH PARASITOLOGY IN AFRICA



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Introduction

Freshwater systems in Africa cover a surface area of almost 30,000 km^2 and include 12 major freshwater habitat types ranging from closed basins, small lakes, floodplains and swamps to large tropical rivers (Van As *et al*. 2012). More than 3,200 species of fish species belonging to 76 of the world's 170 freshwater fish families are known from Africa, with representatives of the families Cichlidae and Cyprinidae dominating African ichthyofauna (Lévêque *et al.* 2008). However, they are irregularly distributed over the continent and include highly diverse systems such as Lake Malawi (800 species) and the Congo River (700 species), as well as low diversity areas further north and south of the tropics.

Diversity of fish parasites in Africa and a brief history of their exploration

In contrast to the known high diversity of the freshwater fish hosts, the relatively low number of identified fish parasites of Africa, with the exception of the species-rich monogenean genus *Cichlidogyrus* Paperna, 1960 (see Pariselle & Euzet 2009), shows a clear paucity of research on this group of parasites. It is even more obvious when the numbers of the parasites reported from African fishes are compared with those in other continents, especially Europe (see, *e.g.*, Scholz *et al.* 2016 for data on fish trematodes). Our knowledge of the diversity of freshwater fish parasites (and the same is valid for parasites of marine bony fishes – Smit & Hadfield 2015) was kick-started in the second half of the 19th century (see Khalil 1971).

In the following paragraphs, the history of studies on the parasites of teleost fishes in Africa is briefly surveyed. However, this account does not represent an exhaustive search of the history of fish parasite exploration in the African continent, partly due to the fact that many papers of African fish parasitologists were published in hardly accessible, regional or local journals, and that nowadays, research from the Global South is unfortunately all too often published in so-called predatory journals. Exhaustive data (updated until 2003) on the parasites of freshwater fishes in Africa can be found in the Host-Parasite Database generated at the Natural History Museum in London (Gibson *et al.* 2005).

Leydig (1853) described the bothriocephalidean cestode *Tetrabothrium polypteri* (now *Polyonchobothrium polypteri*) from bichir, and Wedl (1861) described another bothriocephalidean *Tetracampos ciliotheca* from *Clarias* catfish. Fritsch (1886) described two proteocephalidean cestodes, *Corallobothrium fimbriatum* and *Taenia malapteruri*, from electric catfish *Malapterurus electricus* (Gmelin). At the turn of the 20th century, A. Looss published a series of papers from 1896 to 1907 on the parasitic fauna of Egypt (see Khalil 1971), including the first two species of trematodes recorded from Africa, namely *Acanthostomum spiniceps* (Looss, 1896) and *Haplorchoides cahirinus* (Looss, 1896) found in the bagrid catfishes *Bagrus bajad* (Forsskål) and *Bagrus docmak* (Forsskål). Looss' work on the Nile River in Egypt was continued by T. Odhner (1902-1911), who also dealt mainly with trematodes and described eight new species from the Sudan, based on material collected as part of the Swedish zoological expedition to the Nile River (see Canaris & Gardner 2003).

Khalil (1971) published the first checklist of the helminth parasites of freshwater fishes in Africa. He reported 223 species of adult helminth parasites from African freshwater fishes (86 species of monogeneans, 44 trematodes, 41 cestodes, 41 nematodes and 11 acanthocephalans). Twenty-six years later, an updated checklist (Khalil & Polling 1997) recorded 568 adult helminth parasites, *i.e.*, an increase of 155%, including 342 species of monogeneans (4 times more than in 1971!), 62 trematodes (increase of 41%), 61 cestodes (49%), 80 nematodes (95%) and 21 acanthocephalans (91%). In addition, numerous larval forms have been reported (Khalil & Polling 1997).

However, the species discovery rate over the following years dropped drastically. For example, only five species of trematodes, including one new genus *Malawitrema* Bray et Hendrix, 2007, have since been added to the known fauna of adult trematodes (Scholz *et al.* 2016). In total, 67 species of adult trematodes of 34 genera from 20 families and 35 species of metacercariae of 20 genera from eight families from African freshwater fish are now known (Scholz *et al.* 2016).

Another group of parasites of African freshwater fishes that has received relatively high attention is the parasitic crustaceans, especially the Copepoda. The first record of African freshwater parasitic copepods is that by Cunnington (1914) describing *Lernaea dicerocephala* Cunnington, 1914 and *L. haplocephala* Cunnington, 1914 collected during an extensive expedition to Lake Tanganyika during 1904 to 1905. Since that first description, approximately 45 more species of the family Lernaeidae have been described from freshwater fishes in various localities in Africa, making it the species-richest of all the fish parasitic Crustacea reported from Africa (Oldewage & Avenant-Oldewage 1993).

The largest contribution to our knowledge of freshwater fish parasitic Crustacea was by the British zoologist, Geoffrey Fryer, who described more than 20 branchiuran and copepod species over a twenty-two year period (1955 to 1977). This includes seven species of the African endemic branchiuran genus *Chonopeltis* Thiele, 1900. Fryer (1968) also summarised the known distribution of the parasitic crustaceans of African freshwater fishes, which included just over 80 species at that time, and provided information on the taxonomy, biology and evolution of certain species. Although Fryer's (1968) work is almost 50 years old and in need of an update, it is still considered as one of the most valuable contributions to our understanding of parasitic crustaceans of African freshwater fishes.

Data on protists and myxozoans of African fishes have not been summarised in a form similar to that of the trematodes and crustaceans and thus it is difficult to provide total numbers for these parasites. Several ectoparasitic protists appear to have a cosmopolitan distribution and have also been reported from various places in Africa. Van As and Basson (1984) and Paperna (1996) reported the flagellate *Ichthyobodo necator* (Henneguy, 1883), the pathogenic ciliophorans *Chilodonella hexasticha* (Kiernik, 1909) and *C. piscicola* (Zacharias, 1894), as well

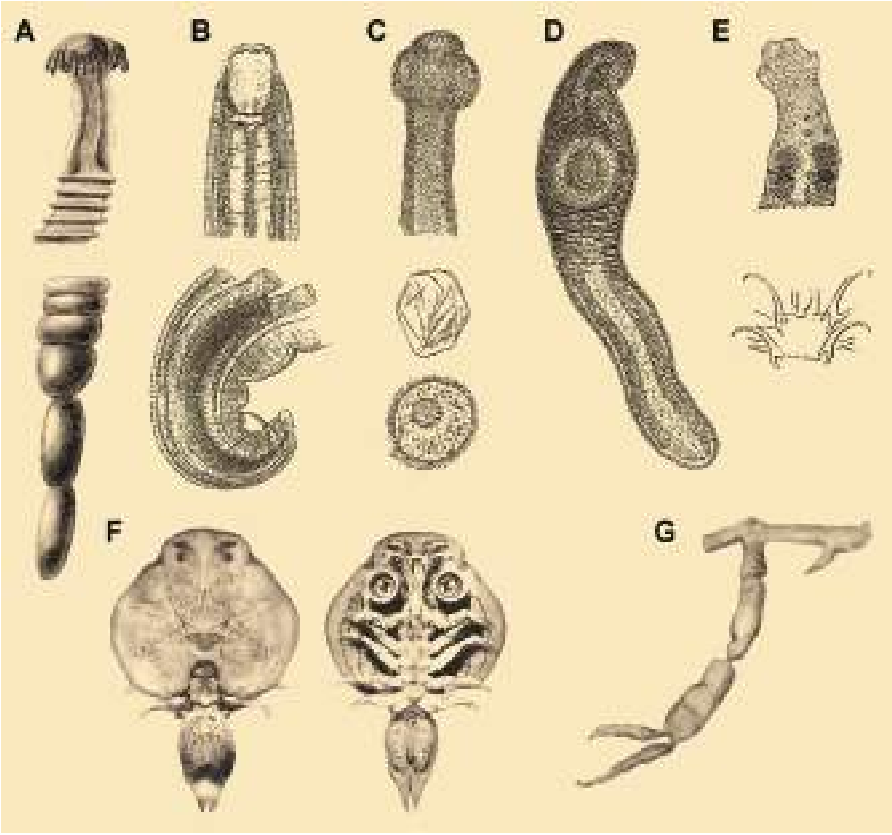

Fig. 1. First documentations of freshwater fish parasites in Africa. **A.** Cestoda – *Tetrabothrium polypteri* Leydig, 1853 (now *Polyonchobothrium polypteri*) from *Polypterus bichir*; **B.** Nematoda – *Cucullanus laeviconchus* Wedl, 1861 (now *Procamallanus laeviconchus*) from *Synodontis schall*; **C.** Cestoda – *Tetracampos ciliotheca* Wedl, 1861 from *Clarias anguillaris*; **D.** Trematoda – *Distoma bagri incapsulatum* Wedl, 1861 (now *Nephrocephalus bagriincapsulatus*) from *Bagrus* sp.; **E.** Monogenea – *Dactylogyrus gracilis* Wedl, 1861 (now *Annulotrema gracilis*) from *Hydrocynus forskahlii*; **F.** Branchiura – *Argulus incisus* Cunnington, 1913 from *Auchenoglanis occidentalis*; **G.** Copepoda – *Lernaeocera diceracephala* Cunnington, 1914 (now *Lernaea diceracephala*) from *Clarias gariepinus*. (Modified from Leydig 1853; Wedl 1861; Cunnington 1913, 1914.)

as the equally notorious pathogenic ciliophoran *Ichthyophthirius multifiliis* Fouquet, 1876 from southern Africa. Paperna (1972) also reported *I. multifiliis* from Uganda. The fish blood trypanosomes also seem to have a wide African distribution, but only three species are reported from a wide range of African freshwater fishes from Egypt, West Africa, the Congo, Sudan, Uganda, Botswana, Mozambique and South Africa (see Smit *et al.* 2004).

Protist groups that have been studied in more detail are the two ciliophoran orders of the subclass Peritrichia, *i.e*., the Sessilida and Mobilida. In the case of the former, there were vague reports of sessilines from fishes in Africa, but more in depth work was done in South Africa in the 1980s by Viljoen and Van As (1983, 1985). These authors collectively described a total of 14 sessiline species (of which 12 were described as new species) representing four genera, *i.e*., *Epistylis* Ehrenberg, 1830, *Apiosoma* Blanchard, 1885, *Scopulata* Viljoen and Van As, 1985 and *Ambiphrya* Raabe, 1952.

The Mobilida have received far more attention, specifically representatives of the family Trichodinidae. The first mention of a fish trichodinid species in Africa was made by Fantham (1918) from a marine host, while the first valid species description was published by Basson *et al.* (1983). Since then, 50 trichodinid species, representing five genera have been described, the vast majority from freshwater fishes and most of these in southern Africa. The southern African trichodinid (from South Africa, Namibia and Botswana) fauna stands at 35 freshwater species due to the contributions of authors such as Basson *et al.* (1983), Basson and Van As (1987, 1989, 1991, 1993, 2002), Basson (1989), and Van As and Basson (1989, 1992), whereas six marine fish trichodinids have been described from South Africa (Basson *et al.* 1990; Van As and Basson 1996) and Namibia (Aljoshkina & Stein 1984). In the rest of Africa, a total of 16 trichodinid species have been found (of which nine were not encountered in southern Africa): three species from marine fish in Senegal (Loubser *et al.* 1995), five species from freshwater fishes in Egypt (El-Tantawy & Kazubski 1986; Kazubski & El-Tantawy 1986; Abdel-Meguid 1995), five species from brackish water in Benin (Maslin-Leny 1988), two species from Kenya (Kazubski 1986; Kazubski & El-Tantawy 1986) and a single species from a freshwater fish in Nigeria (Obiekezie & Ekanem 1995). Probably the least studied fish parasitic protists from Africa are blood protozoans. Currently only a single species, the dactylosomatid, *Babesiosoma mariae* (Hoare, 1930), has been described from African freshwater fishes (Smit *et al.* 2003).

One of the highest numbers of helminths has been reported from the sharp tooth catfish, *Clarias gariepinus* (Burchell), an economically important species occurring in almost all the major river systems. This fish harbours as many as 12 species of adult trematodes, five species of metacercariae, 20 species of monogeneans and at least four species of adult cestodes (Mashego & Saayman 1989; Gibson *et al.* 2005 and references therein; Barson & Avenant-Oldewage 2006; Přikrylová *et al.* 2012; Beletew *et al.* 2016; Scholz *et al.* 2016). Even more impressive, the Nile tilapia *Oreochromis niloticus* (Linnaeus) is infected by 23 trematodes and 20 monogeneans, two cestodes, two acanthocephalans, seven nematodes and it is the African cichlid with the highest number of formally reported helminth species (data from Vanhove *et al.* 2016 and references therein).

The relatively low number of parasites of freshwater fishes in Africa almost certainly does not reflect a naturally low diversity, but rather is due to the lack of dedicated biodiversity studies (Smit & Hatfield 2015; Van As 2015; Scholz *et al.* 2016). The distribution of the currently known fish parasites over the continent is another indication of uneven research. In the case of trematodes, there are no records from almost 40% of African countries. The highest numbers of fish trematode species were reported from the countries where foreign fish parasitologists were active, such as Egypt and the Sudan (Barson & Avenant-Oldewage 2006; Smit & Hadfield 2015).

In 2003, the Sixth International Symposium on Fish Parasites was organised for the first time on the African continent, in Bloemfontein, South Africa (the principal organiser was Jo Van As). This conference was an important stimulus for the development of ichthyoparasitology in the African continent. In addition to stimulation of a new generation of African fish parasitologists, this meeting accelerated intensive international cooperation, including research and teaching visits by foreign experts to Africa and collecting trips with participation of local fish parasitologists. As a result, the number and quality of scientific outcomes dealing with fish parasites in Africa have increased considerably. This latter is clearly demonstrated in the recent review by Jo Van As on the history of freshwater fish parasitology in southern Africa (Van As 2015). In that review, the contribution by South African fish parasitologists from seven different academic institutions was summarised and it was concluded that despite the fact that the country has arguably the largest community of aquatic parasitologists in Africa, the investigation and mapping of fish parasite biodiversity is still in its infancy, because all rivers and their fish parasite fauna of southern Africa have not been explored. Southern African fish parasitology is also visible on the international scene through other ways, *e.g.*, Maxwell Barson from Zimbabwe is a member of the Aquatic Animals Health Standards Commission of the World Organisation for Animal Health (OIE).

Shortly afterwards, in 2009, the Sixth International Symposium on Monogenea was also held in South Africa, namely in Cape Town (principal organisers were Louis Du Preez and Kevin Christison). Monogeneans take up a special place in African fish parasitology, in view of their species richness (see above), but also in view of the large contribution of African researchers to our knowledge of these parasitic flatworms. The first African freshwater monogenean reported from fishes, *Annulotrema gracilis*, however, was described by the Austrian pathologist Wedl (1861) (as *Dactylogyrus gracilis*) from *Hydrocynus forskahlii* (Cuvier) in Egypt. Afterwards, things went rather silent regarding African monogeneans, with the notable exception of some new species from non-fish hosts. *Oculotrema hippopotami* was proposed by the American Stunkard (1924), based on museum specimens probably retrieved from hippopotamus in an Egyptian zoo, and Vercammen-Grandjean (1960) described *Gyrdicotylus gallieni* from the frog *Xenopus victorianus* in the Congolese South Kivu province. Change came in the second half of the 20th century, following the discovery of some typical African monogenean genera infecting fishes. For example, *Macrogyrodactylus polypteri* was described by Malmberg (1957) from *Polypterus senegalus* Cuvier in Gambia (paper dated 1956 but published in 1957) and *M. congolensis* (Prudhoe, 1957) (described as *Neogyrodactylus congolensis* as the author was unaware of Malmberg's aforementioned paper) reported by Prudhoe (1957) from *Clarias lazera* Valenciennes (now *C. gariepinus*) from the Democratic Republic of the Congo, also in 1957. Afterwards, Ilan Paperna became well-known for his substantial contribution to our understanding of African monogeneans (Paperna 1960). He described numerous genera and species, among which the genus *Cichlidogyrus* Paperna, 1960, currently has the most nominal species known in Africa.

In subsequent decades, research efforts on, and knowledge of, monogeneans and other fish parasites proliferated, mainly throughout French-speaking Africa, largely originating from the 'school' of Louis Euzet and his first PhD student Claude Combes. Subsequent generations of French parasitologists active in Africa (not only dealing with monogeneans) include(d) Emile Birgi, Georges Bouix, Laurence Douëllou, Jacques Dupouy, Claude Gabrion, Alain Lambert, Claude Maillard, Guy Oliver, André Raibaut, Jean-Paul Trilles and, more recently, Jean-Lou Justine and Antoine Pariselle. Especially the latter spent much of the last decades based in Africa training a next generation of monogenean workers. Some African (senior) researchers that can be mentioned, at the risk of forgetting people, are Ouafae Berrada-Rkhami, Fouzia El Hafidi and Salwa El Gharbi (Morocco), Faiza Amine and Fadila Tazerouti (Algeria), Christian Dossou (Benin), Charles Félix Bilong Bilong and Jacques Nack (Cameroon), Mohammed El-Naggar (Egypt), Valentin N'Douba and K.G. Blahoua (Ivory Coast), Sylvère Rakotofiringa and Jeanne Rasamy (Madagascar), Austin Ikechukwu Obiekezie and M. Taege (Nigeria), Arfang Diamanka (Senegal), Lotfi F. Khalil (Sudan), Mohamed Hedi Ktari, Lassad Neifar and Lobna Boudaya (Tunisia), Annie Chishawa (Zimbabwe) (many of whom have benefited from the training and advice of L. Euzet), and, of course, the South African teams referred to above. Also, an even younger generation of young African parasitologists has started to publish on monogeneans over the last five years, *e.g.*, Chahrazed Rahmouni and Zouhour El Mouna Ayadi (Algeria), Dieu ne dort Bahanak and Etienne Didier Bassock Bayiha (Cameroon), Fidel Muterezi Bukinga and Gyrhaiss Kapepula Kasembele (Democratic Republic of the Congo), Imane Rahmouni (Morocco), Amira Chaabane (Tunisia), and others.

A brief history of marine fish parasitology in Africa

The history of studies on the parasites of marine fishes in Africa is even longer than that on freshwater fish. The first marine parasite recorded from an African fish was the isopod *Anilocra capensis* Leach, 1818, found in the water around the Cape of Good Hope almost 200 years ago (see Smit & Hatfield 2015). However, the number of parasites reported from marine fishes of Africa is considerably lower than that found in freshwater hosts (see Gibson *et al.* 2005 for an exhaustive database).

The most comprehensive data are available on the parasites of marine fishes of South Africa (Smit & Hatfield 2015). Among the parasites reported from this country, the most famous is the myxozoan *Kudoa thyrsites* (described as *Chloromyxum thyrsites* by Gilchrist in 1924), which causes myoliquefaction of the flesh of commercially important fish and it is responsible for significant economic losses worldwide (Henning *et al.* 2013). Regarding myxozoans of South African fishes, the contribution of Czech parasitologists, especially Jiří Lom and Iva Dyková, should be mentioned (Smit & Hatfield 2015).

Parasites of marine fishes of the other countries/regions of the African continent have not been studied so intensively as in South Africa. In the Maghreb, which belongs to the Palaearctic zoogeographical region, studies on fish parasites are mainly focused on commercially important fish, especially perciforms. Numerous studies, mostly faunal surveys, but also ultrastructural studies, have been published in Morocco, Algeria and Tunisia (see, *e.g.*, papers by Lassad Neifar), often in collaboration with fish parasitologists from Spain, France, Italy and other European countries (*e.g.*, Gargouri ben Abdallah & Maamouri 2002, 2005; Marzoug *et al.* 2012a, b, 2014; Bellal *et al*. 2016). The most recent work on marine fish parasites in Africa focused on those infecting commercially important fishes (reviewed by Reed 2015 and Smit & Hadfield 2015).

Reed (2015) reviewed studies on the parasites of marine fishes in sub-Saharan Africa and also found that information is only available from a few countries where concerted efforts have been made by local parasitologists (*e.g.*, Nigeria, Senegal, South Africa). Reed (2015) concluded her review with the statement that: “Aquatic parasitologists (marine and freshwater) in Africa have a tremendous opportunity to rapidly advance this field of research by documenting new species and also recording species assemblages associated with certain hosts in different regions.”

Prospects

Africa, with its extraordinarily rich fish fauna, especially in freshwater, has a big potential to serve as an important model continent for ecological and evolutionary studies on fish parasites and their interactions. However, fish parasites are still poorly known, which makes it impossible to assess reliably their diversity, interrelations, distribution and potential effects on their fish hosts. Future studies on the evolutionary history of individual parasite groups will certainly yield interesting results as indicated by the very few molecular phylogenetic studies that included African fish parasites (*e.g.*, Barson *et al.* 2010; Chibwana *et al.* 2013; Přikrylová *et al.* 2013; Bartošová-Sojková *et al.* 2015; Brabec *et al.* 2015; Vanhove *et al.* 2015). There is also an urgent need to get much more data on host-parasite interactions with focus on potential pathogens of commercially important fish.

The number of African fishes that have not been examined for parasites is extremely high and thus the present knowledge of the parasite fauna of African fishes is fragmentary and incomplete. Similarly, data from a majority of African countries are scarce or even lacking completely. Therefore, future research efforts should be focused on poorly studied fish hosts as well as the regions from where limited or no information is available. Capacity development through training of, and collaboration with, African scholars is crucial in this regard. The application of adequate methods of collecting fish parasites, their processing and evaluation is critical, because much valuable material of African fish parasites has been lost or is not usable due to the application of inappropriate methods.

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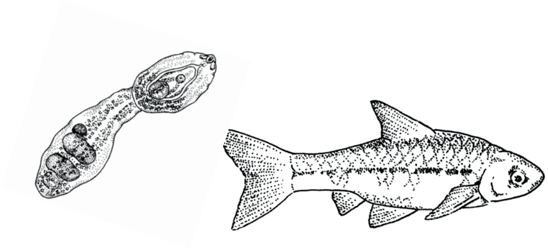
PART 2

FISH AS HOSTS OF PARASITES, THEIR ECOLOGY AND SAMPLING



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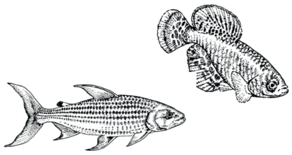

Chapter 2.1.

FISH DIVERSITY AND ECOLOGY

Martin REICHARD

Diversity of fishes in Africa

Fishes are the most taxonomically diverse group of vertebrates and Africa shares a large portion of this diversity. This is due to its rich geological history – being a part of Gondwana, it shares taxa with the Neotropical region, whereas recent close geographical affinity to Eurasia permitted faunal exchange with European and Asian taxa. At the same time, relative isolation and the complex climatic and geological history of Africa enabled major diversification within the continent. The taxonomic diversity of African freshwater fishes is associated with functional and ecological diversity. While freshwater habitats form a tiny fraction of the total surface of aquatic habitats compared with the marine environment, most teleost fish diversity occurs in fresh waters. There are over 3,200 freshwater fish species in Africa and it is likely several hundreds of species remain undescribed (Snoeks et *al.* 2011). This high diversity and endemism is likely mirrored in diversity and endemism of their parasites.

African fish diversity includes an ancient group of air-breathing lungfishes (*Protopterus* spp.). Other taxa are capable of breathing air and tolerate poor water quality, including several clariid catfishes (*e.g.*, *Clarias* spp.; Fig. 2.1.1D) and anabantids (*Ctenopoma* spp.). Africa is also home to several bichir species (*Polypterus* spp.; Fig. 2.1.1A), an ancient fish group endemic to Africa, and bonytongue *Heterotis niloticus* (Cuvier, 1829) (Osteoglossidae), a basal actinopterygian fish. Special adaptations of particular fishes are expected to affect parasite communities.

Functional diversity of African freshwater fishes includes specialised predatory tigerfishes (*Hydrocynus* spp.; Fig. 2.1.1K), weakly electric elephantfishes (Mormyridae; Fig. 2.1.1C), electric catfishes (*Malapterurus* spp.; Fig. 2.1.1B), pufferfishes (*Tetraodon* spp.; Fig. 2.1.1I) and many other specialised forms. Among other unique fishes, Africa has its blind cave fish (*Caecobarbus geertsii* Boulenger, 1921), miniature fishes from rainforest streams (*e.g.*, *Barboides britzi* Conway et Moritz, 2006), small annual killifishes (*Nothobranchius* spp.; Fig. 2.1.1E) that survive annual desiccation of their habitat as dormant embryos encased in dry substrate, or brood parasites that parasitise mouth brooding cichlids and use them as foster parents for their offspring (cuckoo catfish, *Synodontis multipunctatus* Boulenger, 1898; Fig. 2.1.1F). Large functional diversity can evolve even at small temporal and spatial scales, such as in haplochromine cichlids in Lakes Victoria, Malawi and Tanganyika and species of *Labeobarbus* Rüppell, 1835 (Cyprinidae) in

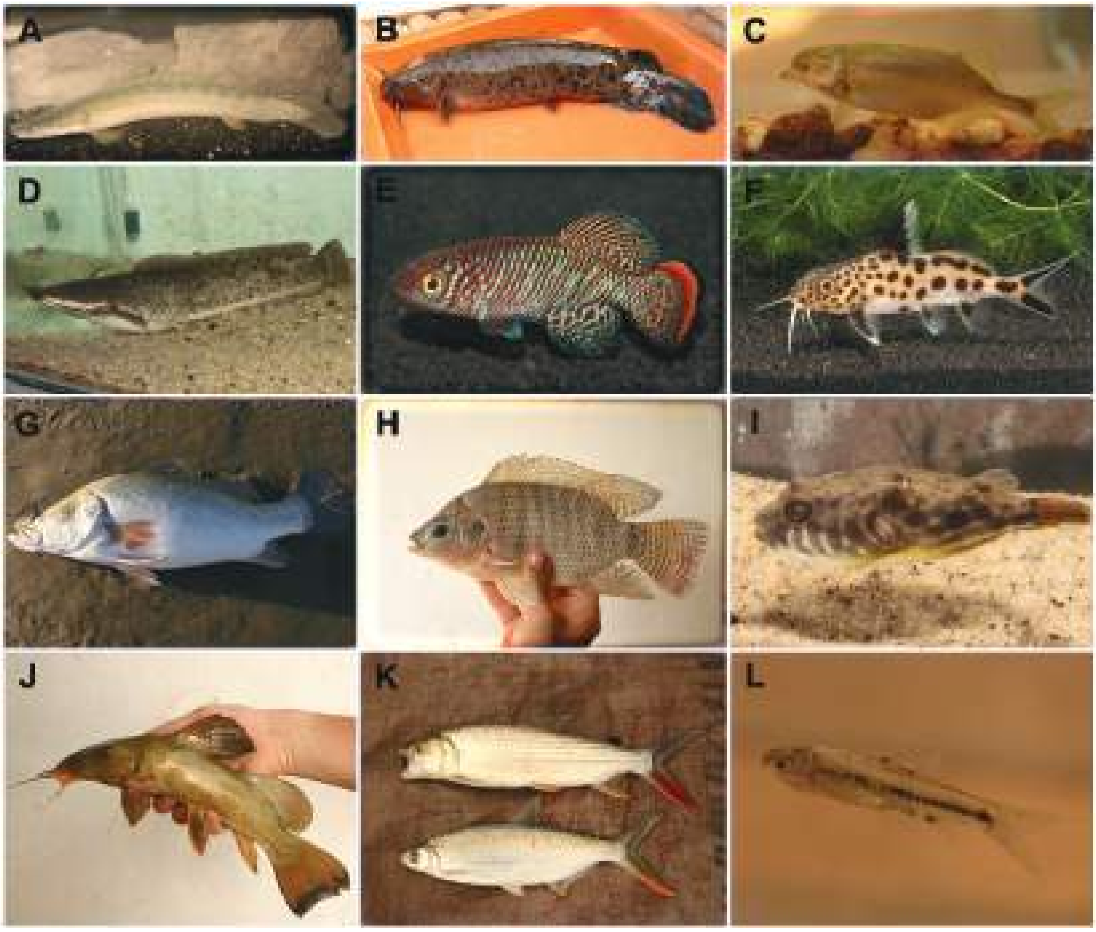

Fig. 2.1.1. Examples of African freshwater fish diversity. **A.** *Polypterus bichir* (Polypteridae); **B.** *Malapterurus occidentalis* (Malapteruridae); **C.** *Marcusenius senegalensis* (Mormyridae); **D.** *Clarias gariepinus* (Clariidae); **E.** *Nothobranchius pienaari* (Nothobranchiidae); **F.** *Synodontis multipunctatus* (Mochokidae); **G.** *Lates niloticus* (Latidae); **H.** *Oreochromis niloticus* (Cichlidae); **I.** *Tetraodon lineatus* (Tetraodontidae); **J.** *Auchenoglanis occidentalis* (Claroteidae); **K.** *Hydrocynus brevis* and *Alestes baremoze* (Alestidae); **L.** *Enteromius niokoloensis* (Cyprinidae). (Photographs by R. Blažek and M. Reichard.)

Fig. 2.1.2. Map of Africa with identification of 10 main ichthyofaunal regions. **1**. Maghreb Province; **2.** Nilo-Sudanian Province; **3.** Congo Province; **4.** Upper Guinea; **5.** Lower Guinea; **6.** Zambezi Province; **7.** East Coast Province; **8.** Southern (Cape) Province; **9.** Quanza Province; **10.** Abyssian Highlands Province (according to Thieme *et al.* 2005). The base map is from Wikimedia Commons: Bamse (self-made) using GMT, CC BY-SA 3.0.

Lake Tana. How such small-scale diversification rates are translated into parasite diversification remains largely unexplored.

The fish diversity in Africa is subject to intense scientific interest, with special attention to understanding their evolution, biology and adaptations, and to explore fish as a resource for local small-scale fisheries and larger scale commercial activities. A better understanding of the diversity and importance of the fish parasite fauna should be based on solid background knowledge of African fish biology and taxonomy.

Zoogeography

The major ichthyofaunal provinces are separated into 10 main continental regions (Roberts 1975; Snoeks *et al.* 2011; Fig. 2.1.2) and Madagascar, though a finer scale resolution to 93 freshwater ecoregions is also available (Thieme *et al.* 2005). The Maghreb Province is the most distinct African ichthyofaunal province. As part of the Palearctic realm, its ichthyofauna displays a high similarity with the European fish fauna (*e.g.*, *Barbus* spp., *Salmo trutta* Linnaeus, 1758, *Cobitis* sp.). The largest province is the Nilo-Sudanian Province, spanning from the River Gambia in the West to the Kenyan coastal drainage in the East. It includes major rivers such as the Nile, the Niger and the Volta, as well as the Lake Chad Basin in its centre. The Congo Province includes the entire drainage of the Congo River, the second largest river basin in the world, with a very high species richness and diversity. It also includes Lake Tanganyika. Two other West African provinces are the Lower and Upper Guinea, separated by the Dahomey Gap and the Volta River. The Upper Guinea includes the coastal rivers of the West African forest region, whereas the Lower Guinea is adjacent to the Congo Province. These regions have been well researched and their ichthyofauna is relatively well known. The Zambezi Province includes rivers flowing eastward to the Indian Ocean from the Zambezi Basin in the North to the Limpopo Basin in the South. It also includes the Okavango Basin. Geographically, Lake Malawi is part of this system, though it has a unique lacustrine ichthyofauna. The East Coast Province includes smaller rivers flowing eastward along the coast of northern Mozambique, Tanzania and southern Kenya, and includes Lake Victoria, with its unique haplochromine cichlid fauna and other lakes in the region. The Southern (Cape) Province includes many temperate rivers south of the Zambezi Province. It has a small number of native (autochthonous) species compared to other provinces (42) and higher-order taxa, but species in the province are often endemic (36 endemic species). The Quanza Province includes a small region of coastal Angolan rivers, with their ichthyofauna being largely unexplored. Finally, the small Abyssian Highlands Province is composed of Lake Tana (with its intra-lacustrine radiation of *Labeobarbus*) and adjacent parts of the effluent rivers.

Main families of fishes

Almost all African freshwater species are continent-endemic and over 40% of the 76 families are restricted to the African continent, which is a relatively high level of endemism at family level. Cichlidae is the most species-rich family (at least 900 species), with the main species diversity in the lakes of East Africa (Tanganyika, Malawi, Victoria), though the number of riverine cichlid species is also high. Cyprinidae (almost 500 species) are typically riverine fishes whose species diversity outnumbers that of the ecologically similar Alestidae (African tetras) by a factor of four (approximately 120 species). Distichodontidae is an endemic family to Africa containing 101 described species. The catfishes are dominated by squeaker catfishes (Mochokidae, 209 species), Claroteidae (86 species) and air-breathing Clariidae (approximately 75 species in Africa). Killifishes are separated into Nothobranchiidae (262 species) and Poeciliidae (about 65 egg-laying species in Africa). The endemic and weakly electric elephantfishes (Mormyridae) include 221 described species (Froese & Pauly 2017). There are also several species from widespread families such as Gobiidae, non-endemic families such as Galaxiidae (in the Southern Province) and many small families endemic to Africa (*e.g.*, Hepsetidae, Pantodontidae). An overview of main freshwater fish families in Africa with estimates of their species richness, general distribution and abundance is shown in Table 2.1.

Ecological guilds

African fishes inhabit all available ecological niches, with examples of species adapted to pelagic and benthic habitats, to strong rapids, swamps, temporary habitats, river margins and deep lacustrine habitats repeated in numerous taxa. Africa harbours native catadromous and anadromous migratory fishes (*e.g.*, Anguillidae) and species with a largely nocturnal lifestyle (*e.g.*, Mormyridae). Several species possess weakly poisonous glands in proximity to sharp fin rays (Mochokidae) and appropriate care should be taken when handling them. Ecological guild largely dictates fish lifestyle and this should aid in the choice of appropriate sampling techniques.

Commercially important fish

Several native African species are commercially exploited in aquaculture in Africa itself and in other continents. Nile tilapia *Oreochromis niloticus* (Linnaeus, 1758) (Fig. 2.1.1H) and a few related species and hybrids are the most popular species in tropical aquaculture, and Nile tilapia is successfully cultured across Africa. Information on their parasite fauna and its dynamics may be critical for the success of aquaculture at high population densities. Some species became important for larger-scale fisheries, including many lacustrine cichlids and the Nile perch *Lates niloticus* (Linnaeus, 1758) (Fig. 2.1.1G) in East Africa, West African pygmy herring *Sierrathrissa leonensis* Thys van den Audenaerde, 1969 in Lake Volta and Tanganyikan kapenta sardine *Limnothrissa miodon* (Boulenger, 1906) in Kariba and Cahora Bassa reservoirs and in Lakes Kivu and Tanganyika. Many other fishes are important for local sustainable fisheries, such as *Clarias* spp. (Fig. 2.1.1D) or riverine migratory species. Particularly, the larvae of digeneans with a complex life cycle may opportunistically infect commercial species as intermediate hosts and may reduce the commercial value of these species. Their final hosts are predatory fish or birds.

Table 2.1. An overview of main African freshwater fish families with estimates ot their species richness, general distribution and abundance

Order	Family	Species richness	Distribution
Lepidosireniformes	Protopteridae	1 genus, 4 species	widespread
Polypteriformes	Polypteridae	2 genera, 10 species	widespread
Anguilliformes	Anguillidae	5+ species	Indian and Mediterranean drainages
Clupeiformes	Clupeidae	13 genera	widespread, abundant
	Denticipitidae	1 species	restricted: West Africa
Osteoglossiformes	Arapaimidae	1 species	widespread, introductions
	Pantodontidae	1 species	relatively widespread: West Africa
	Notopteridae	2 genera, 3 species	widespread: West Africa
	Mormyridae	221 species	widespread, abundant
	Gymnarchidae	1 species	widespread: West Africa + Soudanian
Salmoniformes	Salmonidae	several introduced species	native to Magreb (1 species), introduced to East and Southern Africa
Osmeriformes	Galaxiidae	1 species	endemic to South Africa
Gonorynchiformes	Kneriidae	4 genera, 10 species	geographically widespread but locally uncommon
	Phractolaemidae	1 species	endemic to West Africa
Hepsetidae	Hepsetidae	1 genus, 6 species	West, Middle and South Africa
Characiformes	Alestidae	19 genera, 119 species	widespread, abundant
	Distichodontidae	17 genera, 101 species	widespread, abundant
	Citharinidae	3 genera, 8 species	widespread, abundant
Cypriniformes	Cyprinidae	500+ species	widespread, abundant
	Nemacheilidae	1 species	endemic to Ethiopia
	Cobitidae	1 species	endemic to Maghreb
Siluriformes	Bagridae	1 genus, 11 species	widespread
	Claroteidae	13 genera, 86 species	widespread, abundant
	Schilbeidae	5 genera, 33 species	widespread, abundant
	Amphiliidae	13 genera, 68 species	widespread
	Clariidae	13 genera, 75 species	widespread, abundant
	Malapteruridae	2 genera, 21 species	widespread
	Mochokidae	9 genera, 200+ species	widespread, abundant
Perciformes	Poecilidae	7 genera, 65+ species	widespread, abundant
	Nothobranchiidae	12 genera, 250+ species	widespread, abundant
	Channidae	1 genus, 3 species	West and Central Africa
	Latidae	1 genus, 9 species	widespread
	Centrarchidae	2 genera, 4 species	introduced in South Africa

	Percidae	1 species	introduced in South Africa
	Nandidae	2 genera, 2 species	West Africa
	Cichlidae	900+ species	widespread, abundant
	Anabantidae	3 genera, 32 species	widespread
	Gobiidae	30+ genera, 90+ species	widespread
	Eleotridae	approx. 25 species	widespread
Synbranchiformes	Mastacembelidae	1 genus, 45 species	widespread
Tetraodontiformes	Tetraodontidae	6 species	geographically widespread

Estimates on taxonomic richness, distribution and abundance are based from Skelton (1988), Darwall *at al.* (2005) and Froese & Pauly (2017).

Non-native species and other threats to local fish fauna

Many fish species were translocated within Africa, especially for commercial use in aquaculture. The best-known examples include Nile tilapia (Fig. 2.1.1H) and Nile perch (Fig. 2.1.1G). Nile tilapia is more aggressive and competitively superior to other tilapias (and many other cichlids that share its ecological niche) and has displaced them from many habitats. In addition, Nile tilapia hybridises with native species of *Oreochromis* Günther, 1889, further threatening their existence. Predictably, transport of Nile tilapia includes transport of their parasites, with potential spill-over and spill-back effects on local fish fauna. Nile perch, native to West African rivers and Lake Turkana, has been introduced to other places to supplement local fisheries, sometimes with catastrophic consequences for the local fish fauna (exemplified by the Lake Victoria case). Other fish species being translocated worldwide can be found in Africa, including Eurasian cyprinids such as common carp *Cyprinus carpio* Linnaeus, 1958, Chinese silver carp *Hypophthalmichthys molitrix* (Valenciennes, 1844) or grass carp (*Ctenopharyngodon idella* Valenciennes, 1844) and several North American centrarchids, including largemouth bass *Micropterus salmoides* (Lacépède, 1802). These species, however, are mainly constrained to relatively colder parts of Africa such as the Southern Province. Research on the parasite fauna of non-native species is interesting as missing parasites are often linked to the success of introductions.

The African fish fauna is also threatened by water pollution (especially inorganic pollution near mining sites and sedimentation from soil erosion), river regulation (dams preventing upstream migration), conversion of wetlands to rice paddies and overfishing in particular habitats. Especially water pollution is expected to have a major impact on the parasite fauna. Notably, heavy metals tend to accumulate in parasites and fish parasites might be used as potential biomarkers for mining-related water pollution (Sures *et al.* 1999).

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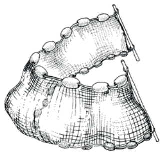

Chapter 2.2.

SAMPLING OF FISH FOR PARASITOLOGICAL INVESTIGATION

Pavel JURAJDA

Freshwater habitats

Freshwater ecosystems are highly diverse habitats that vary in size, depth, bottom substrate (rocks, pebbles, mud, sand), chemistry (acidic, neutral, alkaline, oxygen content, productivity, etc.), availability of cover (woody debris, macrophytes, rocks) and character (flowing, standing). Flowing waters include creeks, streams and rivers, whereas standing waterbodies (stagnant or fresh) may be natural (swamps, pools, backwaters and lakes) or man-made (ponds and reservoirs). Canals are a special case of a man-made, standing or slow flowing, sometimes stagnant, waterbody that may have been stocked with fish.

The combination of these geomorphological and chemical factors will dictate not only which species can exist in the waterbodies, but also which fish sampling methods can be applied. Boats are needed for deeper waters, as sampling by wading can be dangerous and inefficient in waters > 1 m with a strong current. Similarly, obstacles such as rocks and dense aquatic vegetation limit the efficiency of methods such as seine netting. Physico-chemical parameters may also limit sampling efficiency, with fish reacting to sampling activity earlier and escaping in clear waters and visibility severely limited in turbid waters. Electrofishing is also ineffective in waters with very low conductivity (low salt content), which is typical for tropical countries.

Sampling methods

Choosing the most appropriate sampling method (Fig. 2.2.1) is critical for the effectiveness of sampling in a given locality and habitat (Figs 2.2.2, 2.2.3). The sampling methods available may be passive or active (Table 2.2). Passive methods generally involve simple and relatively cheap sampling equipment that is placed in the habitat being sampled and left for a defined period. Fish are generally caught as a result of their own movement. Active methods require an operator or team that actively attempts to catch the target fish. Such methods tend to be costlier than passive methods, both in terms of equipment and manpower, but active methods are generally more efficient and less time-consuming than passive methods.

Gill nets (passive; Fig. 2.2.1) are vertical panels of (usually) monofilament nylon netting that are set in a straight line and held in position by weights and floats. Gill nets are very good at intercepting fish that naturally migrate or move frequently;

they are far less effective in catching sedentary or territorial species. Gill nets may have a single mesh size, in which case they are size-specific, or can consist of several panels with different mesh sizes (Nordic gill nets) in order to reduce size selectivity. Gill nets may be placed near the bottom to sample benthic fish, or can be installed nearer the surface to sample mainly pelagic fishes. Gill nets can be used in a wide range of habitats, though their use is generally limited to areas free of obstructions, snags and floating debris, and to localities with little or no current. As monofilament nylon is very hard to see underwater, even in clear waters, animals other than fish may also get caught in the nets. In their efforts to escape, they may become entangled and, if air breathing, may drown. While observers may be able to release smaller animals or unwanted fish, it may be too dangerous to approach larger species such as crocodiles or hippos.

Traps (passive; Fig. 2.2.1) include various types of fyke net, wicker cages and pots, all of which are constructed with a small funnel opening which fish can enter easily but find very difficult to exit. Traps may be baited to increase the probability of success. They are generally used in shallow regions of lakes and reservoirs but can also be used to sample fish in slow-flowing streams, rivers and backwaters. Unlike most other methods, they may also be used in habitats with relatively dense vegetation such as marshes or swamps. Pot traps tend to be most effective in capturing bottom-dwelling species seeking food or shelter. Traps should be checked periodically (at least every 4 to 6 hours) to prevent a build-up of fish, which could lead to fish damage or predation. High densities of trapped fish can

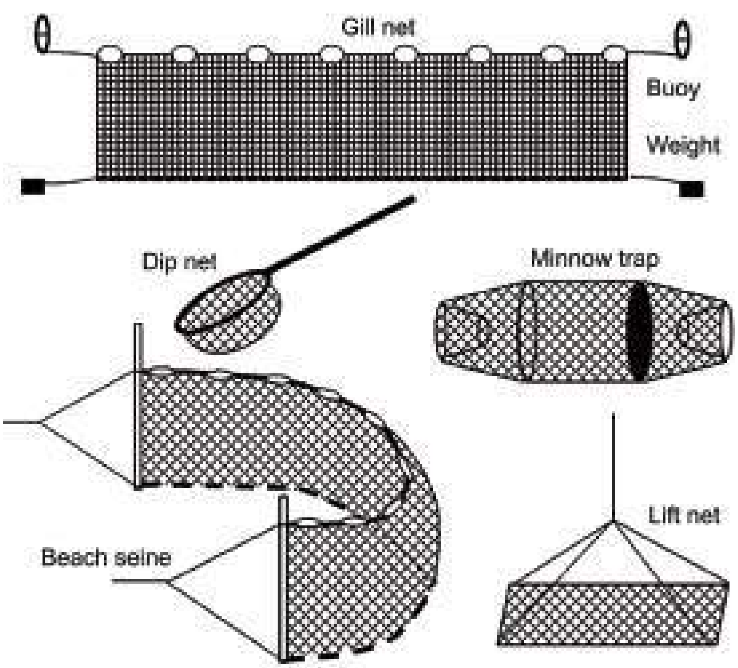

Fig. 2.2.1. Illustrations of some basic active and passive methods of sampling fish.

Fig. 2.2.2. A. A shallow, fast flowing turbid stream, River Nieri Ko, Senegal; **B.** A large slow flowing deep lowland River Gambia, Senegal. (Photographs by R. Blažek.)

Fig. 2.2.3. A. A large deep lake (Lake Tanganyika, Burundi); **B.** A shallow lake with aquatic vegetation, Mozambique. (Photographs by R. Blažek.)

Fig. 2.2.4. A. Beach seining in Lake Turkana, Kenya; **B.** Dip netting in marshes, Mozambique. (Photographs by R. Blažek.)

Fig. 2.2.5. A. Cast net sampling in Lake Turkana, Kenya; **B.** Sampling fish from Uvira fish market, Democratic Republic of the Congo. (Photographs by R. Blažek.)

also lead to the transfer of ectoparasites such as monogeneans between hosts, which may bias subsequent ecological studies.

Rod and line (passive) is a simple angling method that can potentially be used for sampling fish for scientific purposes. Note, however, that use of rod and line tends to be highly selective for particular species (especially predators) and/or sizes of fish, depending on the gear or bait used, and it may take a long time to catch sufficient numbers. On the other hand, the material needed is relatively small and light, making it easy to transport. Efficiency will largely depend on the experience of the user and knowledge of local conditions.

Beach seining (active; Figs 2.2.3, 2.2.4) utilises a large net of uniform mesh size consisting of two ‘wings’ and a ‘purse-like’ central section that holds the catch. As its name implies, a beach seine is typically used in shallow waters, the net generally being set in a semicircle around the target area, either by boat or by wading, and dragging the net back to shore. The net is kept open while deployed by floats on its upper line, while weights on the lower line ensure the net stays close to the bottom substrate. A larger area may be fished by attaching long towing lines to each end of the net. Seine nets are most effective for catching near-shore species or fish that concentrate near-shore periodically. Beaches should preferably be free of obstacles (*e.g.*, rocks, tree stumps) or heavy vegetation.

Push net, dip nets, lift nets and cast nets (active; Figs 2.2.3-2.2.5) are all simple tools that can be used by a single person. Push and dip nets, which are made of netting attached to a round or triangular frame fixed on the end of a pole, are used to collect small fishes along the bank or in places with dense vegetation where other methods may be impractical. Fish are generally pursued by the user. Lift nets or cast nets are used to catch small schooling fish in open waters. Lift nets are left on the bottom (or lowered in deep lakes) by the user who retains hold of the net by a line. Bait (or a light at night) can be used to concentrate fish over the lift net, which are then caught by quickly lifting the net out of the water. Cast nets are circular nets with weighted edges that are thrown so that they cover the fish. The net is closed and retrieved by pulling on a retaining line. The use of cast nets is restricted to areas free of obstacles or plants. Experience is needed to cast the net successfully, allowing it to hit the water completely open over the target fish.

Electrofishing (active) works on the principal of galvanotaxis, whereby direct current (DC, sometimes pulsed) electricity flowing between a submerged cathode and anode causes a muscular convulsion in the fish, causing it to swim toward the anode where it can be caught with a dip net. The cathode, a long, braided steel or copper cable, trails behind the operator and the anode is operated by a switch on a long pole, the operator directing the anode toward the target fish or site. At least two people are required for effective electrofishing, one to operate the anode and the other to catch the fish. The electrical current is produced either by a battery-powered backpack or a petrol-powered generator that remains stationary on the bank or is placed in a boat. The effectiveness of electrofishing will be influenced by a range of biological, technical, logistical and environmental factors. The pulse rate and intensity of the electric field produced can strongly influence the size and nature of catch, whereas conductivity of the water will influence the shape and extent of the electric field, and thus the field's ability to induce galvanotaxis.

Electrofishing is limited to sites with clear water, conductivity of 100-600 µS/cm and a depth of ≤ 1 m. Electrofishing is much less effective in waters with low conductivity (low dissolved salt concentration), whereas a stronger generator will be required in waters with high conductivity. Electrofishing is particularly efficient at sites with obstacles (*e.g.*, vegetation, woody debris, rocks) and in running waters, where other methods may be inefficient or impossible to use. In some cases, electrofishing may harm fish by causing muscle spasms that damage the fish's backbone; a problem more common and severe in longer fish. Used correctly, however, electrofishing causes no permanent harm to the fish, which will recover minutes after being caught. Note that the use of any electrical equipment in and around water is dangerous. For operator safety and for efficient and successful sampling, all equipment should be designed specifically for electrofishing and all personnel adequately trained in its use. Many countries also require that the user is licensed. In some countries, electrofishing is illegal.

Table 2.2. A simplified comparison of passive and active methods of sampling fish

	Method	Advantages	Disadvantages
Passive methods	Gill nets, traps, rod and line	Simple manipulation, cheap, low man power, light, easy to transport	Non-selective for species and size, may damage fish, time-consuming, fish may die in gill nets
Active methods (simple)	Dip nets, scoop nets, hand nets, cast nets	Simple manipulation, cheap, low man power, easy to transport, can target specific species/ sizes	Practice needed, less efficient, time consuming
Active methods (technical)	Beach seine, electrofishing	Mobile, faster, can target specific species/ sizes, greater numbers caught	Expensive, practice and/or training needed, transport difficult, higher man power

For more information about fish sampling methods see Bohlin *et al.* (1989), Murphy & Willis (1996), Lapointe & Corkum (2006) and Pierce *et al.* (1990).

Fish sampling strategy for parasite community studies

In comparison with fish population or community studies, different criteria apply when sampling fish for parasitological surveys. Instead of obtaining a general description of fish assemblage structure (*e.g.*, species richness, density, population structure) for the sample site, the operator aims to obtain a representative sample (number) of specific target species and size categories. Sampling area and timing of sampling should be adapted and aimed specifically at where and when the target fish are most likely to occur.

For ectoparasites found on the skin and/or fins, sampling methods should involve minimal handling of the fish as contact could damage or remove the parasites. Appropriate methods include electrofishing, angling, pot traps or small beach seine nets. For endoparasites and gill parasites, more robust sampling methods (*e.g.*, large seine nets) can be used as they are unlikely to affect parasite numbers. It may even be possible to obtain fresh fish from local fishermen or markets.

Fish transport and treatment

Whatever the method used to sample the fish, they should always be handled carefully prior to dissection to prevent loss or transfer of parasites. The fish should be maintained and transported in water taken from the sampling site and only wet hands or dip nets used for manipulating the fish. During transport and storage, oxygen levels should be maintained with an aerator or oxygen cylinder. Fish should be transported to the laboratory as soon as possible and kept alive until examination. Parasitological examination should be carried out no more than three days after capture; any later and parasites may die or reproduce, biasing infection parameters (Kvach *et al.* 2016). During storage, the fish should not be fed and the density kept relatively low to prevent host mortality and transfer of parasites. The tank should be placed in shade to maintain a stable temperature. In the case of untimely mortality, complete freezing should be avoided if possible as it can affect morphological and ultrastructural observations (for transportation or temporary storage, fish may be kept for a short while in crushed ice).

Fish should always be handled with full regard to the animal's welfare (in line with local regulations) and euthanised using the most 'humane' methods available.

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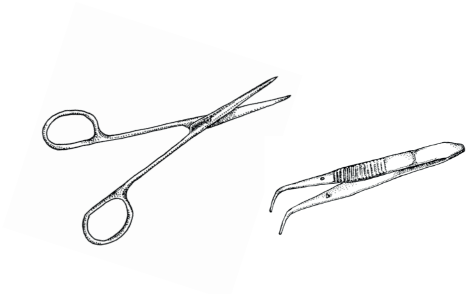
PART 3

BASIC METHODS TO STUDY FISH PARASITES



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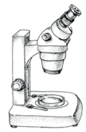

Chapter 3.1.

IMPORTANCE OF SAMPLING DESIGN: HOW TO COLLECT DATA ON FISH PARASITES

Milan GELNAR, Nico SMIT & Maarten P.M. VANHOVE

Introduction

There is no doubt that the importance of fish parasites is related directly to the importance of fish they may affect (Hoffman 1999). It is well known that fish are an excellent source of complex proteins, they provide an important recreational asset, both for sport fishing and as one of the attractions of nature. In addition, a lot of fish species are also very important for development of various types of aquacultures, and finally, fish and their parasites also represent an important and interesting subject for science including ichthyoparasitology investigating parasites as potential causative agents of various fish diseases and also in ecotoxicology and evolutionary ecology (*e.g.*, Woo 1995; Khalil & Polling 1997; Hoffman 1999; Scholz 1999; Alvárez-Pellitero 2008; Eiras *et al.* 2008a,b; Sitjà-Bobadilla 2008; Buchmann *et al.* 2009; Leatherland & Woo 2010; Woo & Bruno 2011; Woo & Buchmann 2012).

Many years ago, Lester (1984) has reviewed methods for studying the effect of parasites on feral and cultured fish. Before fish parasitic diseases are effectively treated and controlled, the study of fish should follow a logical pattern:

- identify the parasite;
- obtain a thorough knowledge of its life history, which may be simple (direct or monoxenous) or very complicated (indirect or complex);
- learn the ecological requirements of the parasite, such as host specificity, optimum temperature, pH, nutrition, and other metabolic requirements;
- map the geographical range of the parasite;
- determine effect of immunological mechanisms of the host on the parasite, and *vice versa*;
- study control and treatment methods.

Hierarchical structure of parasitology

Parasitology and especially evolutionary ecology of parasites can be studied at three hierarchical levels: (1) organism, (2) population and (3) community (see Fig. 3.1.1). The smallest scale of study in parasite ecology is the individual parasitic organism, but parasitologists also deal with populations of parasite individuals of the same species, and with communities made up of several populations of different species (*e.g.*, Kennedy 1976; Esch *et al.* 1990; Esch & Fernández 1993; Rohde 2005; Poulin 2007).

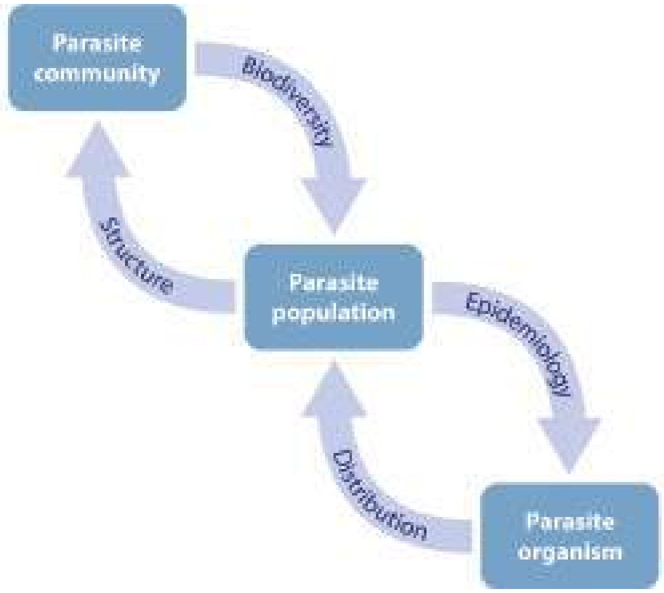

Fig. 3.1.1. A schematic representation for the three hierarchical levels of organisation of parasite-host associations. (Illustration by M. Luo and M. Gelnar.)

Sampling of parasitic organisms

Correct diagnosis is essential not only for parasite species identification but also for effective treatment and control of any fish disease. This means that there needs to be a consensus on the names and terms used in the identification process. Therefore, before we begin to consider a specific parasite, it is necessary to have an understanding of how the taxonomic system works and its relevance to parasitology (*e.g.*, Gussev 1978, 1985; Halton *et al.* 2001; Pugachev *et al.* 2010; Gunn & Pitt 2012). Those who study the classification of organisms are called taxonomists and they arrange organisms into hierarchical categories to reflect their assumed relationships.

Table 3.1.1. Taxonomic hierarchy with specific reference to the monogenean parasite *Paradiplozoon homoion homoion*

Taxonomic division	Taxon name	Common name
Super kingdom	Opisthokonta	
Kingdom	Animalia	animals
Subkingdom	Bilateralia	
Branch	Protostomia	
Phylum	Platyhelminthes	flatworms
	Neodermata	
Class	Monogenea Carus, 1863	
Subclass	Oligonchoinea Bychowsky, 1937	
Order	Mazocreaidea Bychowsky, 1957	
Suborder	Discocotylinea Bychowsky, 1957	
Family	Diplozoidae Palombi,1949	
Subfamily	Diplozoinae Palombi, 1949	
Genus	*Paradiplozoon* Akhmerov, 1974	
Species	*Paradiplozoon homoion*	
	(Reichenbach-Klinke, 1961) Akhmerov, 1974	
Subspecies	*Paradiplozoon homoion gracile*	
	(Bychowsky et Nagibina, 1959) Akhmerov, 1974	

Note: not all taxonomists agree with the same classification scheme. For example, some specialists prefer to divide the Monogenea (or Monogenoidea according to other authors) into different subclasses:

- Monopisthocotylea (= Polyonchoinea) and Polyopisthocotylea (excluding Polystomatidae and Sphyranuridae = Oligonchoinea) – Bychowsky (1957)
- Polyonchoinea, Polystomatinea and Oligonchoinea – Lebedev (1989)
- Polyonchoinea and Heterochoinea (including two infra-subclasses Polystomatoinea and Oligonchoinea) – Boeger & Kritsky (2001)

Selection of proper morphometrical characteristics and effective laboratory techniques

There is no doubt that the usage of selected morphological/anatomical characters and some metrical parameters represents the most important step in parasite species identification (*e.g.*, Rubbi 1994; Rizzuto & Fasolato 1998; Lacey 1999).

As an example, the following morpho-anatomical characteristics can be recommended to be used for the identification of monogeneans (Gussev 1978, 1985; Pugachev *et al.* 2010).

- Shape and size of the body and haptor
- Structure of the anterior end; presence or absence of lobes, lappets, suckers and their number
- Structure of the tegument, its thickness and presence or absence of folds, scales or thorns
- Presence or absence of eyes, their number and structure
- Shape, number, arrangement, orientation and size of haptoral structures
- Structure and size of the copulatory organ and vaginal armament
- Structure of the intestine
- Number of testes
- Shape and arrangement of the ovary
- Relative position of the ovary and testes
- Number, shape and position of the gland reservoir of the copulatory organ
- Course of vas deferens and shape of the seminal vesicle
- Position of the genital and vaginal pores, course and armament of the vaginal duct and seminal receptaculum (if present)

It should also be pointed out that correct identification of the fish host is extremely important. Erroneous identification of hosts or infection site may result in misleading conclusions. It is therefore recommended to always take a picture of the host and to fix a small piece of its tissue (fins, liver or muscle) in molecular-grade ethanol for DNA-based identification, or to fix and preserve the entire host specimen as a voucher.

Sampling of parasite populations

Parasite populations vary in size over short and long-time scales and are affected by biotic and abiotic environmental factors. Some of these factors cause changes in parasite numbers, whereas others reduce the amplitude of fluctuations around an equilibrium population size.

Parasite populations are invariably fragmented into as many subgroups as there are infected individuals in a host population. For practical reasons, it is easier to consider only a single parasite life stage, such as adult parasites only, when defining a population (*e.g.*, Esch et al. 1990; Esch & Fernández 1993; Hanski 1999; Šimková *et al.* 2002; Poulin 2007). Thus, a parasite population consists of all adult parasites in all individual hosts of a host population; it is subdivided into numerous infrapopulations of unequal size, each inhabiting a different host individual. Infrapopulations are ephemeral groups, lasting no longer than the host's lifespan. Offspring issued from different infrapopulations have the opportunity to mix outside hosts and reassemble in new combinations to form new infrapopulations in new individual hosts. The infrapopulation fragmentation is thus temporary and changes continually from generation to generation (for a schematic illustration of factors affecting parasite populations, see Fig. 3.1.2).

To date, the population biology of parasites has been investigated on three different fronts (Poulin 2007):

1. The dynamics of parasite populations can be modelled mathematically, usually with a few simplifying assumptions (*epidemiological approach*).
2. Empirical studies of field populations have highlighted the many density-dependent and density-independent mechanisms acting to regulate parasite abundance over time in specific systems (*ecological approach*).
3. Genetic structure among infrapopulations and among populations allows us to determine transmission processes and estimate the frequency of exchange of individuals among populations (*genetic approach*).

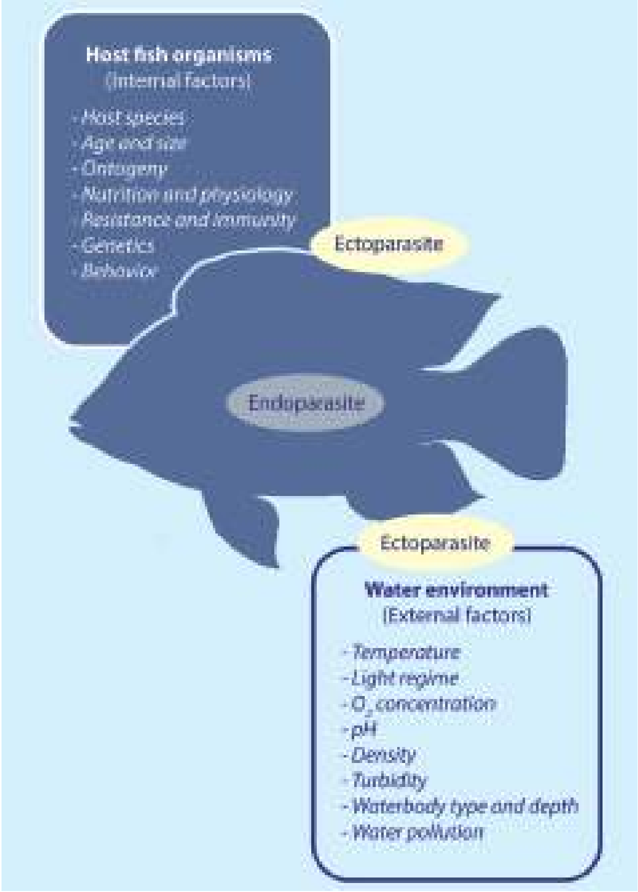

Fig. 3.1.2. A schematic representation of parasite-host interactions in an aquatic environment. (Illustration by M. Luo and M. Gelnar.)

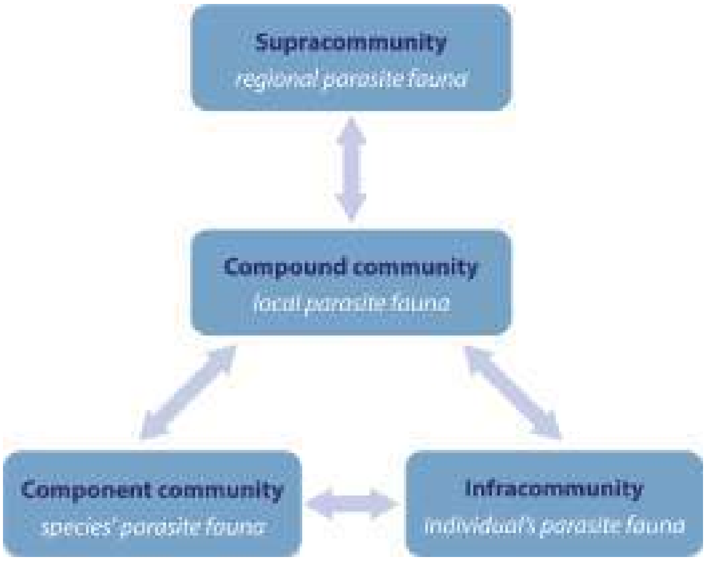

Fig. 3.1.3. A schematic representation of the hierarchical organisation of parasite supracommunity, compound community, component community and infracommunity. (Illustration by M. Luo and M. Gelnar.)

Sampling of parasite communities

The assemblage consisting of all parasites of different species in the same host individual, whether they actually interact or not, forms an infracommunity (*e.g.*, Esch *et al*. 1990; Bush *et al.* 1997). Infracommunities are subsets of the component community, which consists of all parasites exploiting the host population. In theory, infracommunities can range from highly structured and predictable sets of species, to purely stochastic assemblages of species coming together entirely at random (see Fig. 3.1.3 for a schematic illustration of parasite community structure).

Interactions among parasite species are one of the main forces that can shape infracommunity composition and structure and give it a non-random structure. In isolationist parasite communities, where interactions are negligible either because of very narrow niches or small infrapopulation sizes, the co-occurrence of species in hosts is not expected to deviate from that expected by chance (*e.g.*, Esch *et al.* 1990; Esch & Fernández 1993; Rohde 2005; Poulin 2007).

Recommendations for parasite community sampling design

The vast majority of available studies on parasite community ecology are based on the examination of patterns observed in one or a few samples of host individuals, patterns existing among different infracommunities sampled at one point of time. These provide a snapshot of what the parasite infracommunities looked like at the time of sampling, but no information on their development through time, starting from the moment the first parasite arrived on a host. Very few investigations have attempted a longitudinal survey of parasite infracommunities, beginning with uninfected hosts, either young individuals or animals reared in captivity, that were allowed to recruit parasites under natural conditions (*e.g.*, Poulin 1996a,b; Poulin & Rohde 1997; Bagge & Valtonen 1999; Poulin & Valtonen 2002; Šimková *et al.* 2002, 2004; Vidal-Martínez & Poulin 2003). For hypothetical determinants of parasite community structure in real environmental conditions (see Fig. 3.1.2).

Collection of data

Parasitologists, like ecologists and other biologists, collect data to be used for testing hypotheses or describing nature. Modern science including parasitology proceeds by conjecture and refutation, by hypothesis and test, by ideas and data, and it also proceeds by obtaining good descriptions of ecological events. Parasitology like ecology is an empirical science that cannot be done solely on the blackboard or on the computer; it requires data from the real world. However, ecological data on parasites do not say everything about ecology of parasites.

Data represent only one half of this science; ecoparasitological hypotheses are the other half. Some evolutionary parasitologists even feel that hypotheses are more important than data themselves, while others argue the contrary. The central tenet of modern empirical science is that both are necessary. Hypotheses without data are not very useful, and data without hypotheses are wasted (*e.g.*, Krebs 1999; Henderson 2003). One problem that all research fields face is: what to measure? So selection of good, relevant and correct data is essential for the study and understanding of ecological or parasitological systems.

Host fish as habitat and sampling unit

Selection of a suitable and proper habitat unit is among the key questions in sampling design in the ecology of free living animals. In the case of parasites, a host organism represents the environment colonised and inhabited by parasites and due to that host organism, infrapopulation and infracommunity or local host population, metapopulation and component community can be conceptually identical to the concept of habitat and sampling units for free-living animals, respectively (see Fig. 3.1.3).

At the outset, a scientist must be sure about the problem he/she is proposing to investigate. As it is normally impossible to count and identify all the animals in a habitat, it is necessary to estimate data on the population or community by sampling. Naturally, these estimates should have the highest possible accuracy in relation to the effort spent. This requires a plan that includes a sampling program stipulating the number of samples, their distribution and their size. For example, the number of hosts is typically seen as sufficient to characterise a population at a given point in time. The importance of careful formulation of hypotheses to be tested cannot be overstressed (*e.g.*, Southwood & Henderson 2000; Sutherland 2006).

Sampling design and field work

In community studies, preliminary work should explore species richness and potential problems with species identification. The appropriate degree of taxonomic discrimination must be decided as it is important to maintain a consistent taxonomy. Sample sorting and species identification are often the most labour-intensive parts of a study and it may be useful to carry out a pilot trial to assess the effort required. Planning of the timing requires knowledge of life cycles. Preliminary work will be necessary to gain some knowledge of the occurrence of parasites to be studied.

The first decision concerns the scale of the environment to be sampled. A correct definition of the target population or community is essential: if too small, it may not produce results representative of the structure as a whole; if too large, it will waste resources. The second decision must be to define the accuracy or precision of the population estimates required. These decisions must be taken by considering both the objectives of the study and the variability of the system under study.

According to Henderson (2003), the following elements should be considered in any preliminary sampling design for populations of a host fish and for populations and communities of its parasite species.

- The need for sampling
- The scale of the study
- Safety
- Care for the environment and animal welfare
- Taxonomy
- Recording, labelling and noting down observations
- Data security and processing
- Effect of the time of year on sampling
- Effect of the time of day on sampling
- Size of population and community estimate
- Definition of the habitat unit
- Proper selection of unit area for sampling
- Subdivision of the habitat unit
- Statistical considerations

The selection of habitat and sampling unit for parasite ecology research

In general, the criteria for sample unit selection are, for parasites, broadly those of Morris (1955), where the term ‘habitat unit’ is identical with the term metapopulation of the parasites on a local metapopulation of host fish and the term ‘sample unit’ is identical with infrapopulation/infracommunity of fish parasites infecting the above mentioned metapopulation of host fish (*e.g.*, Krebs 1999; Southwood & Henderson 2000; Henderson 2003).

- All units of the environment must have an equal chance of sampling.
- It must have environmental stability.
- The proportion of the population using the sample unit as a habitat must remain constant.
- The sampling unit must lend itself to conversion to unit areas.
- The sampling unit must be easily delineated in the field.
- The sampling unit should be of such a size as to provide a reasonable balance between the variance and the cost.
- The sampling unit must not be too small in relation to the animal's size, as this would have edge-effect errors.
- The sampling unit for mobile animals should approximate the average ambit of an individual.

Conclusions – Top 10 golden rules

- Not everything that can be measured should be.
- Find a problem and state your objective clearly.
- Collect data that will help achieve your objective and make a statistician happy.
- Some ecological questions are impossible to answer at the present time.
- With continuous data, save time and money by deciding on the number of significant Figures in the data before you start field work/an experiment.
- Never report an ecological estimate without some measure of its possible error.
- Be sceptical about the results of statistical tests of significance.
- Never confuse statistical significance with biological significance.
- Code all your ecological data and enter it on a computer in some machine-readable format.
- Garbage in, garbage out.

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Chapter 3.2.

PARASITOLOGICAL EXAMINATION OF FISH (DISSECTION)

Tomáš SCHOLZ, Eva ŘEHULKOVÁ & Roman KUCHTA

Introduction

Parasitological examination, *i.e.*, dissection or necropsy, is the basic method necessary to obtain parasites, especially for endoparasites (some macroscopical ectoparasites can be taken from live fish without their euthanasia). The extent of the examination depends on the purpose of a given study and the group of parasites studied because different methods are used to study eukaryotic microorganisms (parasitic protists and myxozoans), ectohelminths (Monogenea), endohelminths (Trematoda, Cestoda, Acanthocephala and Nematoda), and mostly ectoparasitic crustaceans. Therefore, the methods used in studies of these four principal groups of parasites are described separately in the following chapters (3.3.1-3.3.4). The present text provides only basic information about the most important requirements.

Basic requirements and rules

Equipment and facilities

Examination of fish usually does not require extremely sophisticated equipment and facilities, especially if focused only on those groups of parasites that do not need to be handled with special techniques. Overall, inspecting fish for eukaryotic microorganisms (see chapter 3.3.1) and monogeneans (see chapter 3.3.3) is more complicated; good optics including a light microscope and special chemicals are needed. In contrast, dissection of fish for some large-sized endohelminths can be done even without the use of a dissecting microscope (or just with a simple magnifying glass), but this does not enable the researcher to find all endoparasitic helminths, especially if they are tiny (< 1 mm). Therefore, the best recovery technique for any parasite group is observation of organs with a dissecting (helminths and parasitic crustaceans) and compound (eukaryotic microorganisms) light microscope. Since some helminths, especially monogeneans, are very tiny and translucent, a dissecting microscope equipped with bottom light (transmitted illumination) is preferred to effectively shed light on these parasites.

For dissection of fish in the field, a table is needed on which fish are examined, dissecting tools, several Petri dishes of different sizes, plastic pipettes, sample storage and transport equipment (vials, tubes, microscopic slides, coverslips and boxes) for fixed parasites, nail varnish to fix coverslips, a burner, water and/or saline, fixatives and a camera. Headlights or torches may help find parasites in the organs examined if electricity is unavailable. Containers with aeration to keep living fish should also be available because fish euthanised just before dissection should be used (see below).

Catching fish for examination

Since ectoparasites can be lost during capture and transport of live fish to the place of examination, catch methods that do not damage the external surface, *e.g.*, electrofishing, sport fishing, scap net, small trawl or seine (see chapter 2.2), should be used. Methods that damage the fish (*e.g.*, gill nets) cause substantial injury and fish captured by such a method may suffer high mortality. Care has to be taken not to disturb the outer surface of fish. In particular, the fish surface should not dry up because this would incur the loss of ectoparasitic protists, crustaceans and monogeneans from the skin and fins. To become familiar with the general situation in the fauna of fish parasites in a locality, the fish sample should include at least 10-15 specimens of each fish species.

Condition of fish

The freshness of the hosts examined is a key factor that considerably influences the quality of parasites found, because decomposition and autolysis of their tissue and surface is very fast following the host's death. This negatively affects subsequent processing such as staining and light or scanning electron microscopic (SEM) observations. If fresh hosts cannot be examined, fish should be placed on ice to slow down autolysis of their tissues including their parasites, and examined as soon as possible (within several hours). Examination of dead fish in the field using a provisional laboratory is recommended rather than loosing time by transporting the fish for several hours to the laboratory. However, hosts should not be frozen, because parasites from frozen hosts may be deformed (contracted or artificially relaxed) and their tissues will have disintegrated, making them unsuitable for reliable morphological characterisation and correct species identification. In the case of protists, they can be completely lost. Hosts from fish markets may be suitable for parasitological examination provided they are alive or fresh (the gills should be red and without much mucus), and have not been kept in captivity for a long time or were not previously frozen.

If the number of hosts to be examined is too high for quick processing, the best option is to keep them alive. They can be maintained for some time in large tanks or wide plastic buckets with aerated water from the place of origin (or with dechlorinated water). However, the interval between the capture of hosts and their dissection should not be too long, because parasites may disappear from living hosts within a couple of days, mainly ectoparasites, but also intestinal helminths due to their starvation, stress and different water conditions. In addition, their community composition may change considerably, thus impeding reliable ecological study (changes in infection intensity and hence relative abundance, etc.).

Humane killing of fish

Before parasitological examination/dissection, the fish must be killed humanely in a dissecting dish with local water. Collecting and killing fish always need ethical approval and permits from a relevant authority. It is most important that researchers make sure that they follow the regulations and ethical procedures as prescribed by the country where the research is undertaken. For killing fish, pithing or stunning followed by interruption of the spinal cord should be used. Pithing (also spiking, coring, ikejime) is usually applied to smaller fish. A spike is quickly inserted into the brain of the fish (diagonally through the upper part of the eye or slightly behind and above the eye) and this is immediately followed by physical disruption of brain tissue by rotary movement of the spike. Bigger fish should first be stunned with a stroke on the head and then killed by interruption of the spinal cord immediately beyond the head using scissors or a sharp knife.

As an alternative to killing the fish, the fish can be sedated, anaesthetised or euthanised with chemicals such as tricaine (MS-222), clove oil, quinaldine sulfate, 2-phenoxyethanol, sodium bicarbonate and benzocaine. However, only MS-222, which does not seem to have an effect on parasites, is currently approved for use with fish that are destined for human consumption. More details about sedation, anaesthesia and euthanasia of fish are provided in the monograph by Ross and Ross (2008).

Host identification and labelling

Correct identification of the host is crucial for any parasitological survey or ecological study. Relevant data for the host such as its size (total and standard length), weight and sex should be recorded. Photographs of the host should be taken from a vertical position (not at an angle) with its snout directed to the left. The photos should include a ruler for size estimation and a unique host code (Fig. 3.2.1A). Morphological characters important for identification in individual fish groups such as details of the mouth, the fins and their rays, the number of scales on the lateral line, etc., should also be documented in these photographs. It is highly recommended to take samples of the host's tissues (around 5 mm in diameter, samples of muscles, fins or liver) and fix them with molecular grades 99% ethanol to enable later DNA-based identification. This is important especially in taxonomically complicated groups of fishes.

A unified system of hosts numbering with country codes and consecutive numbers (see Chapter 3.3.3) is strongly recommended because it avoids possible confusion if the same numbers are given to different fish hosts. Widely used abbreviations of fish names as codes may be helpful in some cases, but generally are not recommended because scientific names including genus of fish may change. In addition, this system of host coding is inapplicable when fish cannot be properly identified, which may happen with African fish, *e.g.*, cichlids or species of *Synodontis*.

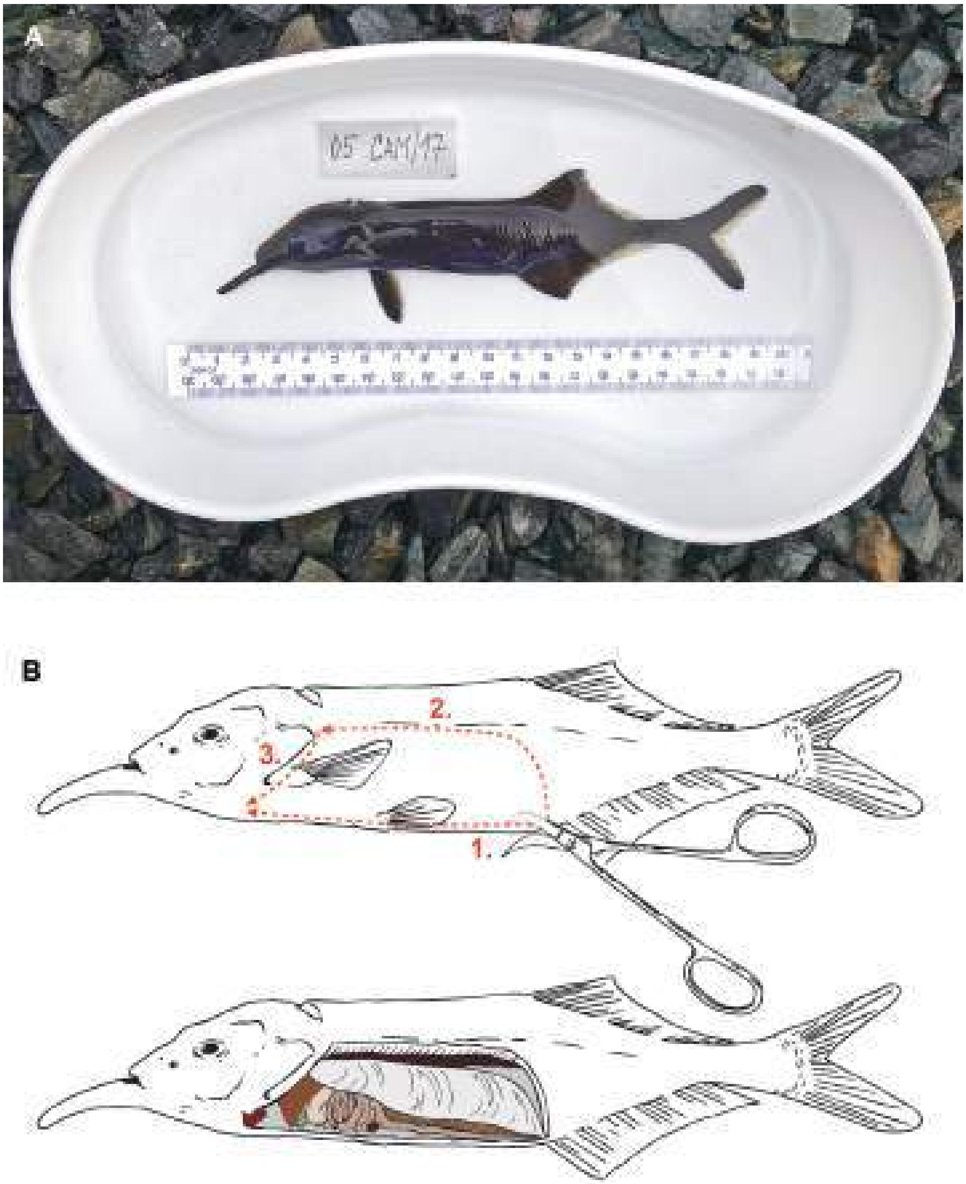

Fig. 3.2.1. A. Labelling fish hosts. Note that the fish snout is positioned to the left side and a ruler is added for estimation of fish size. The surface of the fish should be kept wet during any manipulation and handling of the fish; **B.** Illustration of how to open the body cavity of a fish to reveal the internal organs. (Photograph by E. Řehulková; illustration by M. Luo.)

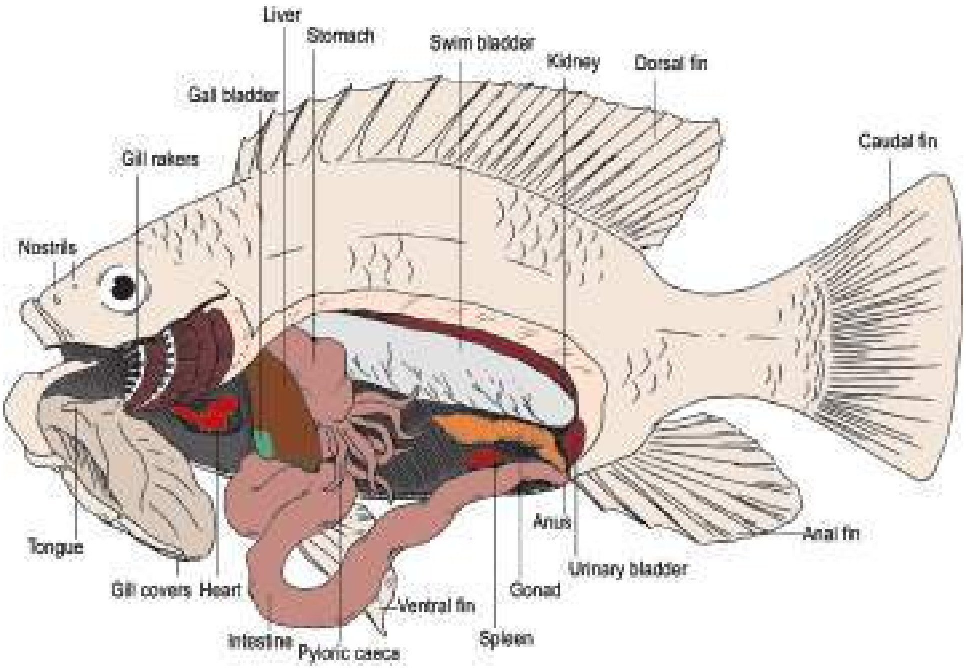

Fig. 3.2.2. External and internal anatomy of a bony fish. (Modified by M. Luo from Hile, R. 1960, U. S. Fish and Wildlife Service, Fishery Leaflet, no. 132, 6 pp.)

Information on the sampling date and locality (GPS coordinates, water temperature, etc.) should be recorded. The scientific name of the host, the infection site, the number of specimens found and fixed, the fixative used, the date of dissection and the name of the collector should be written in a field notebook for all parasites found. Recording of vernacular names (in addition to scientific ones, though) can be useful in interviewing fishermen or people in the market to find a particular species, to learn about its ecology, occurrence, etc. Thereafter, all the data can be transferred to spreadsheets, best as Excel files.

Fish anatomy and handling

Basic knowledge of fish anatomy is necessary before fish examination starts, especially the appearance and location of individual organs (Fig. 3.2.2). For the examination of head organs, the fish should be decapitated (see chapter 3.3.3). Access to the organs of the body cavity can be facilitated by removing one side of the body wall (Fig. 3.2.1B). The organs should be properly excised (avoid cutting them and releasing their contents) and should not be confused. For example, the excretory bladder can be difficult to find in some fish and the examination of kidneys requires scraping them from their location alongside the spinal cord. Superficial organs such as gills and fins, and scrapings from the surface should be placed in water. Internal organs and eyes should be treated in saline.

Reference

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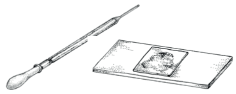


Chapter 3.3.

METHODS TO STUDY THE PRINCIPAL GROUPS OF FISH PARASITES

3.3.1. FISH-INFECTING EUKARYOTIC MICROORGANISMS (EMs)

Iva DYKOVÁ, Tomáš TYML & Astrid HOLZER

Introduction

EMs belong to several taxonomically divergent groups (Kabata 1985; Paperna 1991; Lom & Dyková 1992; Noga 2011; Adl *et al.* 2012). Their identification is traditionally carried out using a series of classical keys (see references to individual groups of parasites below) based upon the morphology of the whole organism, with confirmation or additional classification by DNA sequencing (predominantly 18S rDNA). Fresh smears are of special importance as many taxonomic features are not visible in fixed and stained EMs. However, tissue sections are important to determine the exact location of the parasite in the host and histopathological changes. Ideally, infected tissues are fixed for and studied by all possible methods. Often, light microscopical morphology allows assignment to a group or even genus but species identification requires molecular analyses or detailed ultrastructural studies (Aldrich & Todd 2012).

Groups of EMs

The following EMs are commonly found on freshwater fish:

- **Ciliates** (Alveolata, SAR) – ciliated protists with nuclear dimorphism (micro- and macronuclei). Motile. On external epithelia or inside the host, ranging from harmless to extremely pathogenic. See Figs 3.3.1.1A-I, 3.3.1.2A-G (for further reading, see Lynn 2008; Foissner 2014).
- **Blood flagellates** (Kinetoplastida, Excavata) – highly motile protists with one or two flagella, often forming an undulating membrane, characteristic kinetoplast (single large mitochondrion), associated with flagellar kinetosome. See Fig. 3.3.1.3A-C (Lom 1979; Davies 1995).
- **Amoeboid organisms** (Amoebozoa, Excavata, Opisthokonta, Rhizaria) – protists with amoeboid movement and pseudopodia. Most common are amphizoic amoebae (free living but able to colonise fish) on external epithelia, some other representatives in intestine or internal organs. See Fig. 3.3.1.3F,G (Page 1988; Dyková & Lom 2004; Dyková & Kostka 2013).
- **Coccidia** (Apicomplexa, SAR) – obligate intracellular protists, unsporulated/sporulated oocysts predominantly in enterocytes and faeces,

some other species in parenchymatous organs (*e.g.*, liver, spleen). See Fig. 3.3.1.4A-G (Dyková & Lom 1981, 1983).

- **Microsporidia** (Opisthokonta) – obligate intracellular protists with small, refractile spores with polar tube, which is used for injecting the sporoplasm (infective germ) into the host. Formation of large xenomas (infected and distended host cells) in different organs. See Fig. 3.3.1.5A-F. (Lom 2002; Lom & Dyková 2005).

- **Myxozoa** (Cnidaria) – multicellular (metazoan) parasites forming characteristic spores that contain 1-7 polar capsules, containing a polar filament for attachment to the host. Extremely diverse endoparasites. See Fig. 4.3.2A-M (Lom & Arthur 1989; Lom & Dyková 2006; Okamura *et al.* 2015).

Practical key for preliminary determination of fish-infecting EMs in fresh material

1 (2) Infection detectable as macroscopic whitish aggregations, from tiny dots to cyst-like structures of several mm or even cm in size; on the skin, gills, in or on the internal organs……………………………………………………3

2 (1) No macroscopic changes visible. EMs only detectable by light microscopy…………………………………………………………………………9

3 (4) Microorganisms visible as tiny dots on the body surface and gills. Under the microscope the dot proves to be large (up to 1 mm) slowly rotating cells, uniformly covered with synchronously beating cilia; next to large cells, there may be small ones of different sizes; their cytoplasm is full of granules and contains a large horseshoe-shaped macronucleus. (Fig. 3.3.1.1G-I)………………………………………………………………………………………...…***Ichthyophthirius multifiliis*** (Ciliata, Alveolata, SAR)

4 (3) Dot-, nodule-, or cyst-like structures composed of a mass of small, uniform, refractile bodies (spores or oocysts)……………………………….… 5

5 (6) The spores, typically 7-20 µm in size, most commonly have 2 (1-7) capsules containing a coiled filament, at one or both poles (Fig. 4.3.2A-M)…………………………………………………..…………**Myxozoa** (Cnidaria)

6 (5) Spores without polar capsules…………………………………………………7

7 (8) Spores very small, typically 3-10 µm in size, usually ovoid and often showing a prominent vacuole in the posterior part (Fig. 3.3.1.5A-F)………………………………………….…………..**Microsporidia** (Opisthokonta)

8 (7) Organisms are spherical or ellipsoidal bodies of about 10-20 µm in size, each containing four ellipsoidal bodies, each of which contains two slender cells. Whitish nodules within the body organs are not sharply delimited (Fig. 3.3.1.4A-G)………………………………………………………………………………………**coccidian oocysts** (Apicomplexa, Alveolata, SAR)

9 (10) EMs infecting the surface (skin, fins, nasal pits or gills)……………………11

10 (9) EMs infecting the intestine, other internal organs or blood…………………24

11 (12) Organisms that move…………………………………………………….. 13

12 (11) Sessile or motionless organisms attached to the surface…………………17

13 (14) EMs with flagella or cilia on the cell surface…………………………………15

14 (13) Cells with amoeboid movement and changes of body shape (Fig. 3.3.1.3F,G) ……………………………………………..……………………….. **Amoebae**

15 (16) Cells up to 15 µm in size, possessing two flagella, moving with jerky, creeping motion or swimming spirally forward……………….....……flagellates, *e.g.*, ***Cryptobia*** (Kinetoplastida, Excavata) and ***Ichthyobodo***

16 (15) Cells 20 µm and larger, either covered uniformly with cilia or with several ciliary belts or circular ciliary wreath; they move directly forward, glide over the surface, or roll on the spot (Fig. 3.3.1.1A,B)……………………………… ……………………………….**ciliates,** *e.g.*, scuticociliates (Alveolata, SAR)

17 (18) Pyriform or sac-like cells, attached to the skin or gills of fish………………19

18 (17) EMs attached to surface of host via stalks……………………………………21

19 (20) Transparent, attached pyriform cells not exceeding 15 µm in size …………………………………..…***Ichthyobodo*** (Kinetoplastida, Excavata)

20 (19) Pyriform or sac-like cells, 30-300 µm in size, their cytoplasm yellowish or greenish and containing many refractile granules ....**Dinoflagellata** (Alveolata, SAR)

21 (22) Cells 40-100 µm in size, with cytoplasm dark due to refractile granules, and with bundles of tubules with knob-like ends protruding from their surface……………………………..**suctorian ciliates** (Ciliata, Alveolata, SAR)

22 (21) Goblet-like or cylindrical cells about 40-90 µm in length, each with a wide free end encircled by wreaths of beating cilia; the cells may contract a little (Fig. 3.3.1.1E,F)…………..**sessiline peritriches** (Ciliata, Alveolata, SAR)

23 (24) EMs in internal organs, urinary tract or bile…………………………………25

24 (23) EMs in blood……………………………………………………………. 31

25 (26) Myxozoa (see 5; in any organ, urinary tract or bile), microsporidia (see 7; in any organ), coccidian oocysts (see 8; in intestine); or amoebae (see 14)

26 (25) EMs with surface showing flagella or cilia……………………………………27

27 (28) Cells up to 15 µm in size, with up to 8 flagella, moving about with a jerky motion or swimming directly forward........……………………**flagellates – Diplomonadida** (Excavata)

28 (27) Cells ciliated……………………………………………………………….. 29

29 (30) Spindle-shaped cells, of about 30-140 µm in size, uniformly covered with cilia, with both ends pointed and with sluggish movement…………………………………....**Protoopalina** (Stramenopiles, SAR)

30 (29) Ciliated cells of another shape, up to about 120 µm in length…………………………………………**other ciliates** (Alveolata, SAR)

31 (32) Motile EMs………………………………………………………………….33

32 (31) Non-motile EMs only visible in stained blood smears………………………35

33 (34) Slender cells, typically 10-15 µm long, moving with a wriggling or undulating motion, with 1 or 2 flagella (Fig. 3.3.1.3 A-C) …………………………………… **flagellates – *Trypanosoma* and *Trypanoplasma*** (Kinetoplastida, Excavata)

34 (33) Cells of about 3-15 µm in size, of amoeboid shape, displaying a twitching motion on the spot (Fig. 3.3.1.1E)…………………………………………… ………**developmental stages of some myxosporeans** (Myxozoa, Cnidaria)

35 EMs inside red blood cells (Fig. 3.3.1.3D)…………………………………… …………………………...**Haemogregarina** (Apicomplexa, Alveolata, SAR)

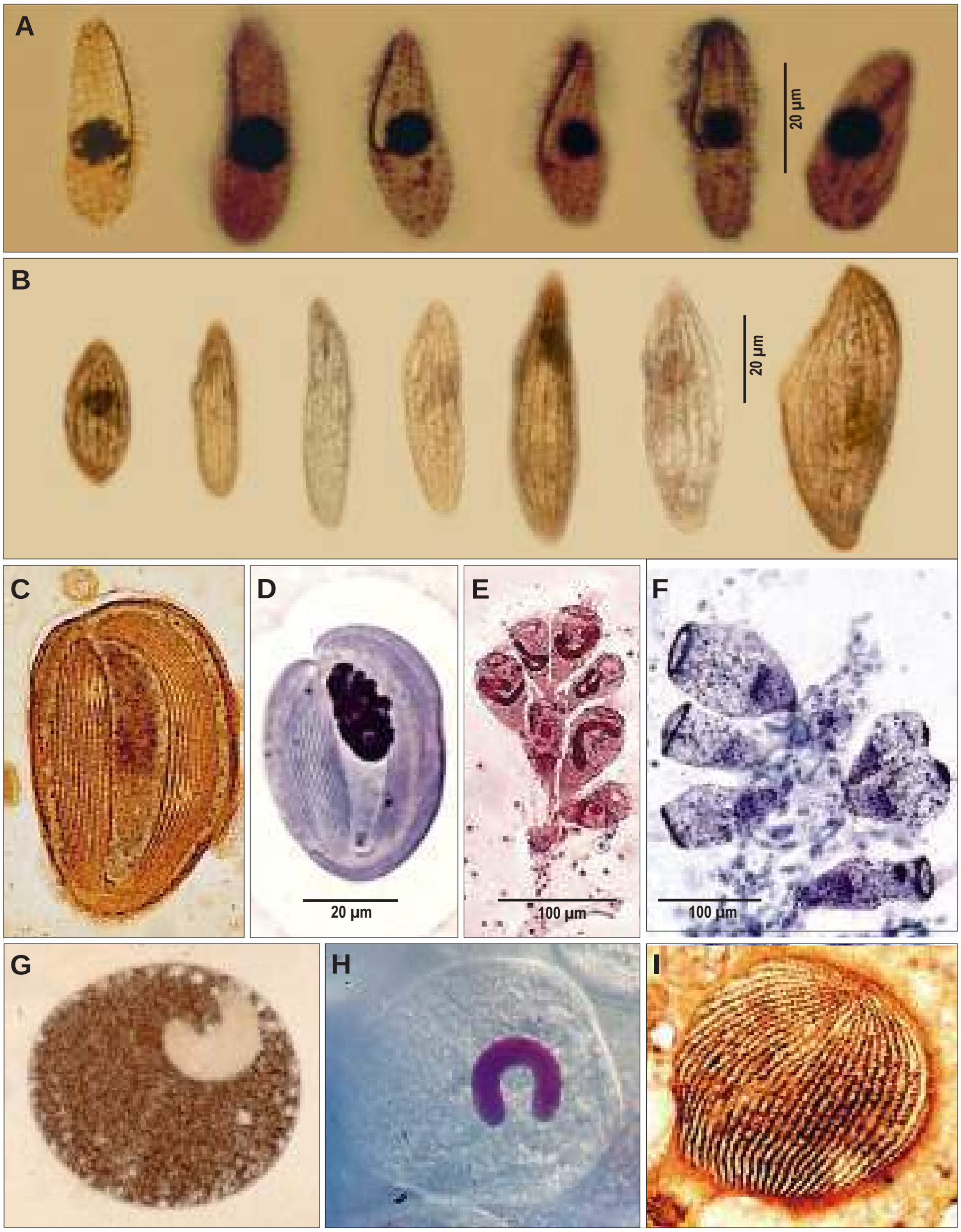
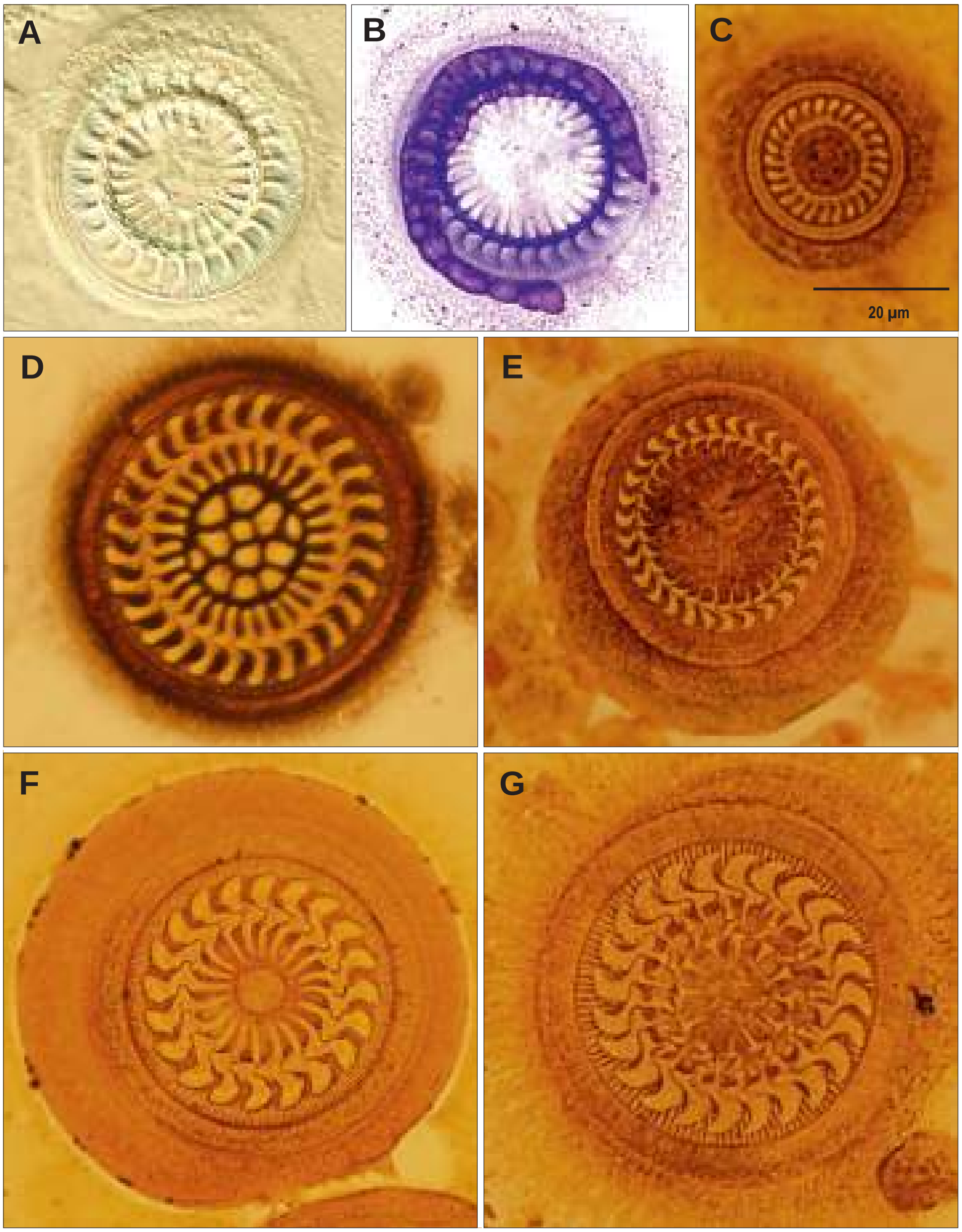


Fig. 3.3.1.1. Fish-infecting ciliates. **A, B.** Scuticociliates. **C, D.** *Chilodonella piscicola* (Zacharias, 1894). **E, F.** *Sessiline peritrichs*. **G-I.** *Ichthyophthirius multifiliis* Fouquet, 1876. Staining: protargol (A, D), 'dry' silver nitrate (B, C), Feulgen (nucleus stain; E,H), Klein's method (I). (All microphotographs by I. Dyková.)



3.3.1.2. Diagnostic features of trichodinid ciliates: skeletal parts of adhesive disc (AD) and shape of nucleus. **A.** AD seen in fresh (Nomarski differential interference contrast); **B.** Horse-shoe shaped macronucleus stained with haematoxylin; **C-G.** ADs stained with Klein's silver impregnation method: (C) *Trichodinella* sp., (D-F) ADs of various species of *Trichodina*. **G.** Dividing *Trichodina* with an outer well developed denticulate ring and a newly formed one indicated by concentrically arranged thorns. Scale bar C applies to all images.

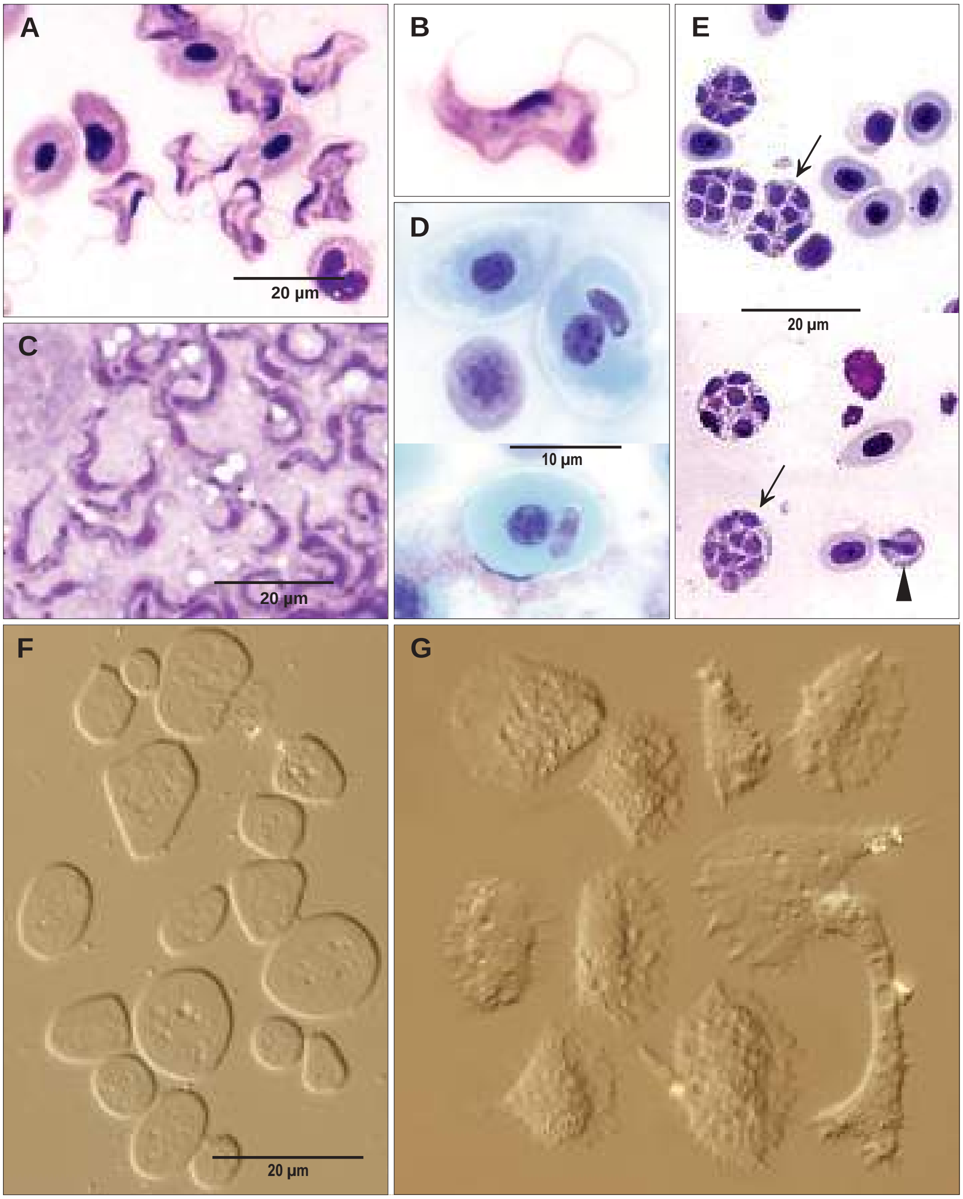


Fig. 3.3.1.3. A, B. *Trypanoplasma borelli* Laveran et Mesnil, 1901 stained with Giemsa. **C.** *Trypanosoma carassii* Mitrofanov, 1883 in Giemsa-stained blood smear. **D.** Intraerythrocytic stages of *Haemogregarina* sp. fixed and stained with Diff Quick. **E.** Proliferative stages of myxosporeans in Giemsa-stained blood smear. **F, G.** Trophozoites of an identical *Flabellula* strain seen under coverslip (**F**) and in hanging drop preparation (**G**). Scale bar F applies also to G.

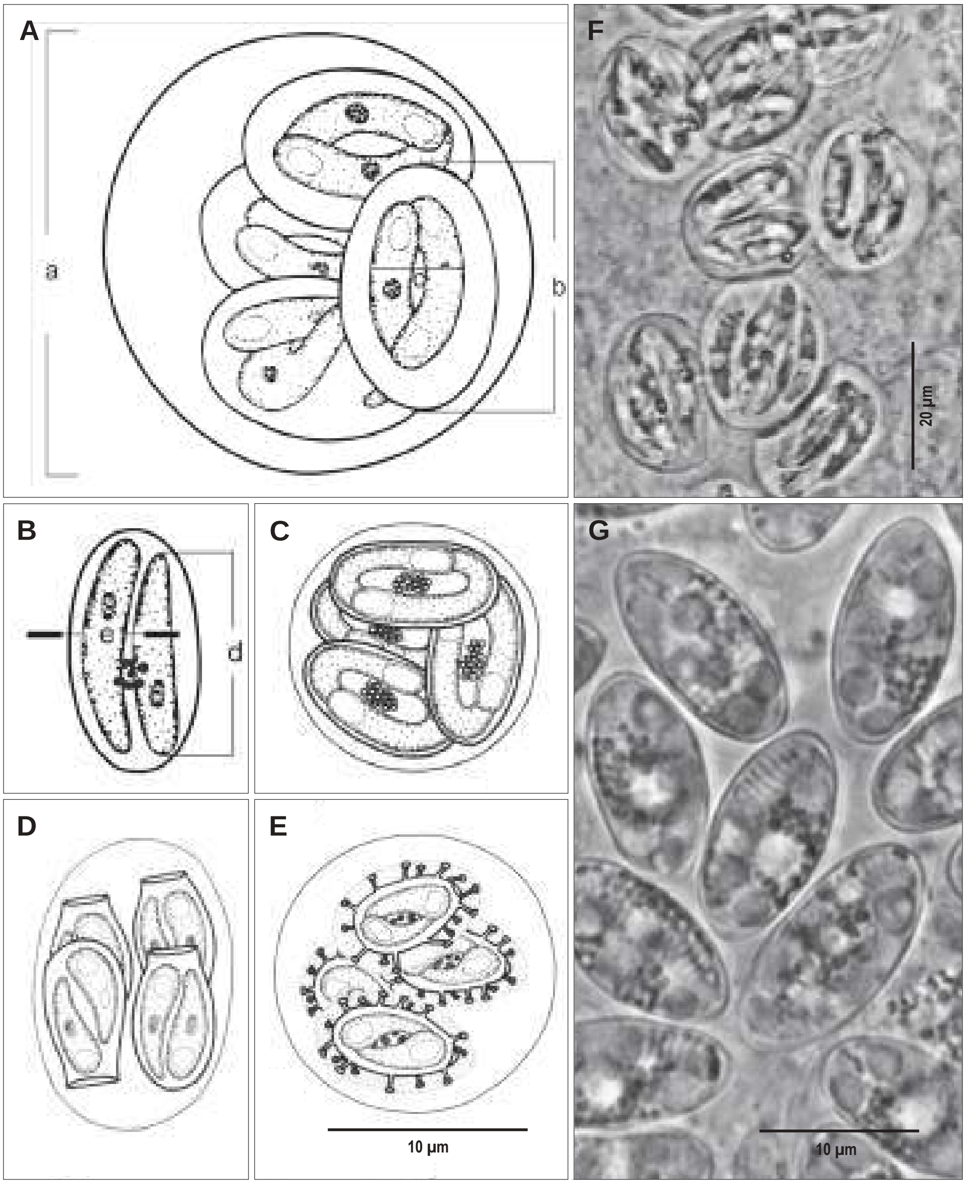


Fig. 3.3.1.4. Coccidia. Spherical oocysts (**A**), diameter (a), (length and width are measured in ellipsoidal oocysts), length and width of sporocyst (b and c, respectively). **B.** Sporozoite measurements (d, e). **C.** Oocyst of *Goussia carpelli* (Léger et Stankovich, 1921) contains sporocysts with residuum body. **D.** Oocyst of *Eimeria rutile* Dogiel et Bychowsky, 1938. **E.** Sporocyst walls of *G. deguisti* (Molnár et Fernando, 1974) bear projections (sporopodia). Scale bar E applies also to C and D. **F.** Sporocysts of *G. leucisci* (Shulman et Zaika, 1964). **G.** *G. subepithelialis* (Moroff et Fiebiger, 1905).

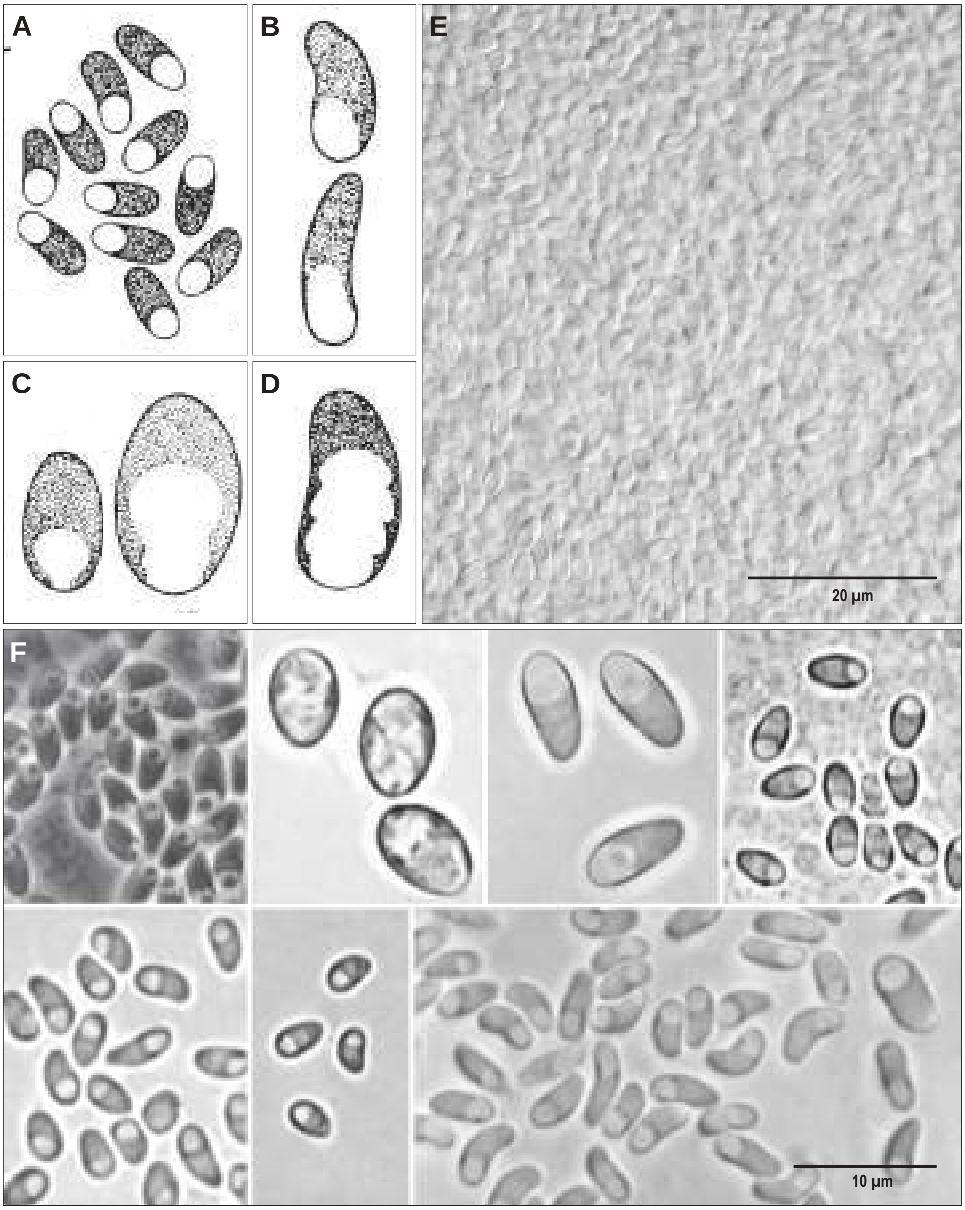


Fig. 3.3.1.5. Microsporidian spores observed in light microscope and documented in fresh state. In spores of fish-infecting species often contain conspicuous vacuole. **A-D.** Line drawings of *Microsporidium* sp., *Glugea* sp., *Pleistophora* sp. and *Heterosporis* sp., respectively. **E.** Fresh spores of *G. anomala* (Moniez, 1897). **F.** Photomicrographs exemplifying vacuoles seen in fresh smear and size differences of microsporidian spores belonging to various genera.

Screening procedure for EMs

Examination of the external surface: skin, fins, nasal pits and gills

Due to the loss of ectoparasites during capture and transport of live fish to the laboratory, the external examination is the most problematic part of the screening for the presence of EMs. Care has to be taken to preserve the outer surface of fish in an undisturbed condition.

PROCEDURE

1. Remove fish from the water using a small dip net and in accordance with relevant national legislation.
2. Scrape mucus from the skin and gills, using a coverslip, either while the fish is still alive or after pithing (anaesthesia is not recommended for the purpose of external examination as it may affect skin parasites).
3. Scrape the gills gently to prevent excess blood in the sample.
4. Spread mucus obtained on a slide and examine the fresh/wet mount for the presence of ectoparasites, at 40x to 1000x magnification (screen large area at low magnification first, then magnify; fix with methanol and store one smear for detailed observation if necessary).
5. Examine scrapings from both sides of the body, fin bases and the belly because the distribution of ectoparasites on the host may not be uniform.
6. Inspect also scrapings from the inner sides of the gill opercula as well as samples from the nasal pits, a special niche for some EMs.
7. Examine macroscopic, cyst-like structures or haemorrhagic areas following the detailed instructions given below.

Blood sampling, detection of blood parasites in fresh blood, blood smears

Venipuncture is the best method to withdraw blood from small fish (immediately after euthanasia). Blood is collected with a heparinised syringe inserted directly into the caudal vein in the area of the peduncule. Samples can also be used for blood chemistry, immunology, etc. Clotting time for fish blood is much shorter than for mammalian blood so always rinse syringes with heparin before use. Haemoflagellates and mobile proliferative blood stages of myxosporeans make themselves apparent by their vigorous movement in fresh blood mounts. Blood flagellate infections of extremely low intensity can be detected if several ml of blood are allowed to clot in a centrifuge tube placed overnight in a refrigerator or by using a haematocrit centrifuge. The following day, the flagellates can be found wriggling in the serum above the blood clot (tube)/compacted cells (haematocrit tube) while myxozoan blood stages occur intermixed with fish leukocytes (top layer after centrifugation). If necessary, the haematocrit tube is cut immediately above the compacted cell layer and the material transferred to a slide, using a micropipette.

PROCEDURE

1. Collect blood from the caudal vein with a heparinised syringe (the size of the needle should correlate with the size of the fish); the needle has to pass through the skin and muscles until it enters the vessel just below the spine.
2. Prepare several blood smears prior to examination of a drop of fresh blood under a coverslip (at a 400x magnification); stained blood smears are a prerequisite for detection of haemoflagellates, haemogregarines and proliferative stages of myxozoans.
3. Stain smears with Giemsa or Diff-Quik for subsequent detailed microscopical examination.

Examination of internal organs and muscle in fresh mounts

PROCEDURE

1. Inspect the internal organs after the body cavity has been opened by an incision made ventrally from the anal opening extending forward to beneath the heart, followed by the removal of one side of the body wall.
2. After macroscopic inspection of the organs, examine fresh mounts (see below).
3. Compress a piece of tissue about 1-2 mm in diameter between slide and coverslip; the coverslip is pressed after placing another slide on top to exert an even pressure over the whole coverslip, then it is removed.
4. Examine the samples under a compound microscope, first at a 100x magnification and then magnifying to 1000x; the number of samples examined from each organ depends on the size of the organ inspected.
5. During routine examination, include gill filaments, liver, spleen, kidney (*i.e.*, trunk kidney and head kidney), gonads, heart, swim bladder, the gall and urinary bladders and their contents (see point 6), muscle and brain.
6. Collect a sufficient quantity of bile and urine from the respective bladders (glass pipette) into a small vial and then examine several drops only for the presence of parasites (thus the bulk of material, if positive, is saved for further processing).
7. Cut open the digestive tract, separate its contents from the tissue and examine scrapings of the stomach, anterior, middle and posterior intestine (and from pyloric caeca, if present).
8. Examine also the rete mirabile at the back of the eyeball.

Examination of organs by histology

Simultaneously or prior to the examination of fresh mounts (squash and scrape preparations) tissue samples should be fixed to ensure adequate structural fixation for histological examination of fish organs infected with EMs (see also Chapter 4.4.). We recommend Davidson's as the best fixative for a well-defined cell architecture in histological sections. However, neutral buffered formalin is also good and, furthermore, allows for parasite DNA detection by *in situ* hybridisation. A guide to the identification of fish protozoan and metazoan parasites in stained tissue sections is available from: https://www.researchgate.net/publication/6911910_Guide_to_the_identification_of_fish_protozoan_and_metazoan_parasites_in_stained_tissue_sections.

Storage of fresh materials

Since the study of living EMs is time consuming and may interfere with the examination of fish brought into the laboratory for routine necropsy, it can be postponed in some cases by storing the fresh sample for later examination. Fresh mounts can be stored for some time in the refrigerator, either in a wet chamber (containers holding slides and some moist tissue) or if the edges of the coverslip are sealed to the slide with nail varnish. Myxosporean or microsporidian spores can be stored in distilled water at 4ºC for up to 12 months.

To prevent bacterial growth, the amount of host tissue debris in the sample should be kept to a minimum. ‘Clean’ spores from large ‘cysts’ can be recovered by puncturing cysts with a capillary tube. Small cysts can be separated from the surrounding tissues using dissecting needles or scissors. They may then be teased open and crushed, releasing the spores, which can then be stored for a limited period of time. As an emergency measure, myxosporean spores collected during long field trips can be studied in a preserved state, either fixed in 10% neutral buffered formalin, or in semipermanent mounts, *e.g.*, glycerol gelatine or glycerine ammonium-picrate.

Storage of material for extraction of DNA

The introduction of DNA-based taxonomy has advanced the identification of EMs as well as the understanding of their phylogenetic relationships. Molecular taxonomy and phylogeny have become an integral part of the EM research. The fixatives used for morphology/histology frequently damage DNA. The negative effects of formalin can be partly reduced if a neutral-buffered formalin solution is used instead of unbuffered or acidic formalin solutions; nevertheless, extraction of good quality DNA cannot be expected, especially after a long-term formalin fixation. Ethanol (95% or higher concentration) is routinely used for DNA preservation. The ratio of any fixative to sample should be at least 10 : 1 to ensure optimal fixation.

Identification of EMs detected and description of new species

The information collected from fresh mounts is of paramount importance; however, the organisms detected in fresh mounts can usually only be assigned to some of the major groups of fish-infecting EMs. The morphology of some of them allows assignment to a genus. Species identification and description of new species require detailed study using methods specific to each organism group, including molecular analyses (see Table 3.3.1.1 and references).

Table 3.3.1.1 Survey of basic methods used in the identification and description of species of EMs

Group	Principal method for morphology	Staining of smears/sections	Additional desirable techniques	DNA-based identification
Ciliates	fresh smears	Giemsa, silver nitrate, protargol	SEM[1], culturing	18S rDNA COI
Blood flagellates	stained slides	Giemsa, Diff-Quik	culturing	18S rDNA gGAPDH
Haemogregarines	stained slides	Giemsa, Diff-Quik	-	18S rDNA
Amoebae	hanging drop (live)	-	TEM[2], culturing	18S rDNA ITS
Coccidia	fresh smears	Giemsa, Diff-Quik, Gram	flotation method	18S rDNA
Microsporidia	fresh smears, TEM sections	PAS, Gram	-	18S rDNA ITS
Myxozoa	fresh smears	Giemsa, Diff-Quik, Gram	-	18S rDNA

[1] Scanning electron microscopy; [2] Transmission electron microscopy

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3.3.2. MYXOZOA

Pavla BARTOŠOVÁ-SOJKOVÁ & Ivan FIALA

Introduction

The present text is focused on myxozoan parasites of freshwater fishes, which can be found on the host surface as well as in internal organs. It is necessary to examine fresh fish individuals because, if dead, the host's tissues that potentially harbour parasites undergo fast degradation and parasites become unsuitable for subsequent studies, especially for transmission electron microscopy and histology. Data on the host species, sampling locality (if possible with GPS location), sampling and fish dissection date, the collector's name, fish condition (alive/dead), and fish total and standard length and weight should be recorded. It is highly recommended to transfer this information to spreadsheets, best as Excel files.

PROCEDURE

1. Assign a unique code to the fish individual examined and write it down in the dissection (field) notebook. If possible, it is highly recommended to perform fish dissection in teams of two, so that one person dissects the fish and fixes the material and the other examines the slides under the light microscope and takes pictures.
2. The dissection starts with the inspection of the fish surface; specifically, skin and gills are target infection sites for myxozoans. Evaluate both organs macroscopically; if you see cyst-like structures (usually whitish in colour), collect them carefully and squash them between a glass slide and coverslip. Scrape the fish skin mucus using a coverslip, flip it over onto a glass slide and prepare a squash slide by gently squashing the sample with another glass, thus equally distributing the pressure on the tissue sample.
3. Anaesthetise the fish using a clove oil solution or MS-222 for a few minutes before it is humanely euthanised (see Chapter 3.2).
4. Cut a small piece of gill filaments (maximum 0.5 cm large) and prepare a squash slide as described previously.
5. If interested in myxozoan extrasporogonic (blood) stages (*e.g.*, *Sphaerospora* spp.), take blood from the caudal vein using a heparinised syringe. Place the blood in a 1.5 ml microtube and collect it in a glass microhematocrit capillary tube which is then centrifuged in a microhematocrit centrifuge at 4000 RPM for 4 minutes. Break the capillary above the white blood cell (WBC) layer which may contain blood stages and collect this layer with a micropipette. Examine the fresh wet mount which is prepared by placing the WBC fraction and a small amount of fish serum onto a glass slide and covering it with a coverslip. For example, *Sphaerospora* blood stages can be distinguished from the host cells by their morphology (Lom & Dyková 1992) and by their specific twitching movement (Hartigan *et al.* 2016).

6. Open the fish by ventral incision starting from the anal opening following the midline of the body to the space beneath the heart. Make another incision from the starting point of the ventral incision close to the anus, and cut upwards to the top of the body cavity. Be careful not to damage the internal organs. Remove the lateral body wall on one side by cutting along the top of the body cavity.

7. Continue with the inspection of fish internal organs among which the kidney, gall bladder, muscles, liver and spleen are the most important locations for myxozoan parasites. The gall bladder must be carefully extracted from the rest of the organs and cut above a 1.5 ml microtube (or larger if necessary) to collect the clean bile, which is then transferred by pipetting a small drop onto a glass slide to be covered by a coverslip. Do not forget to clean the used dissecting tools between the dissection of different fish individuals or even between organs of a single fish individual by washing them under running tap water and subsequently in 70% ethanol or preferably in a 10% hydrogen peroxide solution, to avoid contamination.

8. Observe each sample under a light microscope at 400× magnification. If a parasite is detected, observe the same sample under a higher magnification using immersion oil and (if available) Nomarski differential interference contrast at 1,000× magnification.

9. Take microphotographs of all parasite developmental stages and spores observed immediately. Document at least 10 spores for each myxozoan species to enable later calculation of spore size variations and include a scale bar with each picture. Alternatively, continue with the fish dissection and photograph the parasites later. In the latter case, keep the slides with infected sample(s) in a wet chamber (a large Petri dish with wet tissues inside) in the fridge for a maximum of 24 hours to avoid drying out of the sample. In case the plasmodia or myxozoan blood stages move, a video can also be taken. Later on analyse the spore measurements (see Fig. 4.3.2 in Chapter 4.3) using ImageJ (Wayne Rasband, http://imagej.nih.gov/ij) or another software package.

10. Immediately after microscopic examination, fix a piece of infected tissue in cacodylate buffered 2.5% glutaraldehyde for further processing for transmission electron microscopy (TEM) (Glauert & Lewis 1998). The same fixative is applied for the preparation of samples for scanning electron microscopy (SEM) (Jirků & Bartošová-Sojková 2014); before fixing spores for SEM, separate them from the surrounding tissue on a dextran-polyethylene glycol gradient (Jirků & Bartošová-Sojková 2014), mix them with water or PBS (phosphate buffered saline) and place them on a grease-free poly-d-lysine coated coverslip. Glutaraldehyde-fixed samples can be stored for 24-48 hours in the fridge. Afterwards, the samples should be post-fixed in a 1% osmium tetroxide solution, followed by dehydration in a graded acetone series (in the case of TEM embedded in Spurr resin).

11. For histopathology, fix a sample (maximum size 1 × 1 cm) of the host organ in Davidson's fixative for 24 hours and transfer it to Davidson's stock solution, in which samples can be stored at room temperature for a longer

period (Heil 2009). Alternatively, 10% formalin can be used to fix samples for 24-48 hours, followed by replacement of the fixative by 70%, 80%, 90% and 96% ethanol (each concentration for one hour). Samples can be kept in 96% ethanol in the freezer for a longer period. Afterwards, the samples are embedded in paraffin and cut into slides that can later be stained by haematoxylin-eosin or Giemsa. When fixing the samples, do not forget to label each vial with a tissue sample on the outside and by inserting a label with the code of the host written in pencil.

12. Fix another small part of the infected organ in 96-99% ethanol or, for longer sample storage, in TNES urea buffers (Asahida *et al.* 1996) for subsequent DNA extraction and molecular characterisation of the parasite.

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3.3.3. ECTOPARASITIC HELMINTHS (MONOGENEA)

Eva ŘEHULKOVÁ

Introduction

Monogeneans are common, almost exclusively, ectoparasitic flatworms of freshwater, brackish water and marine fishes. Most monogeneans are tiny, which makes their sampling and further processing more difficult compared with larger-sized endohelminths such as most tapeworms, acanthocephalans or nematodes. Species identification of monogeneans may be difficult and its accuracy depends, to a large extent, on the quality of the material available. Therefore, adequate methods of sampling and processing monogeneans are required. If monogeneans are not collected and fixed correctly, it may affect the reliability of the morphometric data on taxonomically important structures.

Examination of fish for monogeneans

Fish should be examined immediately following their death while the monogeneans are still alive because living monogeneans are more easily detected by their movements. In addition, observations of living parasites may yield valuable information on internal structures (*e.g.*, digestive and excretory system) and the natural configuration of sclerotised hard parts. *Post-mortem* changes of monogeneans, which usually disintegrate quickly after they die, might make taxonomical evaluation of the specimens collected difficult or even impossible. The only disadvantage of collecting the living monogeneans is that they are sometimes harder to isolate because they are difficult to mount and orientate on a slide.

Fixed or preserved fish should be studied in a similar way as described below, but the quality of the specimens obtained is always much worse compared with fresh material; in some cases, a reliable identification of the worms cannot be made. It is important to point out that the surface of the fish should be kept wet during any manipulation and handling of the fish (taking photos, measurements, tissue samples, etc.), because drying up results in the damage or loss of monogeneans on the skin and fins. Therefore, the surface organs (skin, fins, nostrils, mouth and gill cavity) must be examined first after all the necessary data are recorded (see Chapter 3.3.1).

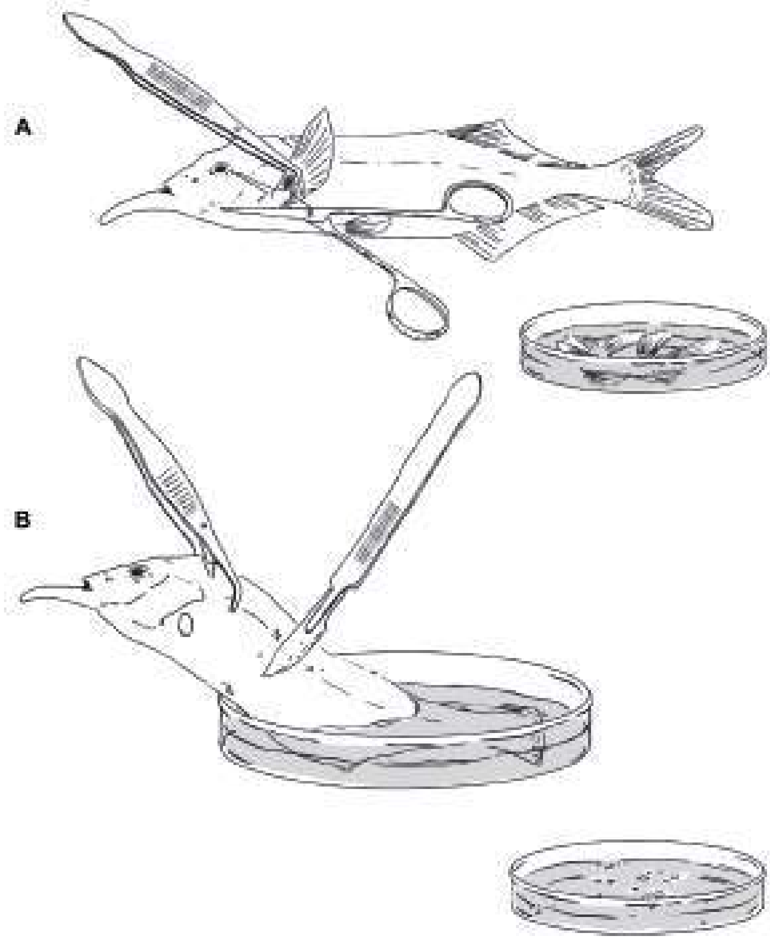

Fig. 3.3.3.1. Examination of fish for monogeneans. **A.** Cutting off fins; **B.** Scraping off mucus. (Illustration by M. Luo and E. Řehulková.)

PROCEDURE

1. Kill the fish using approved methods of euthanasia if it is not dead (*e.g.*, bought at the market or dead after capture).

2. Holding the fish with forceps, cut off the fins using scissors and place them in a Petri dish with water (preferably site water, *i.e.*, from the same source as the fish) (Fig. 3.3.3.1A).

3. Using a scalpel or slide, gently scrape mucus from the whole surface of the fish into a Petri dish with site water (Fig. 3.3.3.1B). If the fish is small (less than 10 cm), examine the whole fish directly under a dissecting microscope (magnification 20×). In this case, an upper illuminator for incident light viewing is required.

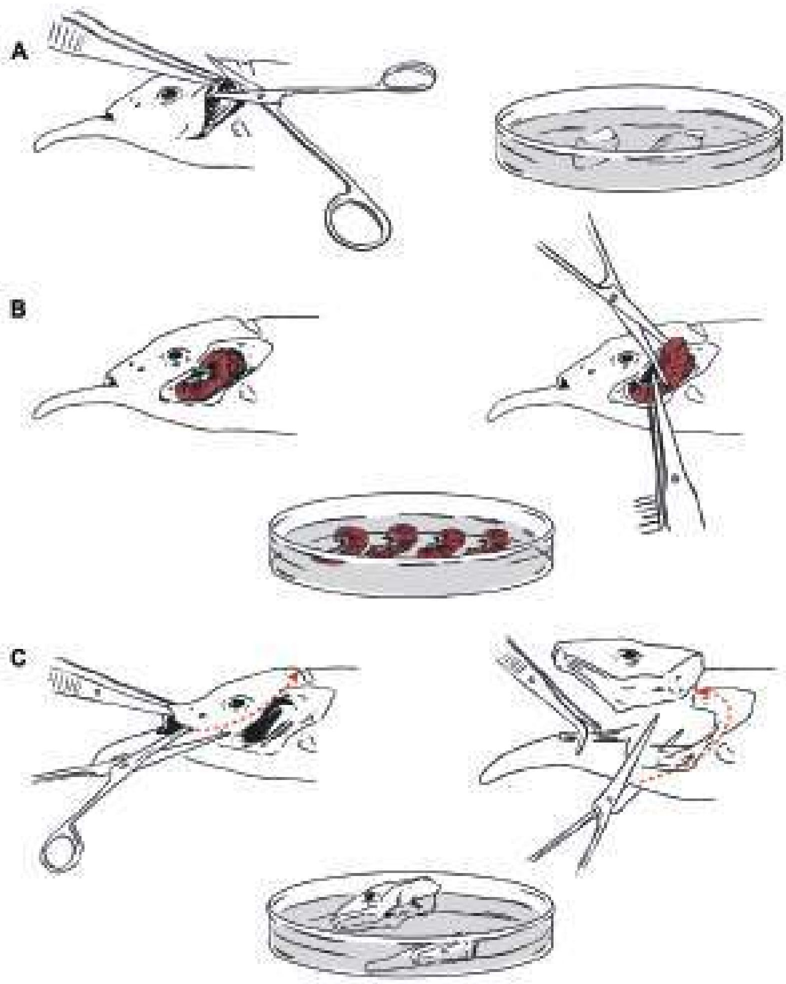

Fig. 3.3.3.2. Examination of fish for monogeneans. **A.** Removing of operculum; **B.** Extraction of gill arches; **C.** Separation of the upper part of the head from the lower part. (Illustration by M. Luo and E. Řehulková.)

4. Remove the operculum of the fish with scissors (Fig. 3.3.3.2A), cut off the gill arches (one by one) from the gill cavity and transfer them to a separate Petri dish with site water (Fig. 3.3.3.2B). If microhabitat preference is studied, each Petri dish should be labelled with the side/number of the gill arch (ideally 1 to 4 from external to internal).

5. Separate the upper part (nostrils, mouth) of the head from the lower part (mouth, pharynx, gill cavity); cut the mouth on both sides of the head towards the oesophagus (scissors following the dorsal side of the pharynx), decapitate the fish just behind the opercula, and place both parts directly in a separate dish with site water (Fig. 3.3.3.2C).

6. Carefully examine the mucus and all organs in Petri dishes with the aid of fine needles under a dissecting microscope at about 20× magnification. Check also the water in each Petri dish for detached monogeneans.
7. Carefully remove each worm from host tissues and place it in a drop of water on a slide, where it is can be fixed immediately (see below) or observed *in vivo* and photographed if the microscope is equipped with a digital camera.
8. After monogeneans from surface organs including gills are collected and fixed, the internal organs should also be examined for endoparasitic monogeneans (*e.g.*, species of *Enterogyrus* in the stomach of cichlids).

Fixation of monogeneans

A variety of methods are used to preserve monogeneans on slides, but some of them do not provide permanent preparations suitable for a deposition in museum collections as types (if a new species is described) or vouchers (faunal surveys and ecological studies). Basically, there are two methodological approaches to processing these parasites. The first one is focused on a study of sclerotised structures, the second one on observations of soft internal structures. To obtain the best results from both these approaches, two different preparation techniques should be used.

For a study of sclerotised structures the method of 'completely flattening' specimens is applied, where monogeneans are flattened under coverslip pressure until their body wall ruptures (see Fig. 3.3.3.3). Using this method, the vitelline follicles disintegrate after the rupture of the body and do not hamper observation of the male copulatory organ and vagina. If monogenean specimens are not sufficiently flattened, the shape of sclerotised structures may not be properly interpreted and their measurements tend to be shorter because of their twisted position. In contrast, coverslip pressure may affect the actual orientation of sclerotised structures with respect to the body axis. For that reason, the orientation of taxonomically important structures should be taken from non-flattened stained specimens.

Fixation to study sclerotised structures

To study the sclerotised structures of the haptor and the distal parts of the reproductive system (*i.e.*, male copulatory organ and vagina), the methods (formalin-glycerine fixative), proposed by R. Ergens in 1956 (in a Czech-written unpublished technical report) and later corroborated by Malmberg (1957; glycerine-ammonium picrate fixative or GAP), should be used. Formalin-glycerine fixative is prepared by mixing five parts of 4% formaldehyde solution and one part of glycerine/glycerol. GAP is prepared by mixing one part of saturated ammonium picrate solution and one part of glycerine.

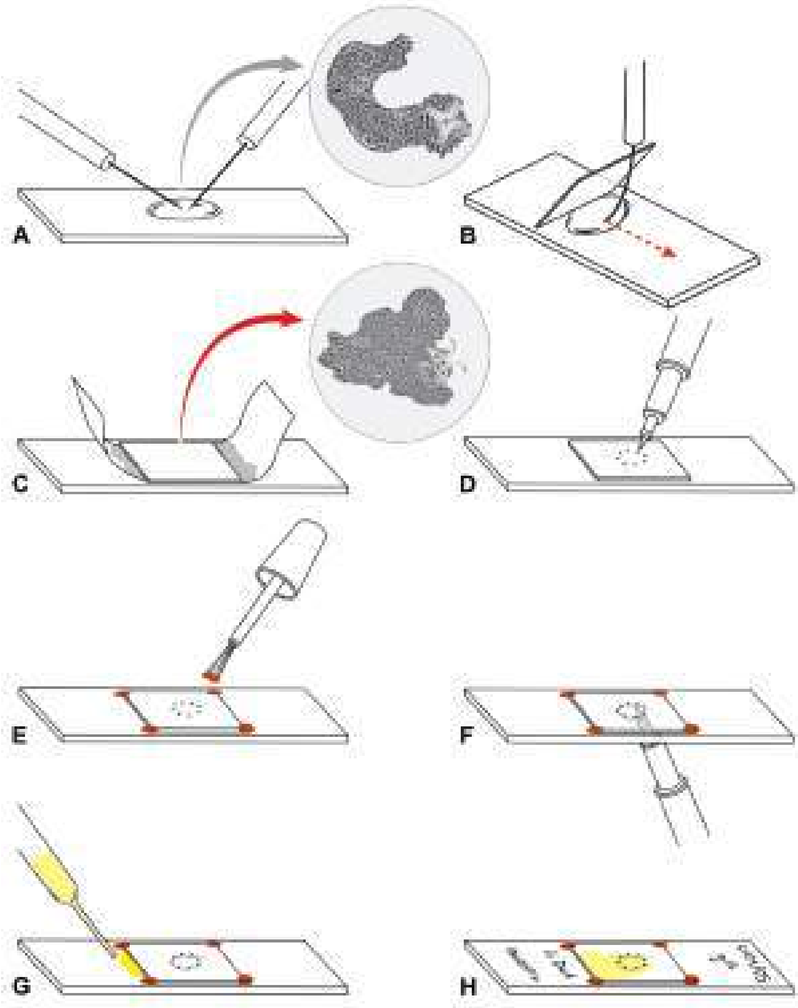

Fig. 3.3.3.3. Slide preparation of monogeneans mounted in GAP (glycerine-ammonium picrate) for subsequent morphological examination of the sclerotised structures. (Illustration by M. Luo and E. Řehulková.)

PROCEDURE (Fig. 3.3.3.3)

1. Place at maximum five clean worms, *i.e.*, worms without host tissue, mucus or any debris, which should be removed using fine needles, in a water drop on a slide using fine needles.
2. Lay a coverslip on the worm(s) while observing its/their position under a dissecting microscope to avoid the loss of the worm(s).
3. Remove excess water from under the coverslip by placing a piece of filter paper at the edge of the coverslip (best from both sides), thus further flattening the specimen(s) until the body wall ruptures.

4. Under the dissecting microscope, gently mark the position of the worm(s) by a dotted circle on the upper side of the coverslip.
5. Seal all four corners of the coverslip with Noyer's lacquer or nail varnish.
6. Trace the dotted circle (using an ethanol-resistant pen) around the worm on the reverse (lower) side of the slide.
7. Add a small drop of formalin-glycerol fixative (or GAP) on the edge of one side of the coverslip. Avoid adding a large volume of fixative as it can lift the coverslip and the flattened/ruptured worm(s) will disintegrate.
8. Label the slide (using an ethanol-resistant pen) with a field number (unique code) of the fish examined, date of collection, infection site (on the host), higher-rank taxon name (usually family) to which the specimen belongs (if known), or unique code of the worm (if part of it was fixed separately for subsequent DNA analysis).
9. Leave the slide on the table in a horizontal position to saturate the worm with formalin-glycerol (or GAP) for a couple of hours (overnight) before storage.
10. Seal the coverslip with enamel paint (nail varnish or Canada balsam) to prevent the mount from drying out.

Since both formalin-glycerine fixative and GAP are semi-permanent mediums, it is necessary for long-term storage, including deposition in museum collections, to remount these preparations using the method of Ergens (1969). This method produces permanent mounts, but some worms may be lost during the remounting procedure, especially if they are broken when the coverslip is detached. It is therefore strongly recommended to make drawings and take measurements from formalin-glycerine or GAP-fixed specimens before remounting them; another option is to take a photo as a photo-voucher.

Fixation to study the soft structures and further processing

To observe the soft internal structures, monogeneans should be relaxed during fixation and then stained with appropriate stains. Fixation with a fixative at ambient temperature (4% formalin or 70% ethanol) is useful only when monogeneans are being (moderately) flattened under a light coverslip pressure. This is best accomplished by placing the worms in a drop of water in a small Petri dish and covering them with a coverslip with a small weight on top (*e.g.*, a metal bolt or nut of approximately 2 g). It is important to note that too much pressure will distort the arrangement/size of the internal organs. Fixation with a hot fixative can avoid this disadvantage. Using hot 4% formalin is the best option (similarly as for trematodes, tapeworms and nematodes – see Chapter 3.3.4), because it penetrates fast into tissues and makes them well-preserved and more suitable for staining compared with samples fixed using hot water. If heating formalin is a practical problem, hot water can be used as described by Justine *et al.* (2012). Hot-water fixation makes it possible to use the worms for both morphological observation (after fixing with 4% formalin or 70% ethanol and subsequent staining) and DNA sequencing (fixed worms are immediately placed in molecular grade 96-99% ethanol).

Fixation of monogeneans for molecular studies

Even though the identification of monogeneans is based mainly on morphological characteristics, molecular data are important for taxonomic, phylogenetic and ecological studies. Therefore, it is strongly recommended always to fix some (parts of) specimens (see below) for genetic analyses (DNA sequencing). Simultaneous infections of fish with several, morphologically similar species represent a serious obstacle in molecular studies because the identity of sequenced worms cannot be ascertained without the availability of a corresponding morphological voucher, *i.e.*, hologenophore (see Pleijel *et al.* 2008 for terminology). In this case, the worms should be divided into three parts; the anterior body part comprising the male copulatory organ and the posterior part with the haptor are prepared for morphological observation as described above (*i.e.*, fixed with formalin-glycerine or GAP), whereas the middle part of the body is fixed in molecular-grade ethanol. However, this procedure is often inapplicable due to the small size of most monogeneans (*i.e.*, species of the families Dactylogyridae and Gyrodactylidae). Therefore, worms are cut just into two parts; that part which enables species-level identification (the posterior part with the haptor in gyrodactylids and diplozoids, the anterior part with the male copulatory organ in dactylogyrids) is fixed for morphological study and the remaining half of the body is fixed for molecular work.

It is important to note that only live or ethanol-fixed monogeneans are suitable for molecular studies. Formalin-fixed worms should not be used because their DNA is fragmented or considerably damaged. The procedure for dividing worms for both morphological and molecular studies is briefly described below.

PROCEDURE (Fig. 3.3.3.4)

1. Place the living or ethanol-preserved worm in a drop of water on a slide.
2. Under a dissecting microscope, divide the body of the worm into two parts using fine needles.
3. Transfer half of the body which does not contain the most important diagnostic structures, to an Eppendorf tube with molecular grades, *i.e.*, non-denaturated 96-99% ethanol and, if possible, store the sample in a refrigerator or freezer.
4. Fix the rest of the body in formalin-glycerine or GAP under coverslip pressure (if the worm is alive) or with Hoyer’s medium, as described below.
5. Use identical labelling for the tube and slide to match the morphological voucher (hologenophore and paragenophores) with the sample to be sequenced.
6. After morphological evaluation, deposit the hologenophore in an internationally accessible collection, ideally together with type (holotype, paratypes) or voucher specimens from the same host.

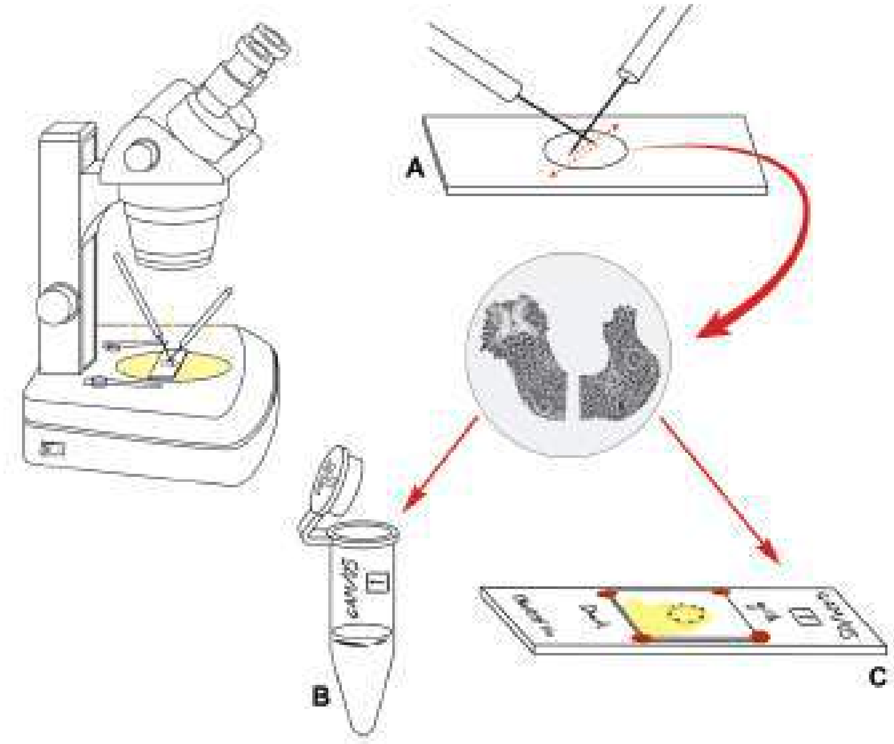

Fig. 3.3.3.4. Collection and identification of specimens for DNA analyses: specimen bisection using fine needles. (Illustration by M. Luo and E. Řehulková.)

Processing of fixed/preserved monogeneans

Study of sclerotised structures

Fixed/preserved monogeneans in vials are observed after being placed into a drop of water on a slide, removing excessive water and mounting them in Hoyer's medium. The slides should be kept in a horizontal position until the medium had solidified. As the worms are cleared rapidly, sclerotised structures and internal organs can be readily observed. Since this is a semi-permanent medium, it is best to ring the coverslip with enamel paint (or Canada balsam) after the medium has solidified. Hoyer's medium is prepared by mixing 30 g Arabic gum, 50 ml distilled water, 20 ml glycerol and 200 g chloral hydrate, followed by filtering the solution through 8-10 layers of cheesecloth or fine gauze before use (Ash & Orihel 1991).

Study of soft internal structures

To study soft parts, the monogeneans should be studied following staining. Different stains, mostly carmine-based, are used to visualise the internal structures and organs of monogeneans, *e.g.*, iron acetocarmine (Georgiev *et al.* 1986; see the procedure below), Schneider's acetocarmine, Mayer's acid carmalum, Gomori's trichrome, etc. (see also Humason 1979; Ash & Orihel 1991 for more details on several staining techniques). After staining, the worms are dehydrated in ascending series (increasing concentration) of ethanol, cleared (with clove oil or xylene), and finally mounted in Canada balsam as permanent preparations, which are suitable for long-term storage in museum collections.

PROCEDURE

1. Prior to staining, rinse the fixed worms in distilled water (30-60 min); worms fixed/preserved in 70% ethanol can be stained directly, without previous rinsing.
2. Transfer the worms to iron acetocarmine in a small Petri dish and keep them in the staining solution until they acquire a deep red colour (1-10 hours).
3. Rinse the worms by placing them into 70% ethanol.
4. Destain the worms in a weak solution of acid ethanol (1 ml or 4 drops of concentrated hydrochloric acid in 100 ml of 70% ethanol); leach the colour from the worms until they turn into a pale pink whereas the internal organs remain red-coloured. Destaining may take from several minutes to several hours, but it must be observed carefully to avoid excessive destaining. If too much stain is removed, rinse the specimens in 70% ethanol and return them to the stain (*i.e.*, start again with step 2), otherwise continue with step 5.
5. Rinse the worms by placing them into tap water until they turn into a deep red colour.
6. Dehydrate the worms through 70% (5 min), 96% (10 min) and 100% ethanol (5 min).
7. Clear the worms in clove oil (eugenol) for 5 min.
8. Mount the worms in Canada balsam as permanent slides.

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3.3.4. ENDOPARASITIC HELMINTHS

Tomáš SCHOLZ, Roman KUCHTA, Šárka MAŠOVÁ & Aneta KOSTADINOVA

Introduction

In the present text, general study methods are briefly described for different groups of endoparasitic helminths, *i.e.*, adults and larvae of flukes (Trematoda, *i.e.*, Aspidogastrea and Digenea), tapeworms (Cestoda), parasitic nematodes (Nematoda) and spiny- or thorny-headed worms (Acanthocephala). The main focus of this section is on the most important steps in searching for endohelminths and on their fixation and processing to ensure adequate quality of the material collected for subsequent evaluation. More detailed information can be found in specialised papers or books on individual groups of endoparasitic helminths.

Examination of fishes for endoparasitic helminths

Parasitological (helminthological) dissection is the basic method to obtain parasites. The extent of the examination depends on the objectives of a given study. The present text is focused on endoparasitic helminths and thus only the examination of internal organs will be described. As mentioned in the introduction to this methodological section (see 3.2), it is necessary to examine fresh hosts because worms, especially tapeworms and tiny trematodes, die quickly following the host's death. As a result, endoparasitic worms from long-time dead or frozen hosts are decomposed and unsuitable for subsequent studies including their reliable identification. Data on the host identity, site of infection, number of specimens found and fixed, fixative used, the date of dissection and the name of the collector should be written in a field notebook. It is highly recommended to record this information digitally on spreadsheets (such as Excel files) following fieldwork.

PROCEDURE (Fig. 3.3.4.1)

1. Take (a) photograph(s) of the host to be examined (the head of the fish should be on the left side) with its unique code (see below) and measurements (usually total and standard lengths). It is strongly recommended to excise a small piece of fish tissue (*e.g.*, muscle, fin – 'finclip', or liver) and fix it in molecular-grade ethanol to allow DNA-based identification of the host or other genetic work on the hosts, *e.g.*, barcoding, co-phylogenetic work, etc.
2. Place the complete digestive tract and other internal organs either in a suitable Petri dish or on a glass plate and add a small volume of saline (0.8-0.9% physiological solution, *i.e.*, 8-9 g of NaCl in 1 l of water). Under no circumstances should the organs dry out. In the tropics, you can add small pieces of ice to the Petri dish with the organs to cool the saline and thus slow down the decomposition of organs and parasites. Add labels with a unique host

code to every Petri dish with individual organs to avoid any subsequent confusion about the host. Check the body cavity of the fish (some helminth larvae can be present there).

3. Examine the surface of the internal organs (heart, liver, spleen, gall bladder, digestive tract, gonads, kidney, swim bladder) for parasites. Then separate the organs into Petri dishes with saline. Examine parenchymatous organs after teasing them apart into small pieces using scissors or forceps.

4. Open the intestine by cutting its wall longitudinally with small sharp scissors, preferably from the posterior part (anus).

5. Observe the content of the intestinal lumen and organs, preferably under a dissecting microscope or at least magnifying glass (good illumination is crucial for dissection; a good headlamp can be useful in the field when electricity is unavailable). The intestinal content should also be gently scraped with a scalpel and observed in a Petri dish with saline under a dissecting microscope.

6. Remove worms carefully (they are usually whitish or pale-white moving organisms) from the intestinal lumen and other organs with the aid of dissecting needles, a brush or a soft (entomological) tweezer or pipette. To detect (and reliably count) tiny worms, it is also possible to press the intestinal content and teased organs between two glass plates after their previous thorough observation.

7. Carefully place the worms in a small Petri dish with saline and wash them gently by flushing with saline using a fine pipette to take out mucus or host tissue. Use decantation (washing and sedimentation of the content in saline) for voluminous gut contents.

8. If time allows, observe the worms when alive (small endohelminths under light microscope), *i.e.*, their shape, movement, presence of structures not observable in fixed worms such as flame cells, *i.e.*, the terminal part of the osmoregulatory system of flatworms. Take a picture or video with a digital camera, make sketches of taxonomically important characteristics or record this information in your field notebook. Keep correct labelling and magnification to each document.

Fixation of endoparasitic helminths

The worms found should be fixed as soon as possible after their isolation and proper cleaning from host tissue or intestinal content. Adults of all endohelminths, except for acanthocephalans, are fixed in a similar way, whereas the fixation of their larvae (metacercariae and metacestodes) requires some modifications. Trematodes and tapeworms should never be flattened because fixation under pressure affects their shape and changes their size. Exceptions are a few special cases such rostellar hooks in larvae of gryporhynchid cestodes or the circumoral spines in trematode metacercariae, which are more visible after flattening. The present authors have found fixation in hot formalin the best choice for morphological studies of trematodes, tapeworms and nematodes including histological sections and scanning electron microscopy (SEM). If heating formalin is not possible, an acceptable alternative is to use hot saline or hot tap water to keep worms in natural shape, not deformed or contracted (see Justine *et al.* 2012). Specimens for morphological studies should afterwards be placed immediately into 4% formalin and samples for DNA sequencing into molecular-grade ethanol.

All fixed samples must be labelled with a unique number/code. We strongly recommend simple codes, preferably unique, consecutive numbers after the country code (*e.g.*, Sud304 for fish No. 304 examined in the Sudan), with small letters (a, b, c, etc.) as subcodes that enable you to distinguish individual samples found in the same host and avoid any confusion.

It is also recommended to use some specimens for both morphological (light microscopy, histology and SEM) observations and genotyping (DNA sequencing), *i.e.*, as hologenophores (see Pleijel *et al.* 2008 for terminology). If there are presumably conspecific worms in the same host, several specimens can be fixed for morphological observations (these are paragenophores), whereas the others should be fixed in 96-99% molecular-grade ethanol for molecular studies.

Fig. 3.3.4.1. Examination of endohelminths. **A.** *Malapterurus electricus* with host code and ruler; **B.** Internal organs removed from *Bagarius bagarius*, India; **C.** Opened intestine of freshly killed *Clarotes laticeps* with alive tapeworm *Proteocephalus sulcatus* (Klaptocz, 1906), Sudan; **D-F.** Examples of differences in fixation of tapeworm *Monticellia amazonica* de Chambrier et Vaucher, 1997 from *Calophysus macropterus*, Peru; **D.** Unnaturally contracted worms fixed after long time of relaxation; **E.** Unnaturally contracted worms fixed in 'cold' fixation (formalin solution); **F.** Worms properly fixed in hot fixative (hot water). (Photographs by R. Kuchta and T. Scholz).

Fixation for morphological, histological studies and SEM

PROCEDURE

1. Place clean worms in a beaker, Petri dish or plastic heat-resistant vial with a small volume of saline (just to avoid drying out).
2. Heat 4% formaldehyde solution (*i.e.*, mix 36-38% aqueous solution of formaldehyde in water or saline in ratio 1 : 9) or water/saline to its boiling point, with bubbles coming up from the bottom. The volume of the fixative should considerably exceed (at least 10 times) that of the saline in which worms are placed to ensure that the worms are heat-fixed.
3. Pour the hot fixative over the worms in saline. Well-fixed worms should be straight immediately after fixation, not contracted or deformed.
4. Once the fixative has cooled, transfer the fixed worms to a vial with the fixative at ambient temperature and add a label with the unique field number of the host (or write the worm's unique code with ethanol-resistant pen on the vial, not its lid).
5. After 1-2 weeks, transfer the worms to 70% ethanol before further processing (long-term storage of worms in formalin makes them hard and fragile).
6. Acanthocephalans are placed, after thorough cleaning (especially of the hooks on the proboscis), in a Petri dish with tap water and are maintained at 4°C for 1-15 hours until the proboscis is everted. Once the proboscis is everted, the worms are fixed with 70% ethanol (suitable for DNA sequencing, even though 96-99% molecular-grade ethanol is preferred) or 4% formalin. Some worms can be flattened between two glass plates and fixed in formalin.
7. Helminth larvae (except gryporhynchids) are usually difficult or impossible to identify based on their morphology and it is recommended to simply place them into a vial with molecular-grade 96-99% ethanol for subsequent molecular identification.

Fixation for molecular study (DNA sequencing)

PROCEDURE

1. Thoroughly rinse the worm (or its tissue sample) in saline to remove all possible traces of host tissue.
2. Place the worm directly in a vial with molecular grades 96-99% ethanol (*i.e.*, non-denaturated ethanol suitable for DNA sequencing). Check that the worm is actually in the vial, not still stuck on the dissecting tools.
3. Place a label with the unique host code (see above) in the vial and keep a morphological voucher (hologenophore or paragenophore – see above) of the same individual or species with the same unique code.
4. If possible, keep samples in ethanol in a refrigerator or freezer until further use.

Processing of fixed endoparasitic helminths

For morphological research, parasitic flatworms (trematodes and cestodes) and sometimes acanthocephalans are stained with carmine or haematoxylin to visualise their internal structures and organs. Following staining, they are dehydrated in an increasing ethanol series, cleared (best with eugenol – clove oil) and finally mounted as permanent preparations (whole mounts), preferably in Canada balsam, which has the best optical properties and does not crystallise as do some of the cheapest synthetic mounting media. These whole mounts are suitable for a deposition in museum collections as vouchers (or type specimens if a new species is described).

In contrast, parasitic nematodes cannot be stained and are observed as temporary mounts after clearing with glycerine (glycerol), which makes it possible to see their internal organs beneath the cuticle. Specimens stored in vials with 70% ethanol are placed on a slide and covered with a coverslip. Thereafter, a mixture of ascending concentration of glycerine: water (1 : 20, 1 : 10, 1 : 5, 1 : 2, pure glycerine) is added at each step after the water has evaporated on a histological heating plate to make clearing gentle. After examination, the nematodes are transferred back to vials with 70% ethanol for further storage.

Other specialised techniques such as gut washing, observation of the anterior end of nematodes (*en face* view), fixation of metacercariae, etc., are described in the specialised literature (Anderson 1958; Jones 1990; Moravec 1994, 2013; Scholz & Aguirre-Macedo 2000; Scholz *et al.* 2004; Cribb & Bray 2010; Oros *et al.* 2010; Justine *et al.* 2012 – see references below).

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3.3.5. PARASITIC CRUSTACEA

Martina DÁVIDOVÁ & Nico J SMIT

Introduction

Parasitic crustaceans (PCs) are very abundant, utilise an extraordinary broad range of hosts (fish primarily) and occupy a similarly broad range of microhabitats on their hosts. Most of these fish parasites are ectoparasites, being found all over the body surface of the host as well as in more sheltered microhabitats that are directly connected to the external environment, including the external nares (nostrils), eyes, oral and branchial cavities, gills and cloaca. A smaller number are mesoparasitic, living with their anterior (cephalothoracic) end embedded in host tissues and their posterior trunk protruding from the host's body surface.

The diversity of morphological forms of PCs (especially parasitic copepods), life cycles and host associations are enormous. Parasitic crustaceans of African fresh- and brackish water fishes belong to three distinct taxonomic groups: Copepoda, Branchiura and Isopoda (see the key in Chapter 4.9). Several genera of African parasitic copepods, branchiurans or isopods also occur outside Africa but other genera, such as *Dysphorus, Lernaeogiraffa* or *Chonopeltis*, are endemic to the African continent. Fryer (1968) recorded 48 species of copepods, 28 species of branchiurans and 3 species of isopods in Africa. Since then, more species have been described, providing better information about the richness and diversity of the parasitic crustaceans on this continent (see Chapter 4.9 for an updated list).

Screening for and collection of parasitic crustaceans on/in fish

The collection of fish hosts, host sedation and euthanasia and external surface examination follow the same protocol as that described in Chapter 3.3.3. Information on the host is vitally important in studies on PCs and every effort must be made not to mix host species following capture, because parasites may be transferred by accident while in the net (Boxshall *et al.* 2016). If it is necessary to transport the fish, they should be stored individually in plastic bags, because of the possibility of ectoparasites being dislodged. In the laboratory, the bag or container with fish should be screened for detached parasites. Although it is always best practice to collect material directly from freshly euthanised hosts, fish markets and fish donated by local fishermen can also be a good source of PCs. However, ectoparasites can be lost during the capture and handling of fish obtained from markets and local fishermen. These losses will affect results for prevalence and intensity of infection.

To find PCs, a macroscopic examination of the external body surface (including fins), mouth cavity, gills, opercula and nasal pits is necessary. It is also important to inspect for PCs first, before scraping for eukaryotic microorganisms (protists and myxozoans) as scraping may damage PCs, especially mesoparasites.

The fish should be examined externally in the following sequence (Kabata 1985):

1. Examine the skin, fins, eyes and nares.
2. Look for signs of external parasites, such as lesions, subcutaneous haemorrhages and missing scales (some copepods produce pouch-like invaginations by burrowing under scales along the side of the host, or into the walls of the alimentary canal, often in the rectal area – Boxshall *et al.* 2016).
3. Nares should be opened and examined as they are a favoured microhabitat for PCs, such as species of the Ergasilidae.
4. Open the mouth and examine the upper buccal cavity and space around the tongue and teeth.
5. Soaking the body in saline for 30 minutes can dislodge small ectoparasites copepods; the sediment from soaked fish should be then examined under a dissecting microscope.
6. Following macroscopical screening and removal of all PCs found, the gills should be screened again under a dissection microscope for small parasitic copepods, which are not always visible to the naked eye (see Fig. 3.3.3.2).
7. Entomological forceps, fine needles and Pasteur pipettes are required for the manipulation of PCs (*e.g.*, removing, cleaning from host tissue and transferring into fixative).
8. Ectoparasitic copepods are typically attached by clawed appendages which are of taxonomic importance. Therefore, care must be taken not to break off the claws when removing the parasite from its host.
9. Mesoparasitic PCs, *e.g.*, members of the Lernaeidae, typically have large metamorphosed females that live with their heads embedded in the muscles of their hosts, forming branching, anchor-like structures. According to Boxshall *et al.* (2016), the best way to extract a mesoparasite with its cephalic holdfast intact is to excise a large portion of the muscle tissue of the host, sufficiently large to enclose the full estimated extent of the holdfast, and place it in 50 ml of saturated potassium hydroxide. Cover it so that it cannot evaporate and leave for one or more days at room temperature, checking every day. The hydroxide digests host tissues surrounding the holdfast so that it can be teased away using dissecting needles. This process also digests the internal tissues of the copepod but the empty exoskeleton is intact and can be used for taxonomic study.

Fixation of parasitic crustaceans

Parasitic crustaceans can be fixed in different ways. For morphological studies, 4% formaldehyde is the most commonly used fixative. To avoid the negative effects of long-term preservation in formalin, specimens should be transferred in 70% ethanol for storage. Ethanol (70-95%) is also a good fixative for morphological evaluation and identification, and for molecular analyses. Davidson's AFA fixative (mixture of 10 ml of 37% formaldehyde, 50 ml of 95% ethanol, 5 ml of glacial acetic acid and 45 ml of distilled water) is recommended for histological sections.

Samples for molecular analysis should be frozen or fixed in 95% or absolute, molecular-grade ethanol. Such material should not be exposed to formalin, which contains methanol.

Processing of fixed material of parasitic crustaceans

The taxonomy of parasitic crustaceans is based mainly on external morphology; therefore, it is necessary to observe the details of the integument. Before identification, the material should be cleared in 90% lactic acid or glycerine to reduce visual interference from internal structures. Lactic acid gives excellent contrast. It is also possible to stain the integument. A good light stain for use with lactic acid is a few drops aqueous solution of lignin pink, added either to the undiluted acid or to 50% aqueous solution. Further information about stains for small crustaceans is available at http://invertebrates.si.edu/copepod/techniques.htm.

The choice of dissecting medium depends on the eventual mounting medium. It is often convenient to dissect the specimen in the eventual mounting medium rather than to attempt transfer of small parts. Dissection is accomplished most easily in glycerine or lactic acid using either fine entomological pins mounted in wooden holders, tungsten needles or a micro-scalpel. The most frequently used mounting media are glycerine, glycerine jelly or lactophenol. The latter medium was recommended by Huys and Boxshall (1991) for type specimens to be deposited in museum collections. Canada balsam can also be used, as for other groups of metazoan parasites. For mounting crustaceans in glycerine jelly or lactophenol, it is also possible to apply procedures used for parasitic nematodes (Ash & Orihel 1991; Moravec 2013). More information on mounting media and procedures for mounting PCs is available at http://invertebrates.si.edu/copepod/techniques.htm.

Preparation of glycerine jelly

Dissolve 10 g gelatine in 60 ml distilled water using moderate heat.

Add 70 ml glycerine and 0.5-1 ml of phenol to the gelatine solution and mix well.

Pour the liquefied glycerine jelly into glass bottles and store in a refrigerator.

Preparation of lactophenol

Mix 20 ml glycerine, 10 ml lactic acid, 10 ml phenol and 10 ml distilled water.

Store the solution in the dark at 2-25°C.

Identification of parasitic crustaceans

Parasitic crustaceans are usually identified using a stereomicroscope and/or a light microscope equipped with differential interference contrast. Taxonomy of PCs is based mainly on their external morphology. For their identification, features such as shape of the body and its individual parts, characteristics of segmentation, size of individual parts of the body, structure of head and thoracic limbs, characteristics of attachment apparatus, etc., are used (see Chapter 4.9).

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Chapter 3.4.

HISTOLOGICAL TECHNIQUES

Iva DYKOVÁ

Introduction

Histology can play an important part in research on fish parasites and parasitic diseases as long as its objectives and limitations are recognised. Histological examination is used mainly for diagnostic purposes, in screening for the presence of parasites in fish hosts and in evaluating their pathogenicity. However, it can also be used in research on specific structures of parasites, including diagnostic characteristics used to distinguish higher taxa, such as families of caryophyllidean and subfamilies of proteocephalid cestodes (see section 4.6).

The aim of histological techniques is to obtain thin sections of tissue samples of interest with as few artefacts as possible. To obtain satisfactory results, some degree of experience and insight is required. Histological techniques have been perfected for years to reach the point of an almost complete automation of sample-processing. This is important in big diagnostic and research centres, however, the prerequisites and individual steps of processing are the same whether automated or performed manually. These basic prerequisites and individual steps with their pitfalls are outlined below. More detailed instructions and recipes can be found in numerous histology manuals and websites, some of which are listed under the references.

Sampling for histology

Correct sampling for diagnostic purposes requires taking samples from freshly killed or moribund fish. To understand pathological processes caused by parasites, macroscopically visible lesions need to be sampled together with the surrounding, presumably intact, tissue. In order to avoid misinterpretation of artefacts, the fragile consistency of parenchymatous organs should be taken into account when tissue samples are extracted by forceps or other instruments. Tissue samples should be large enough to provide good quality information but small enough to be fixed (preserved) well. In the field, fish sometimes cannot be examined while fresh. Fixed tissue samples can be stored in 70% ethanol for a relatively long time (weeks) to be processed and examined later. Then an essential screening for the presence of parasitic infections can be based on histological sections (Figs 3.4.1-3.4.6).

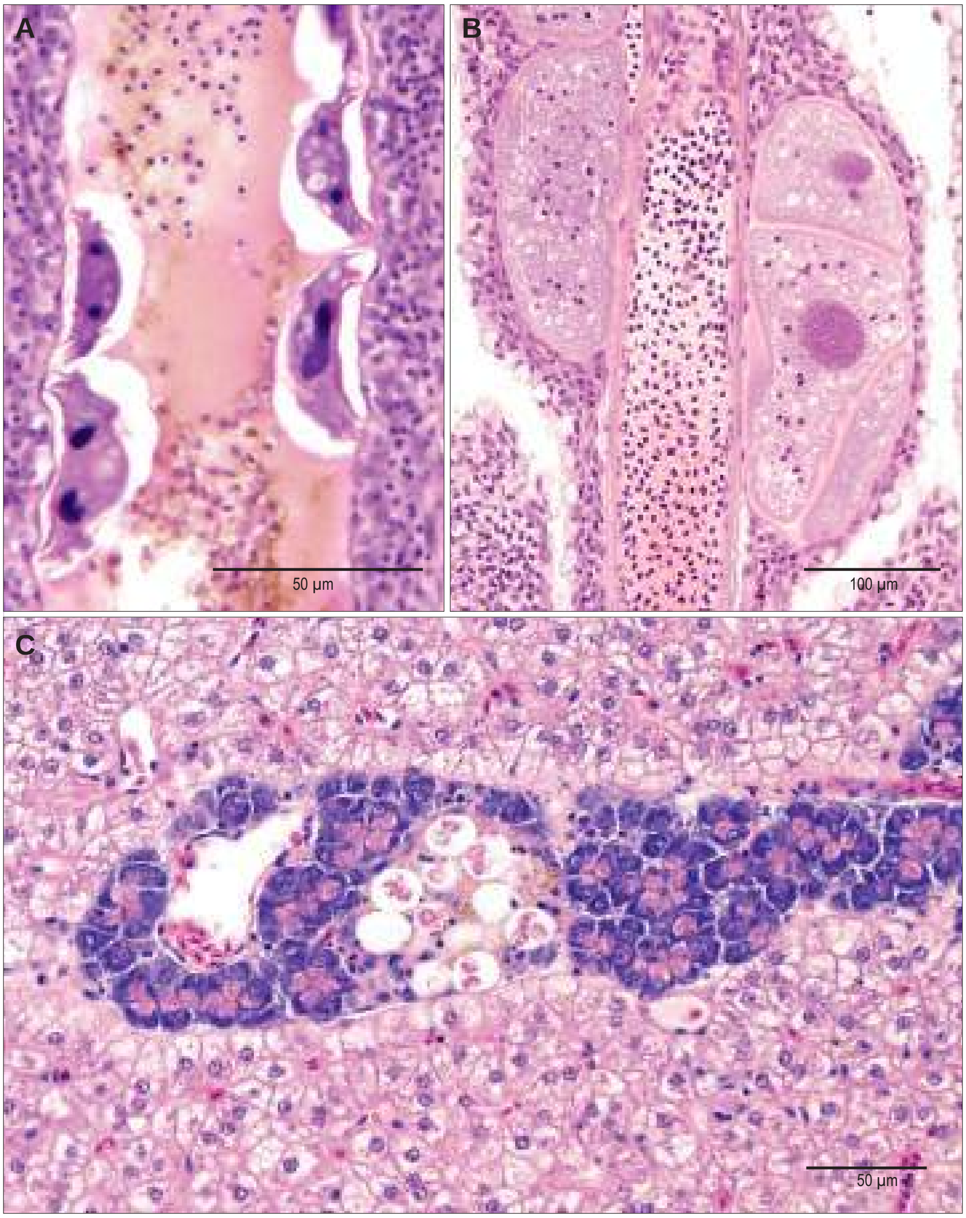

Fig. 3.4.1. A. Trichodinid ciliates seen on the surface of gill filaments of cichlid fish; **B.** Histophagous ciliate *Ichthyophthirius multifiliis* Fouquet, 1876 in the gill filament tissue of *Pseudotropheus* sp.; trophozoites with prominent macronuclei and host cells in the cytoplasm; **C.** Thin-walled oocysts of coccidia in hepatocytes surrounded by pancreatic tissue of *Haplochromis* sp. contain eosinophilic sporozoites. All haematoxylin & eosin. (All microphotographs by I. Dyková.)

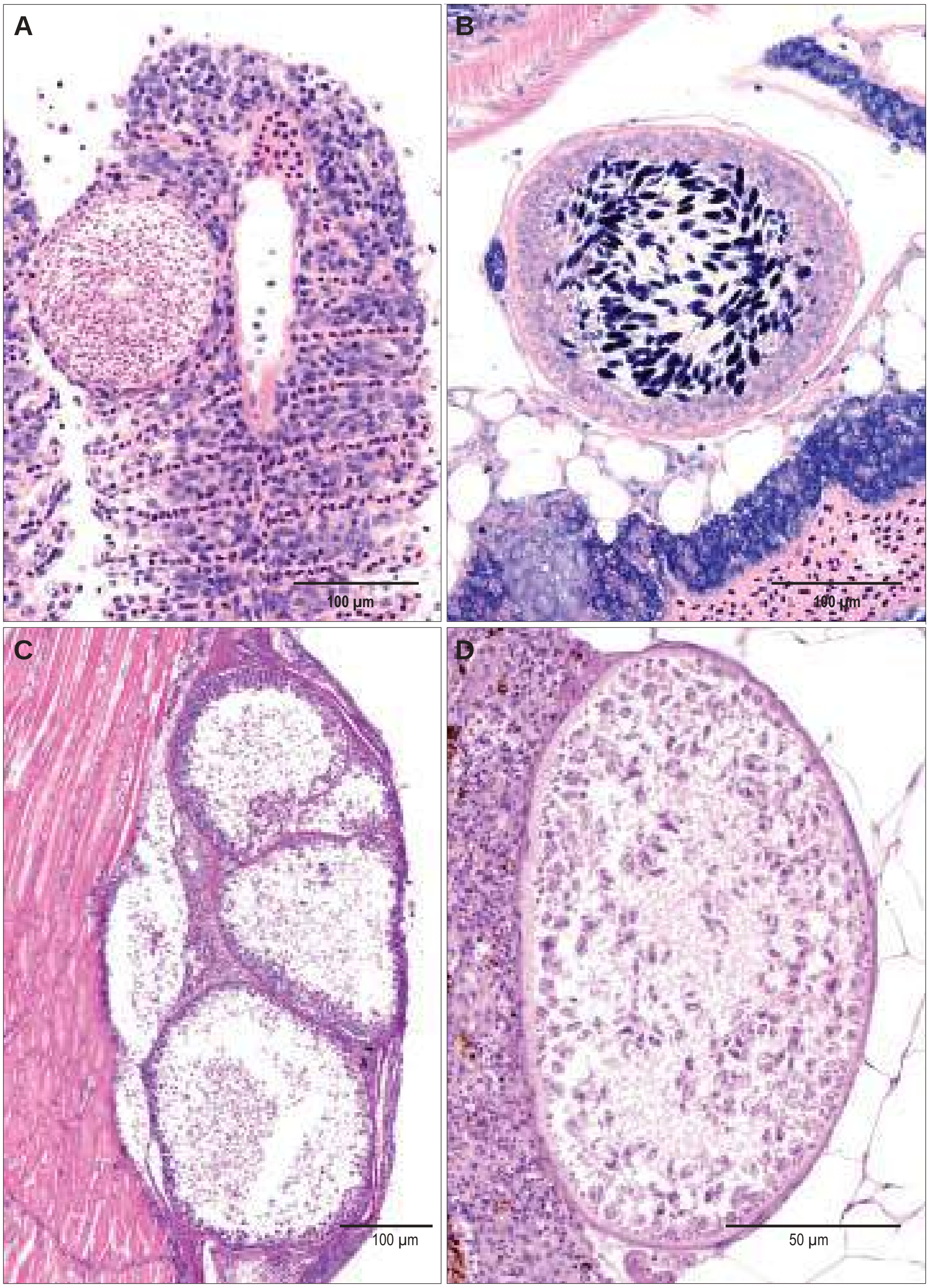

Fig. 3.4.2. A. Polysporic plasmodium of a myxosporean in the gill tissue of *Cichlasoma* sp. H & E; **B.** The plasmodial stage of a myxosporean species localised in the body cavity of *Leporinus* sp. contains intensely stained myxospores. Giemsa stain; **C.** Four myxosporean plasmodia localised subcutaneously in *Haplochromis* sp. H & E; **D.** Myxosporean plasmodium developing in the spleen of *Haplochromis* sp. H & E.

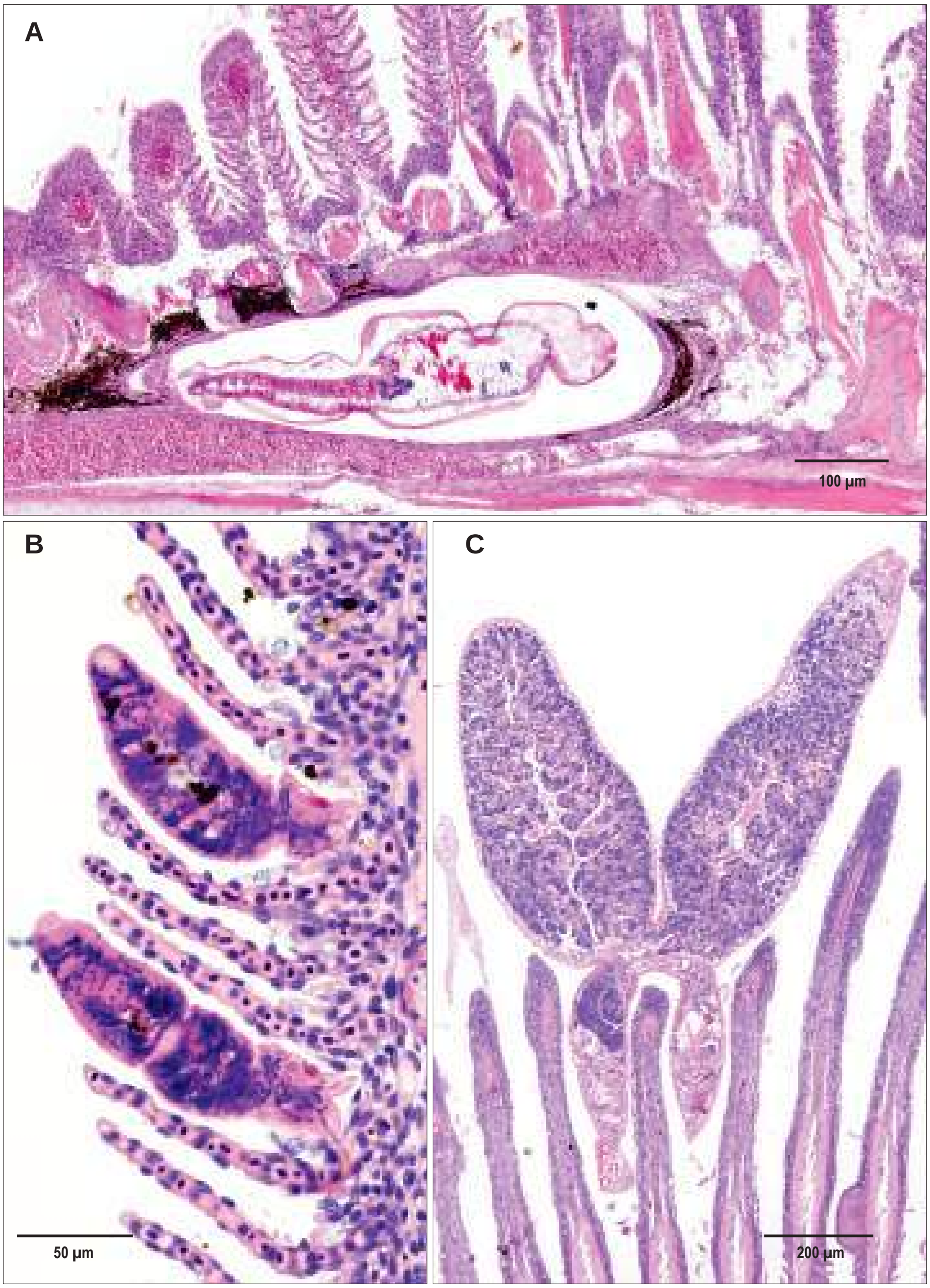

Fig. 3.4.3. A. Metacercarial stage of a digenean (Trematoda) in the gill arch of *Cichlasoma* sp.; **B.** Longitudinal sections of dactylogyrid monogeneans among secondary gill lamellae of *Haplochromis* sp., attached to epithelial tissue of gill filament; **C.** Complete section of a polyopisthocotylid monogenean *Diplozoon* sp. among gill filaments exemplifies the potential of histology in parasite identification. All H & E.

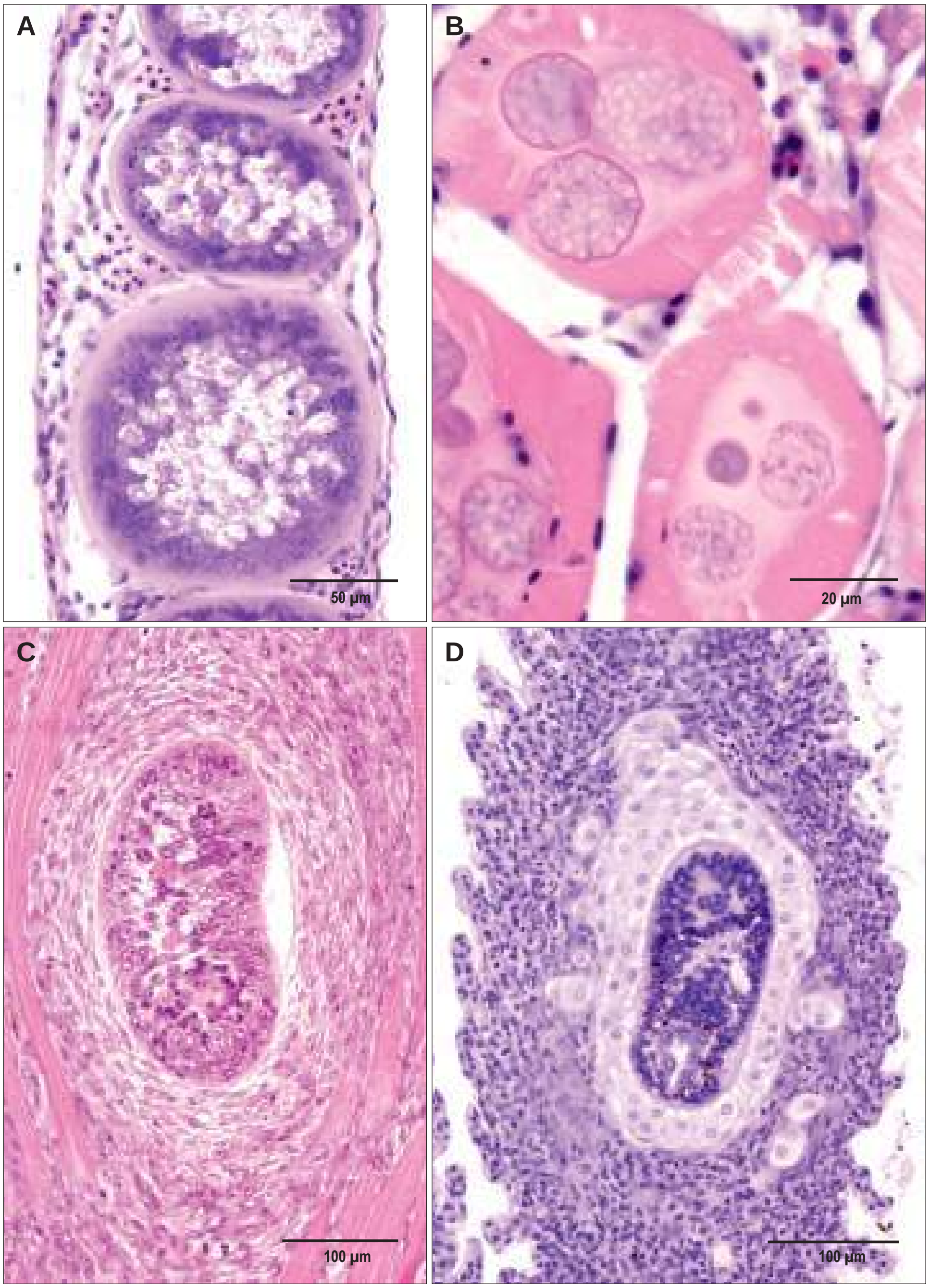

Fig. 3.4.4. A. Xenoma formations induced by microsporidia in connective tissue of *Nothobranchius* sp. Spores concentrated in the centre are almost indistinguishable whereas the whole formation can reach macroscopically visible size; **B.** Developmental stages of microsporidia in muscle fibres of *Paracheirodon innesi*; **C.** Metacercaria of a digenean trematode in muscle tissue of *Haplochromis* sp.; **D.** Metacercaria in cartilage of gill filament. All H & E.

Fig. 3.4.5. A. A sucker-bearing gryporhynchid cestode with hooks on the rostellum in the intestine of *Sandelia* sp.; **B.** Larval stage (plerocercus) of a gryporhynchid cestode in the liver of a cichlid fish; **C.** Cestode *Schyzocotyle acheilognathi* (Yamaguti, 1934) with a pair of dorsoventral grooves (bothria), part of neck and a short part of the strobila in the intestine of *Symphysodon* sp. The fish tissue is autolytic whereas the structures of cestode are well maintained. All H & E.

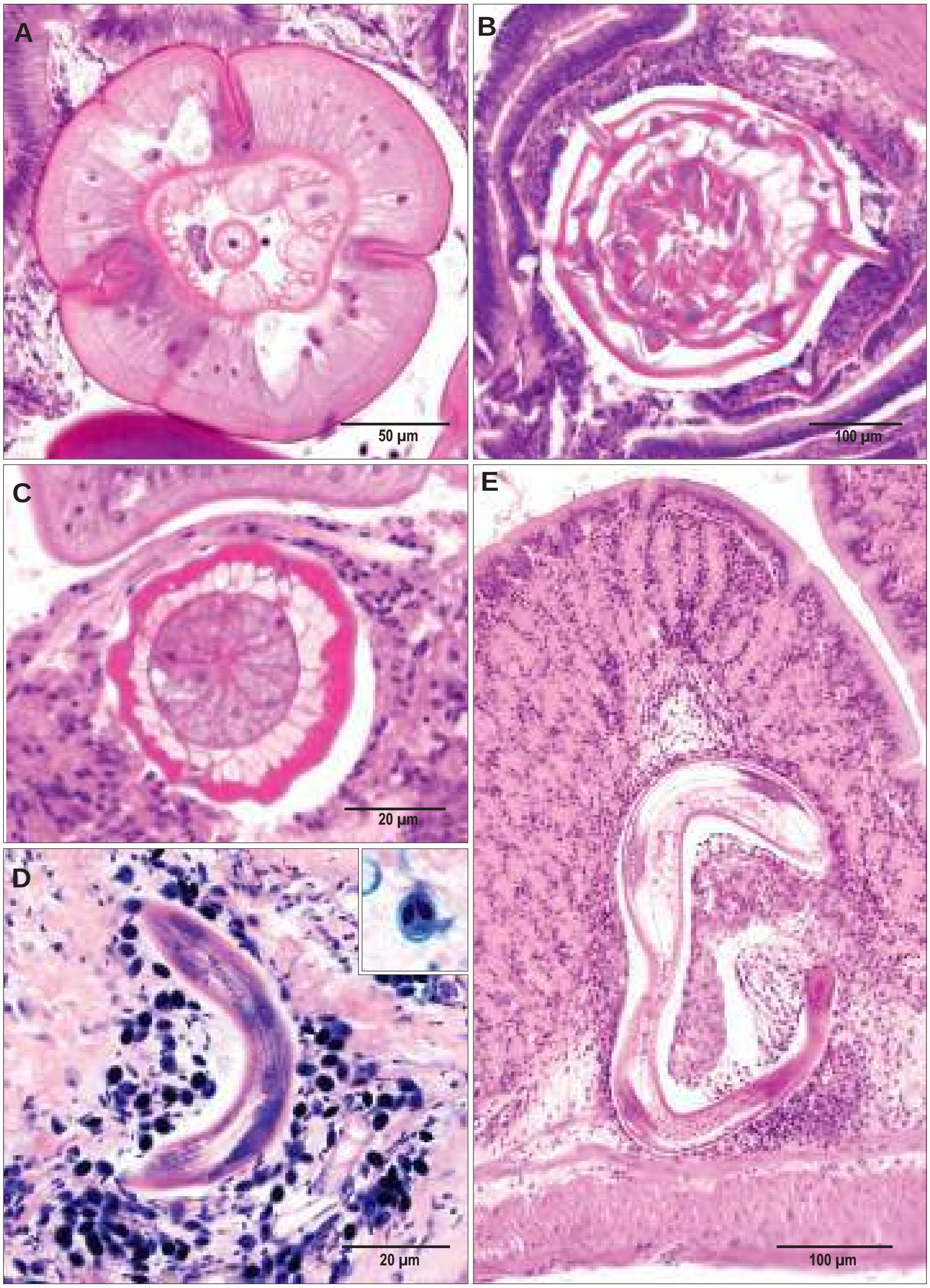

Fig. 3.4.6. A. Transverse section of the anterior part of an acanthocephalan. H & E; **B.** Transverse section of everted acanthocephalan proboscis armed with hooks. H & E; **C.** Transverse section through the oesophagus of a nematode. H & E; **D.** Larval stage of a nematode in host connective tissue. Also note the darkly stained myxospores (inset). Giemsa stain; **E.** Larval stage of a nematode in connective tissue of the stomach. H & E.

Fixation

The objectives of fixation are to preserve tissue samples, preventing autolysis and putrefaction. Fixation has to be adequate and complete. These requirements determine the type of fixative and the method of fixation applied. There are routinely used fixatives which fix tissue samples relatively slowly (*e.g.*, neutral buffered formalin solution), aggressive, rapidly penetrating fixatives (*e.g.*, mixtures of formol, acetic acid and ethanol), and fixatives which preserve specific cell components for specific staining procedures (*e.g.*, non-aqueous fixatives for glycogen). If a fixative causes tissue distortions and deformities, it is recommended to trim tissue samples before the next step (dehydration) starts.

Dehydration

To avoid excessive shrinkage of tissue samples, which ultimately causes difficulties in the evaluation of lesions, water should be eliminated from the samples almost completely by using ascending grades of ethanol before being transferred into an organic solvent. The best results are obtained with adequate concentrations of ethanol and adequate exposure times to ethanol and organic solvent.

Embedding

Embedding following dehydration consists of gradual impregnation of tissue samples with a firm medium (paraffin with a melting point of 56.6°C, mixtures of paraffin with other components, etc.) and blocking out in appropriate moulds. Of the considerations that should be kept in mind in the three-step impregnation procedure (three baths of paraffin), the most important ones are to follow the impregnation schedules given for each paraffin to eliminate remnants of organic solvents (xylene, toluene, etc.) completely. Too long exposures in paraffin and/ or the presence of solvents in the last paraffin bath impair the quality of blocks and sections. The paraffin-impregnated tissue should be oriented with the side of interest facing the bottom of the mould.

Sectioning

The essential equipment required for sectioning properly prepared tissue blocks includes a microtome adjusted for the type of knife used, a water bath, slides and a hot plate (or a safe place to dry sections). To ensure good results from the sectioning, several adjustments may prove necessary. Of those, the crucial one is an appropriate knife angle as specified by the manufacturer. Also important are the temperatures of the water bath, hot plate and oven (with paraffin, with its melting point 56.6°C, neither of these temperatures should exceed 45°C) and drying the sections completely.

Staining of histological sections

The staining procedure completes the preparation of tissue material for histological examination. It includes deparaffinisation of sections with xylene or another organic solvent, their hydration to water (through descending concentrations of ethanol), staining proper and counterstaining, followed by dehydration (through an ascending series of ethanol), clearing (with xylene) and mounting in a medium of choice. It is advisable to have a sufficient number of consecutive sections in order to avoid missing important details which might require special staining.

Haematoxylin and eosin are universally accepted basic dyes used to demonstrate tissue morphology. Haematoxylin stains the nuclear material whereas eosin stains the cell cytoplasm. Some special methods also deserve to be mentioned here, *e.g.*, the Periodic Acid Schiff reaction for demonstration of mucopolysacharides, Trichrom methods with various counterstaining agents for connective tissue, Van Gieson's method for collagen, Giemsa staining for protozoans, Gram's stain for Gram positive and Gram negative bacteria, Ziehl-Nielsen for acid alcohol fast organisms, Von Kóssas for demonstration of calcium salts, Perl's method for iron, Gomori for fungi, etc. There are many methods elaborated by specialists and many modifications of individual techniques. One can find dozens of recipes or modifications of basic staining methods but hardly ever a clear explanation of the chemical processes taking place during the staining. More than 50 staining procedures have been modified for microwave technology, saving time and liquids. For more detailed information, see list of references below.

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Chapter 3.5.

MOLECULAR TAXONOMY

Ivan FIALA & Jan BRABEC

Introduction

Deoxyribonucleic acid (DNA) sequences are a valuable source of information that stores the elementary instructions for how individual parts of an organism should be assembled and operate. DNA-encoded information can also be used to gain insights into the evolutionary history of an organism. Recovering this information has become an essential strategy to study and compare organisms. The field of downstream computational molecular evolution approaches has grown into a complex and rapidly evolving scientific discipline.

Analyses of DNA sequences have become an important part of various studies on the parasites of fish globally, including alpha taxonomy, diagnostics of disease agents, phylogeographical distribution and various studies on the biology of parasites including ecological, life cycle or host specificity-oriented surveys, to name a few. In strong contrast to that, the use of molecular data in studies on African fish parasites remains limited. Studies of Pouyaud *et al.* (2006), de Chambrier *et al.* (2008), Kuchta *et al.* (2012) and Přikrylová *et al.* (2013) are among the few available examples, where analyses of gene sequences assisted substantially in resolving the phylogenetic position of various fish helminths from the African continent. Schaeffner *et al.* (2011) and Chibwana *et al.* (2013) used molecular data to study phylogenetic relationships within individual genera of fish tapeworms and trematodes, respectively.

Co-phylogenetic analyses allowed Mendlová *et al.* (2012) and Vanhove *et al.* (2015) to propose speciation mechanisms in monogeneans infecting African cichlid fishes. Bouzid *et al.* (2013) studied genetic divergence within populations of the diphyllobothriidean cestode *Ligula intestinalis* (Linnaeus, 1758) using highly variable sequences of non-coding regions of DNA, whereas Kmentová *et al.* (2016) used sequence data from the nuclear ribosomal DNA region and the cytochrome *c* oxidase subunit I gene to look at, respectively, host range and intraspecific diversity in the dactylogyrid monogenean *Cichlidogyrus casuarinus* Pariselle, Muterezi Bukinga et Vanhove, 2015. Brabec *et al.* (2016) used next generation sequencing to study intraspecific differences within isolates of the invasive Asian fish tapeworm *Schyzocotyle acheilognathi* (Yamaguti, 1934) parasitising African fishes. Additionally, sequence data are frequently used in species descriptions to support the identification or discovery of parasite species, or to get an idea of their phylogenetic position.

Among the molecular markers most frequently used to study phylogenetic relationships and life history characteristics of fish parasites are ribosomal RNA (rRNA) encoding genetic loci, which include three rRNA encoding genes called 18S, 5.8S and 28S rDNA, according to their molecular weight, as well as the noncoding sequences of so-called internal transcribed spacers 1 and 2 (ITS-1, ITS-2) that are situated between 18S and 5.8S, and 5.8S and 28S rDNAs, respectively. Individual coding and noncoding regions from this genetic locus together form a unit called the nuclear rRNA operon, which is typically found in several hundreds to thousands of copies that are tandemly repeated one unit after another on certain chromosomes, depending on the parasite group. A wealth of information on molecular characteristics of rDNA has been summarised by Blair (2006) for parasitic flatworms and by Fiala *et al.* (2015) for myxozoan parasites.

Given the heterogenetic nature of individual parts of the nuclear rRNA operon, individual regions differ by their relative mutation rate and thus their speed of evolution. Therefore, they can be used across a range of taxonomic levels, spanning from populations of a single parasite species to orders and classes of parasites. Typically, the noncoding regions (such as ITS regions) are used at lower taxonomic levels (*i.e.*, populations and species), whereas the gene sequences (18S, 28S rDNA) are useful at higher levels, typically from genera to orders. However, for many parasitic groups, *e.g.*, Myxozoa or ciliates of the family Trichodinidae, 18S rDNA is a standard universal marker from species to order levels (Tang *et al.* 2013; Fiala *et al.* 2015). Combination of 18S and 28S rDNA or both noncoding and coding regions can be used in studying the phylogenetic relationships of parasites (*e.g.*, Bartošová *et al.* 2009; Přikrylová *et al.* 2017). Thanks to the presence of relatively conserved regions, rRNA loci can be characterised using a universal set of short strands of nucleotides called primers that are necessary to amplify a given region of DNA during polymerase chain reaction (PCR).

Sequences of mitochondrial protein-coding and rRNA-encoding genes (mitochondria are remnants of a primary endosymbiotic event and thus carry their own pair of rRNA genes originally belonging to an alpha proteobacterium) are further examples of commonly used molecular tools. Contrary to the nuclear rDNA, their overall speed of evolution tends to be higher (in some cases roughly comparable to ITS regions of the nuclear rRNA operon). This makes mitochondrial genes useful candidates for lower-level taxonomical studies. However, they may also be used on higher taxonomic ranks, when the protein-coding nucleotide sequences are translated into the corresponding sequence of amino acids. However, the increased mutation rate also means that universal primers are difficult to design. Moreover, flatworms substantially differ from other metazoans in amino acid content over cytochrome c oxidase I, *i.e.*, the sequence homology of flatworm and other metazoans' *cox*1 sequences is generally lower than sequence homology within metazoan *cox*1 (Vanhove *et al.* 2013) and researchers are thus left with no other option than to design a specific set of primers for their parasitic group of interests.

Recently, next generation sequencing techniques have been developed and gained popularity to bulk-characterise sequence data on large scales (*i.e.*, from thousands of loci to entire genomes) without previous knowledge of primer sequences. However, these sequencing approaches remain expensive and require the use of sophisticated technologies and highly trained laboratory staff and bioinformaticians, and are thus not suitable for routine taxonomy.

Fixation of fish parasites for molecular studies

The most critical step that allows successful isolation of DNA and generation of sequence data is quick and correct processing of the dissected parasite tissue and its immediate preservation in a suitable preservative. As a rule of thumb, parasites should be processed after their isolation from the host without any time delays, preferably immediately after the host's death. Extracted parasites (or infected tissues) should either be immediately preserved or kept in conditions that allow parasite survival (*i.e.*, in cool temperatures, appropriate pH and salt concentration). Before being completely submerged in the appropriate preservative (see Chapter 3.3), cells of parasitic protists or tissues of metazoan parasites have to be carefully cleaned of any remnants of the host cells and tissues, eliminating carry-over and subsequent simultaneous extraction of host DNA. Nearly absolute (96-99%) molecular-grade ethanol is used as a preservative of choice, notably in hot weather climate conditions.

DNA sequencing

Sequencing of selected molecular markers includes several steps (principally DNA isolation, PCR amplification and electrophoresis), which require adequate equipment and laboratory experience. A number of essential laboratory skills need to be acquired first to ensure successful and safe work in the laboratory. A good start for those not familiar with basic laboratory practice is to get familiar with individual chapters of the Current Protocols Essential Laboratory Techniques (http://onlinelibrary.wiley.com/book/10.1002/9780470089941) and preferably to obtain practical skills personally in an established laboratory under the supervision of a technician experienced in all relevant methods. Most of these complex issues can be eased through collaboration with an expert parasitologist with a publication record that includes the use of molecular taxonomy and phylogenetic approaches.

DNA isolation

The first step in the entire process of characterising novel sequences is isolation and purification of the DNA from the cells, the basal building blocks of any parasite's body. Within the cells, the DNA is located in membrane-bound organelles, where it is part of high-molecular complexes that consist of DNA itself together with a number of associated proteins. The goal of the DNA extraction step is to get the DNA out of these cells, into a protein- and other contaminant-free water solution called a buffer. It is essential to obtain well-purified DNA in this step, otherwise the following step (*i.e.*, PCR amplification) is likely to fail.

Generally, there are two basic, frequently used ways of isolating DNA. The first involves the use of a commercial DNA extraction kit (basically a box that includes all the chemicals and silica membrane spin columns necessary for DNA isolation, commercially available from many biotech companies). The second option is to go through a more traditional procedure called phenol-chloroform extraction. Both of these methods can vary slightly from one another according to the company that manufactures the kit and the authority that originally established the actual phenol-chloroform protocol. General principles and practical descriptions of sample protocols can be found in Dowhan (2012). As an oversimplification, both DNA extraction strategies are based on the digestion of the cells or tissue and the separation of the DNA from its associated proteins in a clean, water-based buffer.

Independent of the extraction protocol, all workflows start with transferring a certain volume of parasite cells or a small piece of tissue from the ethanol preservative into a new, clean 1.5 ml Eppendorf tube. The transferred material needs to be ground into as small pieces as possible (in the case of tissue) without risking the actual loss of the tissue, especially when you possess only small snippets of, for example a tapeworm strobila (often barely seen with the naked eye). In the case of larger parasites, you should cut up to 5 mm^3 of the tissues with clean, sterilised stainless steel dissecting scissors or a blade, and after a transfer into the new tube, cut it into as small pieces as possible immediately, before the ethanol evaporates and the tissue becomes solid, hard to cut and starts moving because of static electricity. Between processing individual specimens, make sure to thoroughly clean the forceps and scissors/blade used for transferring and cutting the tissue, to avoid cross-contamination of the sample by exogenous DNA that would be impossible to discover in later steps. A recommended method of cleaning is rubbing the forceps/scissors/blade well with a sterile piece of tissue soaked with absolute ethanol, and sterilising the steel tools over a laboratory burner. The tools should be cooled down before processing the next tissue sample.

PCR (polymerase chain reaction) amplification

Polymerase chain reaction is a method to amplify, starting from the solution of parasite DNA, a selected molecular marker that will be used, *e.g.*, to reconstruct the phylogeny of the studied parasite taxon. To amplify the chosen marker (*e.g.*, 18S rDNA), the following chemicals and tools are needed: Taq DNA Polymerase, Taq Reaction Buffer, dNTPs, forward and reverse primers, PCR-grade water and DNA template; thermocycler, pipets, tubes, tips and gloves. PCR is a routine method in many molecular laboratories and detailed protocols can be found elsewhere (*e.g.*, Sambrook *et al.* 1989). For successful amplification, good quality DNA and well-designed primers are crucial.

Electrophoresis

Agarose gel electrophoresis is the most effective way of separating DNA fragments of varying sizes ranging from 25 kb to 100 bp. The phosphate backbone of the DNA (and RNA) molecule is negatively charged. Therefore, DNA fragments will migrate to the positively charged anode when placed in an electric field. Since

DNA has a uniform mass/charge ratio, DNA molecules are separated by size within an agarose gel in a pattern such that the distance travelled is proportional to their molecular weight.

Sequencing

The PCR product of the proper size must be cleaned from unused nucleotides and primers. The product is directly sequenced using a DNA sequencer if available or making use of the services of commercial DNA sequencing companies. The result is a chromatogram file with the desired sequence of nucleotides of the genetic marker.

Phylogenetic analysis

BLAST analysis

The chromatogram sequence file should be checked to confirm that the sequences obtained actually belong to the studied organism. PCR may accidentally amplify the host gene instead of the desired gene of the parasite species. This usually happens when the primers are not specific enough for the studied parasitic group. The easiest way to clarify the sequence origin is to perform a BLAST (Basic Local Alignment Search Tool) search at the web page: https://blast.ncbi.nlm.nih.gov/Blast.cgi. BLAST search of the nucleotide sequences will find the closest match with the sequences stored in GenBank.

Aligning and tree reconstruction

The phylogenetic relationships of the studied organism can be revealed by aligning the sequence obtained with a selected number of sequences downloaded from GenBank at www.ncbi.nlm.nih.gov. Such a dataset of sequences is subjected to tree reconstruction analyses using several methods of choice. The most commonly used methods are maximum likelihood, maximum parsimony and Bayesian inference.

There are plenty of phylogenetic programs that can be used for phylogenetic analysis. One of the best programs including all methods is Geneious, which is a very user-friendly programme. A trial version can be downloaded and used for a limited time (https://www.geneious.com/). Another option is to use MEGA – a free programme with very good user-friendly interface (http://www.megasoftware.net). A very useful manual for beginners called “Introduction to Walk through MEGA” can be obtained at: http://www.megasoftware.net/web_help_7/hc_introduction_to_walk_through_mega.htm

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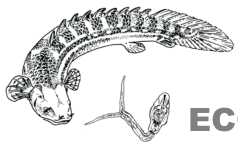

Chapter 3.6.

ECOLOGICAL STUDIES

Andrea VETEŠNÍKOVÁ ŠIMKOVÁ & Jiří JARKOVSKÝ

Introduction

Basic ecological studies in fish parasitology focus on parasite distribution in host populations, the structure of parasite communities and host-parasite interactions. The effects of some abiotic or biotic factors on parasite distribution (usually measured by parasite prevalence, abundance or aggregation) or parasite diversity have been analysed. The most commonly studied abiotic factors are season, time, water temperature, habitat types and environmental pollution. The most commonly studied biotic factors associated with hosts are species, body size, age, sex, food spectrum, stress, reproduction, immunity, or genetic diversity of hosts. The presence and abundance of a given parasite species in the parasite community may also be strongly affected by other parasite species currently occurring (or coexisting) in the parasite community.

General challenges in ecological studies on fish parasites

The protocol of an ecological study basically depends on the hypothesis to be tested, *i.e.*, predictions and objectives should be set prior to any ecological study. As parasite abundance and diversity can be affected by multiple abiotic and biotic factors, the ecological study should be designed to eliminate these effects. Before starting to investigate ecological patterns in fish parasites, the correct identification of host specimens should be confirmed by a specialist. In case of doubt over host identification or if hybridisation between phylogenetically related host species seems to play a role, molecular markers should also be applied to confirm morphology-based identification.

Sample size is important when investigating parasite diversity (for example, when studying the structure of parasite communities or in the case of comparative analyses of determinants of parasite diversity), investigating parasite distribution in host populations or delimiting host specificity of parasites. However, there are mathematical methods that allow correction for unequal sampling (rarefaction method or simulated random sampling of given sample size). Another confounding effect may be the host body size as parasite diversity (and parasite abundance) generally increases with increased host body size due to allometric relationships. Larger hosts represent a larger and more stable habitat for parasite colonisation. Therefore, when comparing the parasite communities of a given host species between different sites, hosts of similar body size should be selected.

For parasites with a complex life cycle, the presence and abundance of intermediate hosts is another biotic factor influencing the composition of parasite communities and should be taken into account even if the study includes sites with equal sample size and fish hosts of similar body size. Furthermore, parasite diversity and parasite abundance may vary in time and space. In addition, the biotic variables linked to hosts also exhibit temporal and spatial variability (*e.g.*, seasonal changes in water temperature induce changes in fish immunity which affect the level of parasite infection). Therefore, when investigating spatial variability in parasite diversity, the ecological study should be performed under similar environmental conditions (*e.g.*, when comparing the parasite diversity of a given host species among different sites, the fish from all sites should be sampled in the same season, with similar water temperature or water flow).

A very important part of ecological studies on parasite diversity is fish storage following sampling and the time between the collection and processing of fish specimens (*i.e.*, fish dissection and parasite collection). Fish should be quickly transported to the laboratory and placed into containers with the original water and aeration. All fish should be dissected and parasites should be collected and fixed within 48 hours after capture. Alternatively, fish may be frozen and dissected later, but in that case most parasites found are useless for a detailed morphological study. In addition, parasites cannot be detected based on their movement. Finally, host phylogenetic relationships should also be considered. Two congeneric hosts may share parasite species due to common ancestry.

Fish in the life cycle of parasites

Parasites exhibit direct or complex life cycles. In the case of a direct life cycle, parasites require only one host species to complete their ontogenetic development. All monogeneans, some nematodes and most arthropods have a direct life cycle. Parasites with a complex (or indirect) life cycle have one or more obligatory intermediate host species in different stages of their life cycle in which the parasites undergo some developmental and morphological changes (*i.e.*, multiplication of infective stages in intermediate hosts) and definitive hosts (parasites reach sexual maturity in definitive hosts). For many endoparasites with a complex life cycle (*e.g.*, trematodes and nematodes maturing in fish-eating birds), fish act as intermediate hosts. Some endoparasites (*e.g.*, heterophyid metacercariae in the brain of fish and plerocercoids of diphyllobothriidean cestodes in the body cavity) are able to manipulate the behaviour of their intermediate host (here, a fish) to successfully reach the definitive host (PITT – Parasite Increased Trophic Transmission).

Population ecology of parasites – basic terminology

> **Population:** a group of individuals belonging to the same species living at a given time and in a given space; each individual host is parasitised by one or more parasite infrapopulations. The following types of parasite populations have been defined (Margolis *et al.* 1982; Bush *et al.* 1997, 2001; Morand & Šimková 2005).

Infrapopulation: the group of all individuals of a given parasite species infecting a single host specimen; each individual host is parasitised by one parasite population of a single parasite species or more parasite infrapopulations of different parasite species; an infrapopulation is short-living, *i.e*., its maximal life span is equal to (but usually shorter than) the life of the individual host harbouring this infrapopulation. Parasite infrapopulations are subunits of a metapopulation.

Metapopulation (sometimes termed component population): consists of all infrapopulations of a given parasite species in all host individuals of the same host species in an ecosystem.

Suprapopulation: consists of all parasites of a given species including all developmental stages of this parasite in all hosts in a given ecosystem.

Population ecology of host-parasite interactions is analogous to metapopulation theory. The principal idea of metapopulation theory is that the local populations are interconnected, *i.e.*, there is migration of specimens among local populations. Each individual host represents the equivalent of a habitat patch, which usually includes the infrapopulations of more metapopulations of different parasite species infecting a given host population.

To describe the size and distribution of a parasite population in a given host population, Margolis *et al.* (1982) and Bush *et al.* (1997) proposed the **basic epidemiological parameters** describing the level of parasite infection in a host population:

Prevalence: the proportion of hosts infected by a given parasite species (*i.e.*, the proportion of hosts infected in the whole sample of host specimens examined).

Intensity of infection: the number of parasite specimens found in/on a given host specimen infected.

Mean intensity of infection: the mean number of parasites of a given parasite species over all infected hosts in the sample.

Mean parasite abundance: the mean number of parasites per host specimen in a given host population, *i.e.*, the mean number of parasite specimens calculated when considering both infected and uninfected hosts in the sample.

Parasites are typically aggregated within a host population, which means that many hosts are parasitised by one or very few parasites or are uninfected, and a few hosts are infected with many parasite specimens. The simplest way for the description of this parasite distribution is to calculate the variance/mean ratio. A ratio equalling 1 indicates random distribution, a ratio below 1 indicates a uniform distribution and a ratio higher than 1 indicates an aggregated distribution.

Parasite communities – basic terms

Several types of parasite communities have been defined (Bush *et al.* 1997, 2001; Poulin 2007):

> **Infracommunity:** all populations of different species of parasites in the same host individual.
>
> **Component community (or metacommunity):** all parasite species exploiting a host population.
>
> **Compound community:** all parasite communities in an ecosystem.
>
> As infracommunities are subsets of the component community, the maximum number of species in an infracommunity is equal to the number of species in the component community (however, this maximum number of parasite species in an infracommunity is typically not reached and usually no single infracommunity contains all species that are locally available). Infracommunities are short-lived, their maximum life span is equal to that of the host. As component communities are subsets of the parasite fauna, the maximum number of parasite species in a component community is equal to the number of species in the parasite fauna (however, this maximum number of parasite species in a component community is typically not reached). Component communities are longer-lived assemblages than infracommunities as the host population persists in time (Poulin 2007). Component communities are often saturated (expressed by a curvilinear function) by parasite species (the saturation by species is below the number of species in the parasite fauna).
>
> Parasite species are not randomly distributed among infracommunities due to species interactions or other structuring forces. Parasite infracommunities may exhibit so-called **nested patterns** of parasite species distribution when a common parasite species (*i.e.,* usually a parasite with high prevalence and abundance) is distributed in all infracommunities, but rare parasite species occur only in species-rich infracommunities (Patterson & Atmar 1986). This nested pattern is usually explained by different colonisation and extinction rates of species.

Parasite interactions: competition versus coexistence in parasite communities

There are two types of parasite communities:

(1) non-interactive (isolationist) communities, in which niche space is not saturated with parasite individuals and thus interspecific interactions do not play a role (parasites may coexist in the communities);

(2) interactive communities, in which niche space is saturated and interspecific competition plays an important role (Rohde 1977, 1991).

The ecological niche of a given parasite species is the multidimensional habitat volume occupied by specimens of this parasite species. It is defined by physical and biotic variables (Hutchinson 1957 and modified for parasites by Poulin 2007). The comparison of basic niche (measured for a single species infection) and real ecological niche (measured for a multispecies infection) under experimental conditions is the basic way to reveal ongoing competition. The ecological niche of a parasite species is determined by host specificity, microhabitat, macrohabitat (*i.e.*, the habitat of the host), geographical distribution, host age, host food and rarely by host sex (Rohde 1979).

Host specificity

The most widely used descriptor of parasites in their communities is the host specificity. According to the most widely accepted definition, host specificity is the extent to which a parasite taxon is restricted in the number of host species used at a given stage in the life cycle (Poulin 2007). Using a basic measure of host specificity (*i.e.*, host specificity measured by the number of host species), a specialist (or strictly host-specific parasite) is restricted to a single host species, while a generalist (*i.e.*, parasite species with low host specificity) is able to infect at least two host species. Host specificity decreases with an increasing number of host species (*i.e.*, with increasing host range).

Special attention should by paid to parasite species with a complex life cycle. A parasite species with a complex life cycle is often restricted to a single intermediate host species (*i.e.*, it is a specialist at the intermediate host level), but is able to infect a wide range of definitive hosts (*i.e.*, it is a generalist at the final host level). Host specificity may also be expressed by including quantitative ecological data (like abundance), phylogenetic relatedness of hosts or the geographical distribution range of parasite species (Poulin *et al.* 2011). When evaluating host specificity, the scale of the study should be taken into account. Some parasites may exhibit strict host specificity at the local level, but are recorded on a wide range of host species at the regional level.

Analyses of parasite communities – biodiversity indices

Diversity of parasite communities is expressed by species richness or by the relative abundance of species. Species richness is a simple count of the number of species in the community. Relative abundance specifies the number of individuals per species. Biodiversity indices are frequently used to express the diversity in parasite communities (see Maguran 2003). The Shannon index and its evenness have been widely applied for parasite component communities. In contrast, the Brillouin index is useful at the level of the infracommunity. Species dominance in parasite communities can be evaluated using the Simpson index or the Berger-Parker index (see Table 3.6.1 for equations).

Table 3.6.1 Overview of biodiversity indices (S – number of species, N – number of individuals, n_i – number of individuals of the i-th species).

Index	Equation
Margalef index	$D_{Mg} = \frac{(S-1)}{\ln N}$
Menhinick index	$D_{Mn} = \frac{S}{\sqrt{N}}$
Shannon index	$H' = -\sum p_i \ln p_i$, where $p_i = \frac{n_i}{N}$
Brillouin index	$HB = \frac{\ln N! - \sum \ln n_i!}{N}$
Simpson index	$D = \sum \left(\frac{n_i(n_i-1)}{N(N-1)} \right)$
Berger-Parker index	$d = \frac{N_{max}}{N}$ where N_{max} – abundance of the most abundant species

Parasite communities are compared by calculating the similarity between parasite communities (*e.g.*, similarity between two parasite component communities of the same host species collected from two different sites). The coefficient of associations is calculated with or without taking into account the problem of double zero values (asymmetrical and symmetrical coefficient, respectively). Binary or quantitative data are used to evaluate the similarity between parasite communities. The most often applied asymmetrical indices are the Jaccard index of similarity for binary data and the Sørensen index for quantitative data (see Table 3.6.2 for equations).

Table 3.6.2 Basic similarity indices.

Index	**Equation**
Jaccard similarity coefficient	$S = \dfrac{a}{a+b+c}$, where *a* is the number of species occurring at both sites and *b*, *c* is the number of species occurring only at one of the sites
Sørensen quantitative coefficient	$C_N = \dfrac{2jN}{aN+bN}$, where *aN* and *bN* are the abundance of species at sites A and B, and *jN* is the sum of abundances of species occurring at both sites

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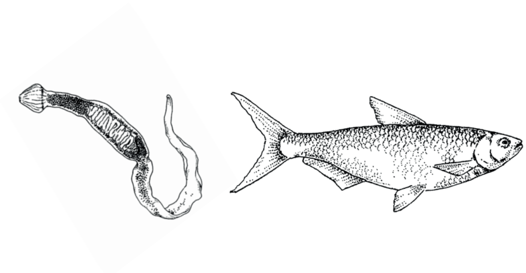
PART 4

A SYSTEMATIC SURVEY OF THE PARASITES OF FRESHWATER FISHES IN AFRICA



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Chapter 4.1.

KEY TO THE PRINCIPAL GROUPS OF THE PARASITES OF FRESHWATER FISHES IN AFRICA*

Roman KUCHTA

1 (2) Microscopic organisms, mostly unicellular, may form cysts containing spores that are not visible to the naked eye, cysts sometimes macroscopic……………………….**Protista and Myxozoa** (see key in Chapter 3.3.1)

2 (1) Organisms visible to the naked eye (may nonetheless be quite small and larvae may be microscopic), multicellular, may or may not be aggregated into clusters of individuals ..………………………………………………3

3 (4) Worm-like organism, lacking an articulated exoskeleton with segmented appendages……………………………………………………………......5

4 (3) Organisms with articulated exoskeleton and segmented appendages (appendages may be minute requiring a microscope to be observed)…………………………………………………………………..15

5 (6) Organisms with dorsoventrally flattened body, not round in cross-section…………………………………………………………………...7

6 (5) Organisms not dorsoventrally flattened, round in cross-section, endoparasitic …………………………………………………………………….13

7 (8) Organisms with the anterior and posterior attachment organ…………….9

8 (7) Organisms without the posterior attachment organ, usually proglottised [Fig. 4.1F]………………………………………**Cestoda** (see Chapter 4.6)

9 (10) Anterior and posterior attachment organs sucker-like, without armatures…………………………………………………………………….....11

10 (9) The posterior attachment organ (haptor) comprising various sclerotised structures (hooks, clamps, squamodiscs) present [Fig. 4.1A, B]…………………………………………**Monogenea** (see Chapter 4.4)

11 (12) Anterior and posterior attachment organs present with well-defined posterior sucker; intestine not bifurcate; always ectoparasites [Fig. 4.1E] ……………………………………………**Hirudinea** (see Chapter 4.10)

* The key does not include encysted helminth larvae; these larvae have to be taken out from the cyst before identification or fixation, usually using fine preparation.

12 (11) Posterior attachment organ usually not present, circumoral and ventral suckers present (except for blood-dwelling species and Aspidogastrea); intestine mostly bifurcate; always endoparasitic [Fig. 4.1C, D]……………………………………………..**Trematoda** (see Chapter 4.5)

13 (14) Anterior end with retractable spined proboscis; intestine absent [Fig. 4.1G] …………………………………………..**Acanthocephala** (see Chapter 4.7)

14 (13) Anterior spined retractable proboscis absent; intestine present [Fig. 4.1H] ………………………………………………..**Nematoda** (see Chapter 4.8)

15 (16) Body not covered by carapace……………………………………………17

16 (15) Almost whole body covered by carapace; four swimming legs [Fig. 4.1I] ……………………………………………….**Branchiura** (see Chapter 4.9)

17 (18) Organisms with two compound eyes; body dorsoventrally flattened, segmented; more than 4 legs [Fig. 4.1J]……………**Isopoda** (see Chapter 4.9)

18 (17) Organisms with one compound eye; body shape variable [Fig. 4.1K] ………………………………………………..**Copepoda** (see Chapter 4.9)

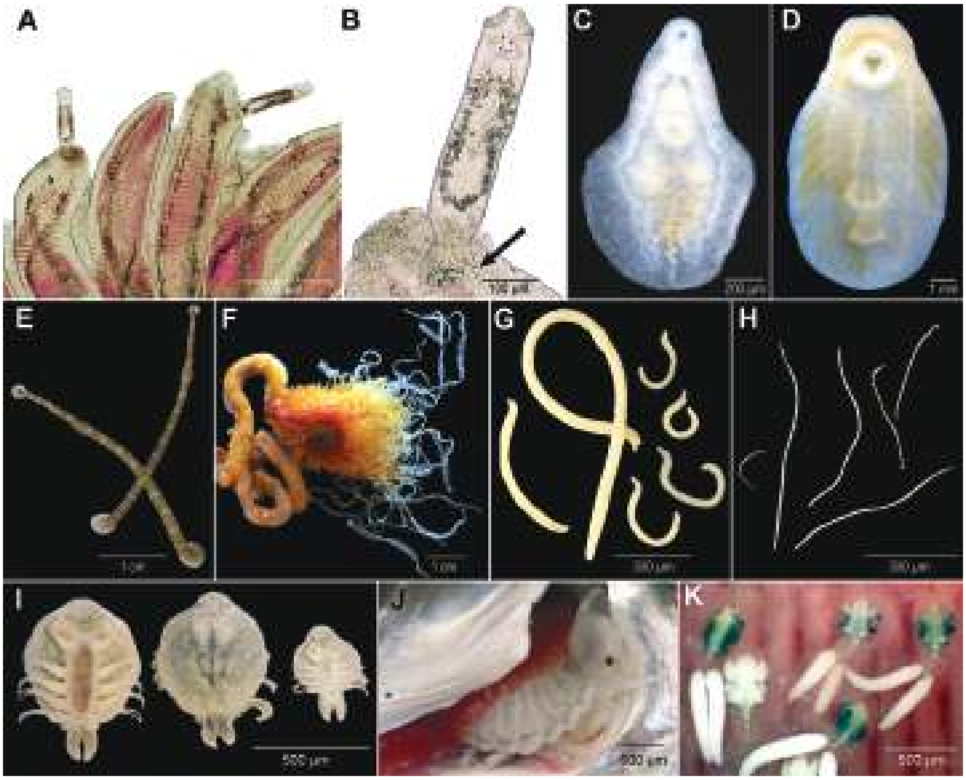

Fig. 4.1. Principal groups of metazoan fish parasites. **A, B.** Monogenea – *Thylacicleidus serendipitus* Wheeler et Klassen, 1988 from *Dichotomyctere nigroviridis*; arrow indicates position of the haptor; **C.** Digenea – *Phyllodistomum* sp.; **D.** Digenea – *Euclinostomum* sp., metacercaria; both from *Clarias gariepinus*; **E.** Hirudinea – *Piscicola geometra* Linnaeus, 1761 from *Cyprinus carpio*; **F.** Cestoda – *Ichthybothrium* sp. in the intestine of *Mesoborus crocodilus*; **G.** Acanthocephala – *Echinorhynchus* cf. *gadi* Zoega in Müller, 1776 from *Microgadus proximus*; **H.** Nematoda – *Procamallanus* sp. from *C. gariepinus*; **I.** Branchiura – *Dolops ranarum* (Stuhlmann, 1892); **J.** Isopoda – *Mothocya renardi* (Bleeker, 1857) from *Strongylura leiura*; **K.** Copepoda – *Ergasilus* sp. on the gills of *C. carpio*. (Photographs by R. Kuchta, O. Kudlai, D. Modrý & E. Řehulková).

Chapter 4.2.

PROTISTA

Linda BASSON & Courtney COOK

Protists parasitising freshwater fishes – basic characteristics, life cycles, classification and principal diagnostic features

Protists do not represent a distinct and monophyletic group of organisms. According to Adl *et al.* (2005) Haeckel's taxon Protista (Haeckel 1866) is no longer formally recognised. However, the popular term "protist" is retained to describe eukaryotes with a unicellular level of organisation (eukaryotic microorganisms or EMs; see Chapter 3.3.1). Therefore, this term will be used throughout this chapter, but with no taxonomic validity. The various groups discussed below belong to supergroups as proposed by Adl *et al.* (2012). The only characteristic these organisms share is the fact that they are all unicellular. Very scant information on protist fish parasites in Africa exists.

Each taxonomic group is discussed separately throughout. Host names are presented according to Froese & Pauly (2017). For the purpose of this chapter, the classification system proposed by Adl *et al.* (2012) has been followed. A brief outline of this system is presented in Table 4.2.1, limited to groups of parasites recorded from African freshwater fishes. A generalised key to major groups is presented in Chapter 3.3.1.

In cases where a representative species for every genus could be obtained from the relevant African literature, these species are presented in diagrammatic drawings. However, in several instances only records of genera are provided, with no species identification and/or micrographs or diagrams. In these cases, a representative species from elsewhere in the world was selected and diagrammatically presented.

Practical key for preliminary determination of fish-infecting EMs in freshly prepared material

1 (2) Protists detectable as macroscopic whitish aggregations, from tiny dots to cyst-like structures of several millimetres in size; on skin, gills, in or on internal organs……………………………………………………………….3

2 (1) No macroscopic changes visible; protists only detectable by light microscopy………………………………………………………………………….17

3 (4) Microorganisms visible as tiny dots on the body surface and gills, under the microscope dot proves to be one or several large (up to 1 mm) slowly rotating cells, uniformly covered with beating cilia; smaller cells may be

present, next to large ones; cytoplasm full of granules, containing large macronucleus [Fig. 4.2.4E]……………………***Ichthyophthirius multifiliis***

4 (3) Dot-, nodule-, or cyst-like structures composed of mass of small, uniform, refractile bodies (spores or oocysts)………………………………………5

5 (6) Spores very small, typically 3-10 µm in size, usually ovoid and often showing prominent vacuole in posterior part (Microsporidia)………………9

6 (5) Spores spherical or ellipsoid-spherical……………………………………7

7 (8) Spherical spores with a large central vacuole/light refracting bodies [Fig. 4.2.2B]………………………………………………***Dermocystidium***

8 (7) Organisms spherical or ellipsoidal bodies of about 10-20 µm in size, each with 4 sharply delimited (coccidian oocysts)……………………………… 15

9 (10) Microsporidian not directly associated with fish, hyperparasite [Fig. 4.2.2F] ……………………………………………………………………..***Unikaryon***

10 (9) Microsporidians associated directly with fish………………………………11

11 (12) First merogony stages with diplokarya [Fig. 4.2.2D]……***Neonosemoides***

12 (11) No diplokaryon in the developmental series……………………………….13

13 (14) Xenoma wall consists of granulo-fibrillar layer, spores throughout xenoma [Fig. 4.2.2C]………………………………………………………… ***Loma***

14 (13) Merogony and sporogony stages with conspicuous envelope [Fig. 4.2.2E] ……………………………………………………………….***Pleistophora***

15 (16) One pole of sporocyst bearing special structure (Stieda body) [Fig. 4.2.2I] …………………………………………………………………….. ***Eimeria***

16 (15) Sporocyst without Stieda body [Fig. 4.2.2J]………………………..***Goussia***

17 (18) Protists infecting surface (skin, fins, nasal pits or gills)………………….. 19

18 (17) Protists infecting intestine, other internal organs or blood…………………55

19 (12) Organisms that move………………………………………………………21

20 (11) Sessile or motionless organisms attached to surface……………………29

21 (14) Protists with flagella or cilia on the cell surface ……………………………23

22 (13) Cells with amoeboid movement and changes of body shape [Fig. 4.2.2A] …………………………………………………………………***Entamoeba***

23 (16) Protists possessing 2 flagella, moving with jerky, creeping motion or swimming spirally forward (flagellates)……………………………………...25

24 (15) Protists 20 μm and larger, either covered uniformly with cilia or with several ciliary belts or circular ciliary wreath; they move directly forward, glide over the surface, or roll on the spot (ciliophorans)……………………………..…41

25 (26) No mitochondrion present [Fig. 4.2.5F] …………………………..***Hexamita***

26 (25) Mitochondrion present …………………………………………………… 27

27 (28) Long tubular mitochondrion contains numerous nucleoids so that there are many small kinetoplasts throughout the body [Fig. 4.2.5G]………………… ………………………………………………………………***Ichthyobodo***

28 (27) Single branched mitochondrial ribbon forms massive, elongate kinetoplast on the ventral surface [Fig. 4.2.5H]………………………………. ***Cryptobia***

29 (30) Refractile granules in cytoplasm…………………………………………31

30 (29) Goblet-like or cylindrical, each with wide free end and encircled by wreaths of beating cilia; cells may contract a little (sessilines)……………………. 33

31 (32) Pyriform or sack-like flagellated protist, cytoplasm yellowish or greenish (parasitic dinoflagellates) [Fig. 4.2.2H]…………………….***Piscinoodinium***

32 (31) Cytoplasm dark due to refractile granules, with bundles of tubules ending in knob-like shapes (suctorians) [Fig. 4.2.4D]……………………..***Capriniana***

33 (34) Sessilines attach directly to substrate via scopula…………………………35

34 (33) Sessilines attach to substrate via a stalk……………………………………39

35 (36) Permanent locomotory equatorial fringe present [Fig. 4.2.5D]……………… …………………………………………………………………***Ambiphrya***

36 (35) Locomotory fringe of cilia only present in free-swimming larval stage …… ……………………………………………………………………… 37

37 (38) Body elongate, macronucleus compact, conical or ellipsoidal [Fig. 4.2.5B] …………………………………………………………………..***Apiosoma***

38 (37) Body cylindrical, macronucleus sausage-shaped [Fig. 4.2.5C]…………… ……………………………………………………………..***Riboscyphidia***

39 (40) Stalk highly contractile and unbranched [Fig. 4.2.5E]……………***Vorticella***

40 (39) Stalk non-contractile, bearing a small colony of several zooids [Fig. 4.2.5A] ……………………………………………………………………***Epistylis***

41 (42) Cilia in distinct rows……………………………………………………43

42 (41) Cilia limited to aboral wreath (around concave adhesive disc) and an adoral spiral of cilia (feeding organelles at the opposite side of adhesive disc); aboral side with distinct adhesive disc consisting of prominent interlinking

denticles (mobilines)……………………………………………………………45

43 (44) Ciliary rows limited to one surface of the organism…………………………53

44 (43) Pyriform ciliophorans with 2-30 meridional kineties [Fig. 4.2.4F]……………
……………………………………………………………………***Tetrahymena***

45 (46) Adoral spiral makes a full turn or slightly more………………………………47

46 (45) Adoral spiral makes less than one full turn…………………………………49

47 (48) Denticles have well-developed rays and blades [Fig. 4.2.6C]………………
………………………………………………………………………***Trichodina***

48 (47) Denticles have stunted blades [Fig. 4.2.6A]………………***Hemitrichodina***

49 (50) Denticles have well-developed rays………………………………………51

50 (49) Denticles have rays that merely form small hooks [Fig. 4.2.6D]……………
……………………………………………………………………***Trichodinella***

51 (52) Denticles interlinked only by central parts [Fig. 4.2.6B]………………………
…………………………………………………………………… ***Paratrichodina***

52 (51) Denticles interlinked by central parts, as well as by a prominent anterior projection of blades, fitting tightly into corresponding notches in blades of preceding denticles [Fig. 4.2.6E]……………………………***Tripartiella***

53 (54) One side bears longitudinal or strongly arched ciliary rows [Fig. 4.2.4B]
……………………………………………………………………***Amphileptus***

54 (53) Ventral ciliature reduced to two longitudinal belts close to body margins [Fig. 4.2.4C]…………………………………………………….. ***Chilodonella***

55 (56) Protists in internal organs or urinary tract……………………………………57

56 (55) Protists in blood………………………………………………………………63

57 (58) Microsporidia (see 5; in any organ), coccidian oocysts (see 8; in intestine), or amoebae (see 22)

58 (57) Protists with surface showing cilia……………………………………… 59

59 (60) Cilia uniformly covering body of ciliophoran………………………………..61

60 (59) Cilia limited to aboral wreath as well as an adoral spiral of cilia. The aboral side with distinct adhesive disc consisting of denticles (endoparasitic trichodinids) [Fig. 4.2.6C]………………………………………… ***Trichodina***

61 (62) Spindle-shaped cells, with both ends pointed, showing sluggish movement; two to many monomorphic nuclei [Fig. 4.2.2G]………………***Protoopalina***

62 (61) Ciliophorans covered uniformly in longitudinal rows of cilia; single elongate macronucleus and single spherical micronucleus [Fig. 4.2.4A]………………………………………………………………………………….. ***Balantidium***

63 (64) Motile protists, slender cells, typically 10-15 μm long, moving with a wriggling or undulating motion, with 1 or 2 flagella [Fig. 4.2.3D]……………………………………………………………………………...***Trypanosoma***

64 (63) Non-motile protists only visible in stained blood smears, found within blood cells………………………………………………………………………… 65

65 (66) Intraerythrocytic meronts (division stage showing more than one nuclei) and gamonts (sexual stage showing a single nucleus)………………….. 67

66 (65) Intraerythrocytic gamonts only [Fig. 4.2.3C]……………………….***Desseria***

67 (68) Intraerythrocytic meronts rounded [Fig. 4.2.3A]……………..***Babesiosoma***

68 (67) Intraerythrocytic meronts vermicular (wormlike) [Fig. 4.2.3B]………***Cyrilia***

Fig. 4.2.1. A. Life cycles of the ciliophoran *Ichthyophthirius multifiliis* Fouquet, 1876 (direct life cycle without intermediate hosts); **B.** The blood kinetoplastid *Trypanosoma* sp. (indirect life cycle where leeches serve as intermediate hosts). (Illustration by M. Luo.)

Table 4.2.1. Classification system for the protists according to Adl *et al.* (2012).

Supergroup	First rank	Second rank – examples
AMOEBOZOA	Archamoebae	Entamoebidae (*Entamoeba*)
OPISTHOKONTA	Holozoa	Ichthyosporea (*Dermocystidium*)
	Nucletmycea	Fungi (*Eimeria*, *Glugea*, *Loma*, *Neonosemoides*, *Pleistophora*, *Unikaryon*)
EXCAVATA	Diplomonanida	Hexamitinae (*Hexamita*)
	Euglenozoa	Prokinetoplastina (*Cryptobia*, *Ichthyobodo*, *Trypanosoma*)
SAR	Stramenopiles	Opalinata (*Protoopalina*)
	Alveolata	Dinoflagellata (*Piscinoodinium*)
		Apicomplexa (*Babesiosoma*, *Cyrilia*, *Desseria*, *Eimeria*, *Goussia*, haemogregarines)
SAR	Alveolata	Ciliophora; Trichostomatia* (*Amphiletus*, *Balantidium*) Phyllopharyngea* (*Chilodonella*) Suctoria** (*Capriniana*), Oligohymenophorea*; Hymenostomatia** (*Ichthyophthirius*, *Tetrahymena*) Oligohymenophorea*; Peritrichia** (*Ambiphrya*, *Apiosoma*, *Epistylis*, *Hemitrichodina*, *Paratrichodina*, *Riboscyphydia*, *Trichodina*, *Trichodinella*, *Tripartiella*, *Vorticella*)

* fifth rank; ** sixth rank

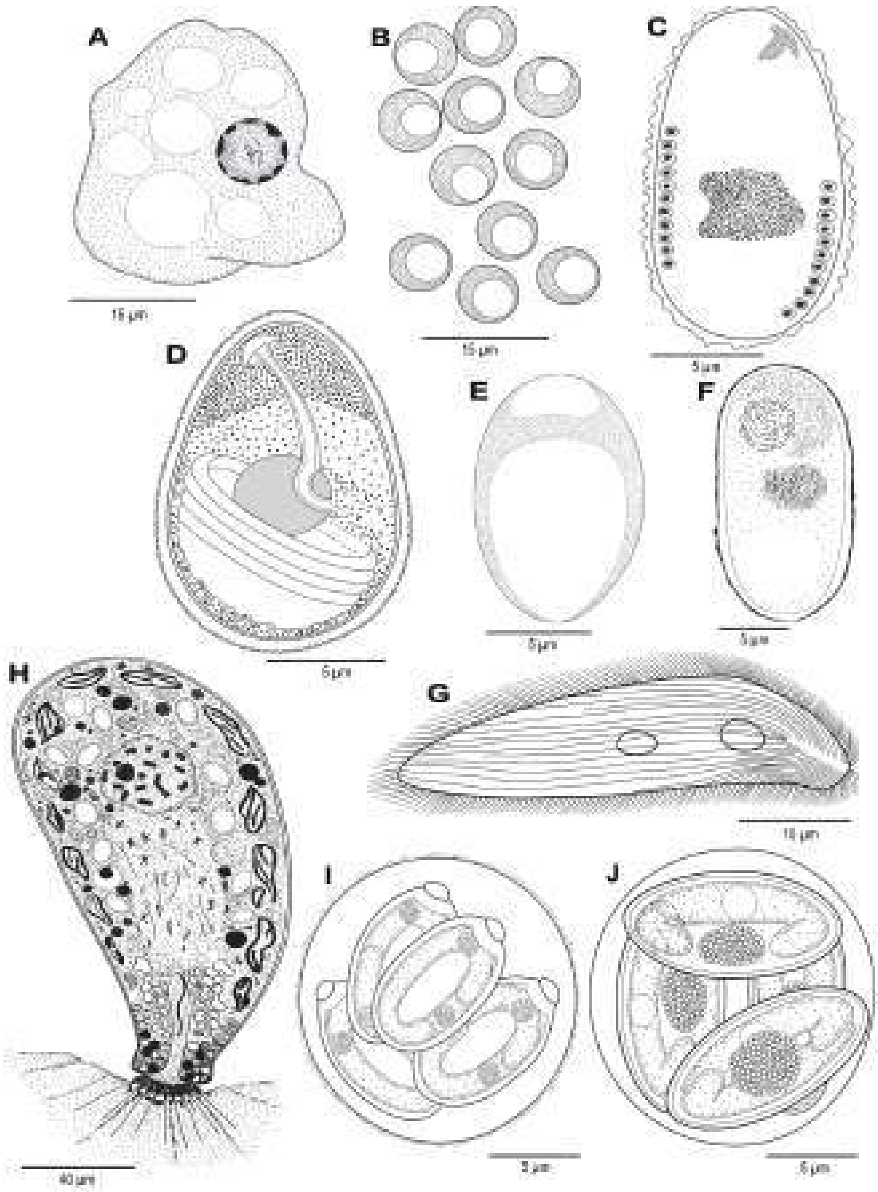

Fig. 4.2.2. Schematic line drawings of fish-infecting eukaryotic microorganisms (EMs). **A.** *Entamoeba salpae* (Alexeieff, 1912) from *Boops salpa*; **B.** *Dermocystidium branchiale* Léger, 1914 from *Salmo trutta*; **C.** *Loma camerounensis* Fomena, Coste et Bouix, 1992 from *Oreochromis niloticus*; **D.** *Neonosemoides* sp. from *Chrysichthys auratus*; **E.** *Pleistophora elegans* Auerbach, 1910 from *Alburnus alburnus*; **F.** *Unikaryon nomimoscolexi* Sene, Ba, Marchand et Toguebaye, 1997 from *Clarotes laticeps*; **G.** *Protoopalina symphysodonis* Foissner, Schubert et Wilbert, 1974 from *Symphysodon aequifasciata*; **H.** *Piscinoodinium pillulare* (Schäperclaus, 1954) from *Poecilia reticulata*; **I.** *Eimeria variabilis* (Thélohan, 1893) from *Cottus bubalis*; **J.** *Goussia anopli* Molnár, Avenant-Oldewage et Székely, 2004 from *Enteromius anoplus*. (Modified from Davies 1978; Fomena *et al.* 1992; Lom & Dyková 1992; Sene *et al.* 1997; Molnár *et al.* 2004; Reda 2010.)

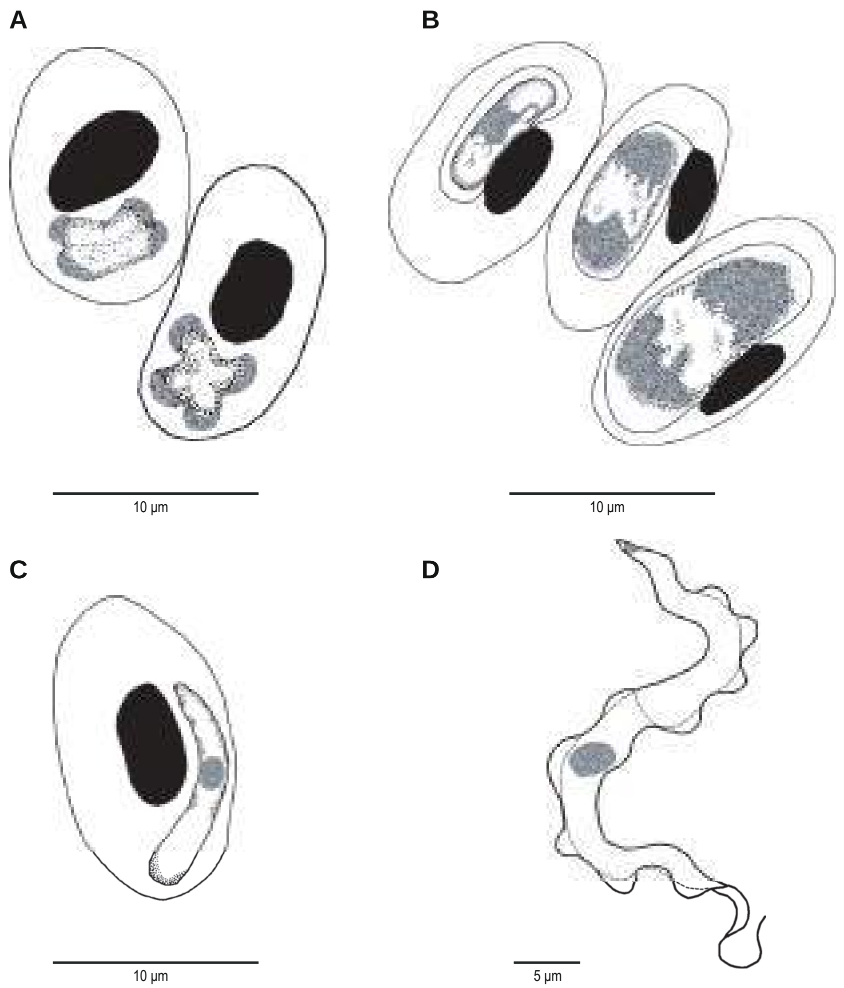

Fig. 4.2.3. Schematic line drawings of blood parasites reported from the peripheral blood of African freshwater fishes; **A.** Left to right: young meront in division and characteristic mature cruciform meront with four merozoites of *Babesiosoma mariae* (Hoare, 1930) from various freshwater fish species; **B.** Left to right: gamont, young meront and mature meront stage of *Cyrilia gomesi* (Neiva et Pinto, 1926) from *Synbranchus marmoratus*; **C.** Gamont stage of *Desseria* sp. from *Mugil cephalus*; **D.** Trypomastigote stage of *Trypanosoma mukasai* Hoare, 1932 from a freshwater fish. (Modified from Hoare 1930, 1932; Lainson 1981; Smit *et al.* 2002.)

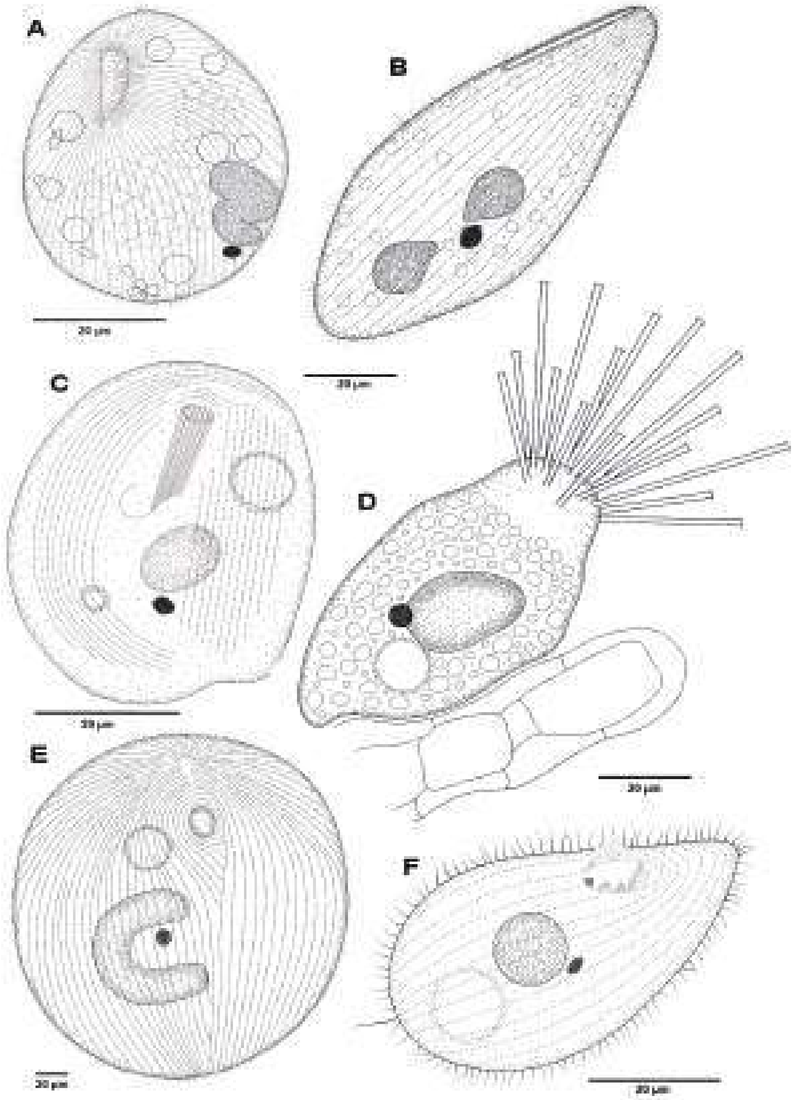

Fig. 4.2.4. Schematic line drawings of ciliophorans. **A.** *Balantidium polyvacuolum* Li, 1963 from *Xenocypris argentea*; **B.** *Amphileptus branchiarum* Wenrich, 1924 from a freshwater fish; **C.** *Chilodonella piscicola* (Zacharias, 1894) from *Tilapia sparrmanii*; **D.** *Capriniana piscium* (Buetschli, 1889) from *Perca fluviatilis*; **E.** *Ichthyophthirius multifiliis* Fouquet, 1876 from *Oreochromis mossambicus*; **F.** *Tetrahymena corlissi* Thompson, 1955 from a freshwater fish. (Modified from Li 1963; Lom & Dyková 1992.)

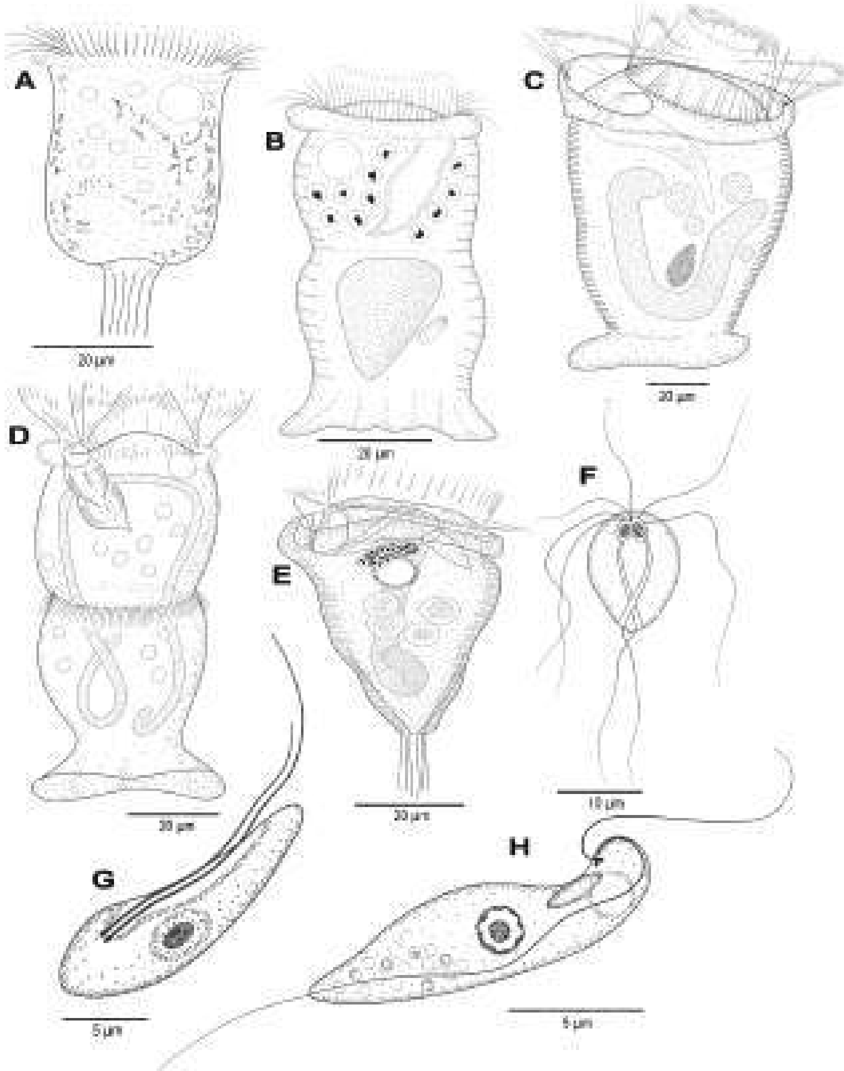

Fig. 4.2.5. Schematic line drawings. **A.** *Epistylis transvaalensis* Viljoen et Van As, 1983 from *Pseudocrenilabrus philander*; **B.** *Apiosoma dermatum* Viljoen et Van As, 1983 from *Oreochromis mossambicus*; **C.** *Riboscyphidia arctica* (Zhukov, 1962) from *Liparis* sp.; **D.** *Ambiphrya neobolae* Viljoen et Van As, 1985 from *Neobola brevianalis*; **E.** *Vorticella* sp. from a freshwater fish; **F.** *Hexamita salmonis* (Moore, 1923) from *Salmo trutta*; **G.** *Ichthyobodo necator* (Henneguy, 1883) from *Cyprinus carpio*; **H.** *Cryptobia branchialis* Nie in Chen, 1955 from *Clarias gariepinus*. (Modified from Viljoen & Van As 1983, 1985; Lom & Dyková 1992; Lom & de Puytorac 1994.)

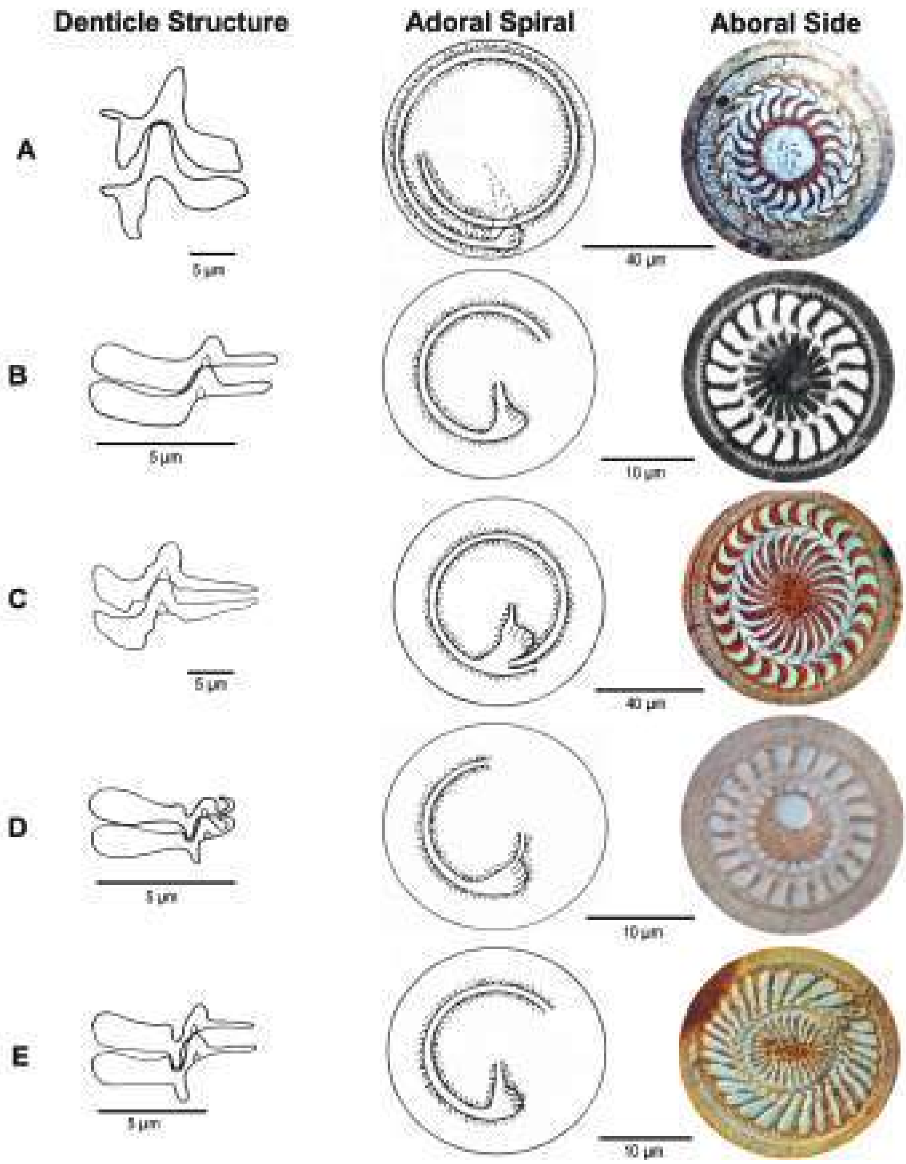

Fig. 4.2.6. Schematic line drawings and silver impregnated adhesives discs of five genera of trichodinids. **A.** *Hemitrichodina robusta* Basson et Van As, 1989 from *Marcusenius macrolepidotus*; **B.** *Paratrichodina corlissi* Lom et Haldar, 1977 from *Gobio* sp.; **C.** *Trichodina magna* Van As et Basson, 1989 from *Oreochromis mossambicus*; **D.** *Trichodinella epizootica* (Raabe, 1950) from *Enteromius paludinosus*; **E.** *Tripartiella ctenopomae* Basson et Van As, 2002 from *Ctenopoma multispine*. (Modified from Basson & Van As, 1989; micrographs by L. Basson except for B, which was provided by the late J. Lom.)

AMOEBOZOA Lühe, 1913

Archaomoebae Cavalier-Smith, 1983 – **Entamoebidae** Chatton, 1925

Amoeboid organisms – basic characteristics

- amoebae infecting fish either specific endocommensals or free-living (see Dyková 2008)
- only species of *Entamoeba* reported from African fishes
- cilia and centrioles absent
- contain mitosomes instead of classical mitochondria
- peroxisome-absent
- mitosis closed with endonuclear centrosomes and spindle
- reduced Golgi dictyosome

List of amoeboid protists in African freshwater fishes

Note: Dyková *et al.* (2005) characterised 17 strains of flattened amoebae from fishes obtained from various sources and deposited at the Institute of Parasitology, Biology Centre of the Czech Academy of Sciences. Mention is made of two freshwater strains imported from Africa, both from the gills of *Clarias angolensis* that formed part of the analyses in their study.

Entamoeba Cassagrandi et Barbagallo, 1895

Entamoeba synodontis Imam, Ramadan and Derahlli, 1987 from ***Synodontis schall*** (Egypt)*

Entamoeba sp. on the skin and gills of *Clarias gariepinus* [Fig. 4.2.2A]

OPISTHOKONTA Cavalier-Smith, 1987

Ichthyosporea Cavalier-Smith, 1998 – **Rhinosporidaceae** Mendoza *et al.*, 2001

Ichthyosporeans – basic characteristics

- posterior cilia, if cilia present
- flat mitochondrial cristae present (folds of mitochondrial inner membrane)
- parasitic forms spherical phenotypes with several endospores 2-20 μm in diameter eventually released, becoming mature cells with endospores to continue with the parasitic cycle

List of ichthyosporeans from African freshwater fishes

Dermocystidium Pérez, 1908

Dermocystidium sp. on the skin of *Carassius auratus* and *Cyprinus carpio* [Fig. 4.2.2B]

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*The type species of parasite genera and type host of species are highlighted in bold. The country where the type locality lies is also provided if known.

Fungi Moore, 1980 – **Microsporidia** Balbiani, 1882

Microsporidia – basic characteristics

- parasites of nearly all animal phyla, with the majority of species associated with arthropods and fishes
- obligate intracellular parasites, usually of animals
- mitochondria reduced to mitosomes
- spores with inner chitin walls and outer proteinaceous walls
- without kinetosomes, centrioles or cilia
- centrosomal plaque
- extrusive specialised polar tube for host penetration
- reproduction sexual, asexual or both
- systematic subdivisions uncertain at this time
- many of those genera that are found as parasites in fishes exhibit a complex coexistence with their host cell that includes a special type of hypertrophy, forming structures known as xenomas
- about 100 microsporidian species known from fishes (see Lom 2002)

List of microsporidians from African freshwater fishes

Loma Morrison et Sprague, 1981

Loma camerounensis Fomena, Coste et Bouix, 1992 in subepithelial connective tissue of the intestine of ***Oreochromis niloticus*** (Cameroon) [Fig. 4.2.2C]

Neonosemoides Faye, Toguebaye et Bouix, 1996

Neonosemoides tilapiae Faye, Toguebaye et Bouix, 1996 in the stomach of ***Coptodon guineensis*** (Senegal)

Neonosemoides sp. in the gills of *Chrysichthys auratus* [Fig. 4.2.2D]

Pleistophora Gurley, 1893

Pleistophora-like species in the swim-bladder of *Haplochromis angustifrons* and *H. elegans* [Fig. 4.2.2E]

Unikaryon Canning, Lai et Lie, 1974

Unikaryon nomimoscolexi Sene, Ba, Marcand et Toguebaye, 1997 in the cestode *Nomimoscolex* sp. from ***Clarotes laticeps*** (Senegal), *i.e.*, hyperparasite [Fig. 4.2.1F] [the cestode was certainly misidentified as species of *Nomimoscolex* occur in the Neotropical region; it was most likely *Proteocephalus sulcatus* – see Chapter 4.6]

Unidentified microsporidia

Microsporidia gen. sp. from *Clarias gariepinus* (cystozoic), *Oreochromis niloticus* (skin and gills), *Parachanna obscura*, *Synodontis schall*, *S. ocellifer* (both in stomach and intestine)

Stramenopiles Patterson, 1989 – **Opalinata** Wenyon, 1926

Opalins – basic characteristics

- slowly swimming large protists
- covered with numerous cilia arranged in longitudinal, or slightly helicoidal, densely spaced rows
- multiciliated cells with cilia originating from anterior morphogenetic centre, the falx, forming oblique longitudinal rows or files
- microtubular ribbons supporting longitudinal pellicular ridges between ciliary rows
- two to many monomorphic nuclei
- life cycle complex, with sexual processes induced by hormones of host and linked to host's life cycle
- endobionts in amphibians and some fishes (Adl *et al.* 2012).

List of opalins from African freshwater fishes

Protoopalina Metcalf, 1918

Protoopalina sp. in the intestine of *Clarias gariepinus* [Fig. 4.2.2G]

Alveolata Cavalier-Smith, 1991 – **Dinoflagellata** Bütschli, 1885

Dinoflagellates – basic characteristics

- dinoflagellates in fishes with the parasitic stage in the life cycle predominating over the dinospore stage
- cells with two cilia in motile stage
- nucleus typically a dinokaryon (nucleus where chromosomes are fibrillar in appearance and condensed)
- closed dinomitosis (mitosis specifically involving dinokaryon) with extra nuclear spindle

List of dinoflagellates from African freshwater fishes

Piscinoodinium Lom, 1981

Piscinoodinium sp. in the skin and gills of *Clarias gariepinus* [Fig. 4.2.2H]

Apicomplexa Levine, 1980 **– Coccidia** Leuckart, 1879 **(Eimeriorina** Léger, 1911**)**

Coccidia – basic characteristics

- elaborate and intricate apical complex always present
- oocysts always formed

- sporozoites with a three-layered pellicle
- micro- and macrogametes develop independently
- microgamonts produce large numbers of ciliated microgametes
- zygote nonmotile
- sporozoites always enclosed in sporocyst within oocysts

List of coccidia from African freshwater fishes

Eimeria Schneider, 1875

Eimeria sp. from *Clarias* spp. (intestine) and *Synodontis schall* [Fig. 4.2.2I]

Goussia Labbé, 1896

Goussia anopli Molnár, Avenant-Oldewage et Székely, 2004 in mucus and epithelium of the foregut of ***Enteromius anoplus*** (South Africa) [Fig. 4.2.2J]

Goussia cichlidarum Landsberg et Paperna, 1985 in the swim bladder of *Coptodon zillii*, *Oreochromis aureus*, *O. niloticus*, ***Oreochromis* sp.** (Uganda)

Goussia molnarica El-Mansy, 2008 in the intestine of ***Clarias gariepinus*** (Egypt)

Goussia vanasi (Landsberg et Paperna, 1987) [syn. *Eimeria vanasi* Landsberg and Paperna, 1987] in the intestine of *Oreochromis mossambicus*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii* (South Africa)

Coccidia sp. in the intestine of *Chrysichthys nigrodigitatus*

Apicomplexa Levine, 1980 **– Coccidia** Leuckart, 1879 **(Adeleorina** Léger, 1911**)**

Haemogregarines – basic characteristics

- obligate endoparasitic intracellular protists in the blood of a range of vertebrates
- transmitted by invertebrate haematophagous vectors (flies, bugs, ticks and leeches)
- around 400 species recorded globally in vertebrates and invertebrate vectors
- apical complex in infective stages (merozoites, sporozoites)
- heteroxenous, asexual development (including merogony and gamogony) in vertebrate host
- sexual development (including gametogenesis, syngamy, ookinete formation and sporogony) in invertebrate vector
- transfer of infective stages to and from the invertebrate vector presumed to be inoculative
- leeches implicated as the invertebrate vector for most African genera

List of haemogregarines from African freshwater fishes

Babesiosoma Jakowski et Nigrelli, 1956

Babesiosoma hannesi (Paperna, 1981) from *Chelon dumerili*, *Chelon richardsonii*, ***Mugil cephalus*** (South Africa) [Fish inhabiting marine, freshwater and brackish systems.]

Babesiosoma mariae (Hoare, 1930) [syn. *Dactylosoma mariae* Hoare, 1930] from *Astatoreochromis alluaudi*, *Haplochromis cinereus*, *H. nubilus*, *H. serranus*, ***Haplochromis* spp**. (Uganda), *Labeo victorianus*, *Oreochromis esculentus*, *O. niloticus*, *O. variabilis*, *Serranochromis angusticeps* [Fig. 4.2.3A]

Cyrilia Lainson, 1981

Cyrilia nili (Wenyon, 1909) [syn. *Haemogregarina nili*] from ***Parachanna obscura*** (Sudan) [Fig. 4.2.3B]

Desseria Siddall, 1995

Desseria sp. from *Mugil cephalus* [This species was described in detail by Smit *et al.* (2002), but not named. As such it has been provisionally included here.] [Fig. 4.2.3C]

Haemogregarine gen. sp. from *Synodontis schall*

Ciliophora Doflein, 1901

Ciliophorans – basic characteristics

- among the most common and widely distributed symbionts of fishes, whether as parasites or as ecto- and endocommensals
- monograph by Lynn (2008) gives an overview of these organisms
- highly organised protists with a pellicle covering cell body
- pellicle covered by cilia, may be grouped to form compound ciliary organelles
- ciliature may be reduced in some groups, or completely absent
- infraciliature (complex fibrillar network) associated with cilia’s basal bodies
- one to several diploid macronuclei and one to several polyploid macronuclei
- transverse (homothetogenic) binary fission, rarely budding or multiple fission
- conjugation occurs as sexual reproduction process
- complex buccal apparatus used for feeding, but some groups secondarily astome
- found on the surface or inside animal hosts (variety of symbiotic associations with hosts)

List of ciliophorans from African freshwater fishes

In the following survey, ciliophorans are listed according to their 5th and 6th taxonomic rank (see Table 4.2.1), *i.e.*, Trichostomatia (genera *Amphileptus* and *Balantidium*), Phyllopharyngea (*Chilodonella*), Suctoria (*Capriniana*), Oligohymenophorea – Hymenostomatia (*Ichthyophthirius* and *Tetrahymena*), Oligohymenophorea – Peritrichia (Sessilida: *Ambiphrya*, *Apiosoma*, *Epistylis*, *Riboscyphidia*, *Vorticella*), and Oligohymenophorea – Peritrichia (Mobilida: *Hemitrichodina*, *Paratrichodina*, *Trichodina*, *Trichodinella*, *Tripartiella*).

Trichostomatia Bütschli, 1889

Amphileptus Ehrenberg, 1830

Amphileptus niloticus El-Tantawy, Abdel-Aziz, Abou El-Nour, Samn, Shaldoum et Rady, 2016 on the skin and gills of ***Lates niloticus*** (Egypt)

Amphileptus sp. on the gills of *Clarias gariepinus*, *Lates niloticus*, *Sarotherodon galilaeus* [Fig. 4.2.4B]

Balantidium Claparède et Lachmann, 1858

Balantidium sp. from *Clarias gariepinus* and *Synodontis schall* [Fig. 4.2.4A]

Phyllopharyngea de Puytorac *et al.*, 1974

Chilodonella Strand, 1926

Chilodonella hexasticha (Kiernik, 1909) on the skin, fins and gills of *Carassius auratus*, *Clarias gariepinus*, *Coptodon rendalli*, *C. zillii*, *Enteromius paludinosus*, *Oreochromis mossambicus*, *O. niloticus*, *Pseudocrenilabrus philander*, *Sarotherodon galilaeus*, *Tilapia sparrmanii*

Chilodonella piscicola (Zacharias, 1894) [syn. *C. cyprini* (Moroff, 1902)] from *Coptodon rendalli*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii* [Fig. 4.2.4C]

Chilodonella sp. on the skin and gills of *Clarias gariepinus*, *Clarias* sp., *Cyprinus carpio*, *Heterobranchus bidorsalis*, *H. longifilis*, *Oreochromis mossambicus*, *O. niloticus*, *Synodontis schall*

Suctoria Claparède et Lachmann, 1858

Capriniana Mazzarelli, 1906 [syn. *Trichophrya* Claparède et Lachmann, 1858]

Capriniana sp. on the skin and gills of *Clarias gariepinus* [Fig. 4.2.4D]

Oligohymenophorea de Puytorac *et al.*, 1974 – **Hymenostomatia** Delage et Hérouard, 1896

Ichthyophthirius Fouquet, 1876

Ichthyophthirius multifiliis Fouquet, 1876 on the skin, fins and gills of *Anguilla mossambica*, *Carassius auratus*, *Chrysichthys auratus*, *Clarias gariepinus*, *Cyprinus carpio*, *Enteromius paludinosus*, *Heterobranchus longifilis*, *Labeobarbus aeneus*, *Oncorhynchus mykiss*, *Oreochromis mossambicus*, *O. niloticus*, *Poecilia reticulata*, *Salmo trutta* [Fig. 4.2.4E]

Ichthyophthirius sp. on the skin and gills of *Carassius auratus*, *Clarias gariepinus*, *Oreochromis niloticus*, *Synodontis schall*

Tetrahymena Furgason, 1940

Tetrahymena corlissi Thompson, 1955 on the skin of *Sarotherodon galilaeus* [Fig. 4.2.4F]

Tetrahymena pyriformis (Ehrenberg, 1830) on the skin and gills of *Coptodon zillii*, *Cyprinus carpio*, *Enteromius paludinosus*, *Oreochromis leucostictus*

Tetrahymena sp. on the skin and gills of *Clarias gariepinus*, *Lates niloticus*

Oligohymenophorea de Puytorac *et al.*, 1974 – **Peritrichia** Stein, 1859 (Sessilida Kahl, 1935)

Ambiphrya Raabe, 1952

Ambiphrya ameiuri Davis, 1947 on the skin and gills of *Lates niloticus*, *Sarotherodon galilaeus*

Ambiphrya neobolae Viljoen et Van As, 1985 on the skin of ***Mesobola brevianalis*** (South Africa) [Fig. 4.2.5D]

Ambiphrya sp. on the skin and gills of *Clarias gariepinus*, *Lates niloticus*

Apiosoma Blanchard, 1885 [syns *Glossatella* Bütschli, 1889; *Scopulata* Viljoen and Van As, 1985]

Apiosoma amoebae (Grenfell, 1887) [syn. *Glossatella amoebae* Grenfell, 1887] on the gills of *Lates niloticus*

Apiosoma caulata Viljoen et Van As, 1985 on the skin and gills of ***Mesobola brevianalis*** (South Africa)

Apiosoma conica Shulman, 1984 on the gills of *Coptodon zillii*

Apiosoma constricta (Viljoen et Van As, 1985) [syn. *Scopulata constricta* Viljoen et Van As, 1985] on the skin of *Coptodon rendalli*, *Enteromius trimaculatus*, *Marcusenius macrolepidotus*, *Micralestes acutidens*, ***Oreochromis mossambicus*** (South Africa)**,** *Pseudocrenilabrus philander*, *Tilapia sparrmanii*

Apiosoma curvinucleata Viljoen et Van As, 1985 on the skin of ***Oreochromis mossambicus*** (South Africa)

Apiosoma dermatum (Viljoen et Van As, 1983) [syn. *Scopulata dermata* Viljoen et Van As, 1983] on the skin of *Coptodon rendalli, Enteromius trimaculatus*, *Marcusenius macrolepidotus*, *Micralestes acutidens*, ***Oreochromis mossambicus*** (South Africa), *Pseudocrenilabrus philander*, *Tilapia sparrmanii* [Fig. 4.2.5B]

Apiosoma doliaris Timofeev, 1962 on the gills of *Lates niloticus*

Apiosoma epibranchialis (Viljoen et Van As, 1983) [syn. *Scopulata epibranchialis* Viljoen et Van As, 1983] on the skin of *Lates niloticus*, *Micropterus dolomieu*, *Oreochromis mossambicus*, ***Pseudocrenilabrus philander*** (South Africa), *Sarotherodon galilaeus*

Apiosoma micralesti Viljoen et Van As, 1985 on the skin of ***Micralestes acutidens*** (South Africa)

Apiosoma mothlapitsis Viljoen et Van As, 1985 on the skin of ***Labeobarbus marequensis*** (South Africa)

Apiosoma nasalis (Timofeev, 1962) [syn. *Glossatella nasalis* Timofeev, 1962] on the skin and gills of *Pseudocrenilabrus philander*

Apiosoma obliqua Viljoen et Van As, 1985 on the skin of ***Labeo cylindricus*** (South Africa)

Apiosoma phiala Viljoen et Van As, 1985 on the skin of ***Enteromius trimaculatus*** (South Africa), *E. marequensis*, *E. paludinosus*, *E. unitaeniatus*, *Labeo capensis*, *L. cylindricus*, *Mesobola brevianalis*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander*

Apiosoma piscicola Blanchard, 1885 on the skin and gills of *Coptodon rendalli*, *C. zillii*, *Enteromius paludinosus*, *E. trimaculatus*, *Labeo cylindricus*, *Lates niloticus*, *Marcusenius macrolepidotus*, *Micropterus dolomieu*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander*

Apiosoma poteriformis (Timofeev, 1962) [syn. *Glossatella poteriformis* Timofeev, 1962] on the gills of *Lates niloticus*

Apiosoma viridis Viljoen et Van As, 1985 on the skin of *Chetia flaviventris*, *Coptodon rendalli*, *Oreochromis mossambicus*, ***Pseudocrenilabrus philander*** (South Africa), *Tilapia sparrmanii*

Apiosoma sp. on the skin and gills of *Oncorhynchus mykiss*, *Synodontis schall*

Epistylis Ehrenberg, 1830

Epistylis transvaalensis Viljoen et Van As, 1983 on the skin of ***Pseudocrenilabrus philander*** (South Africa)

Epistylis sp. on the skin and gills of *Clarias gariepinus*, *Enteromius paludinosus*, *Oreochromis leucostictus*, *O. niloticus* [Fig. 4.2.5A]

Riboscyphidia Jankovski, 1985 (syn. *Scyphidia* Dujardin, 1841, *partim*)

Riboscyphidia doliaris (Chernova, 1977) [syn. *Scyphidia doliaris* Chernova, 1977] on the skin and gills of *Lates niloticus*

Riboscyphidia globularis (Solomatova, 1977) [syn. *Scyphidia globularis* Solomatova, 1977] on the skin and gills of *Lates niloticus*

Riboscyphidia mansourensis (El-Tantawy, Abdel-Aziz, Abou El-Nour, Samn, Shaldoum et Rady, 2016) [syn. *Scyphidia mansourensis* El-Tantawy, Abdel-Aziz, Abou El-Nour, Samn, Shaldoum et Rady, 2016] on the skin and gills of ***Lates niloticus*** (Egypt)

Riboscyphidia sp. on the gills and skin of *Chrysichthys auratus*, *Lates niloticus* [Fig. 4.2.5C]

Vorticella Linnaeus, 1767

Vorticella sp. on the gills and skin of *Clarias gariepinus*, *Sarotherodon galilaeus* [Fig. 4.2.5E]

Oligohymenophorea de Puytorac *et al.*, 1974 – **Peritrichia** Stein, 1859 (order Mobilida Kahl, 1933)

List of trichodinids (Mobilida) in African freshwater fishes

Hemitrichodina Basson et Van As, 1989

Hemitrichodina robusta Basson et Van As, 1989 on the skin and fins, occasionally gills of *Hepsetus cuvieri*, ***Marcusenius macrolepidotus*** (South Africa), *Micralestes acutidens* [Fig. 4.2.6A]

Paratrichodina Lom, 1963

Paratrichodina africana Kazubski et El-Tantawy, 1986 on the gills, rarely body surface of *Lates niloticus*, ***Oreochromis niloticus*** (Egypt), *Sarotherodon galilaeus*, *Tilapia* sp. [Fig. 4.2.6B]

Trichodina Ehrenberg, 1838

Trichodina acuta Lom, 1961 on the skin of *Oncorhynchus mykiss*

Trichodina anabantidarum Basson et Van As, 2002 on the gills, sometimes skin and fins of *Ctenopoma multispine*, ***Microctenopoma intermedium*** (Botswana)

Trichodina centrostrigeata Basson, Van As et Paperna, 1983 on the gills, sometimes skin and fins of *Brycinus lateralis*, *Coptodon rendalli*, *Cyprinus carpio*, *Enteromius* sp., *Hemichromis elongatus*, *Labeo cylindricus*, *Lates niloticus*, *Oreochromis andersonii*, *O. mossambicus*, *O. niloticus*, ***Pseudocrenilabrus philander*** (South Africa), *Serranochromis angusticeps*, *Synodontis leopardinus*, *Tilapia sparrmanii*

Trichodina compacta Van As et Basson, 1989 on the skin and fins of *Chetia flaviventris*, *Chiloglanis pretoriae*, ***Coptodon rendalli*** (South Africa)**,** *Cyprinus carpio*, *Enteromius eutaenia*, *E. radiatus*, *E. trimaculatus*, *Labeo cylindricus*, *Labeobarbus kimberleyensis*, *L. marequensis*, *Lates niloticus*, *Marcusenius macrolepidotus*, *Mesobola brevianalis*, *Micropanchax johnstoni*, *Nannocharax multifasciatus*, *Oreochromis andersonii*, *O. mossambicus*, *Petrocephalus catostoma*, *Pharyngochromis darlingi*, *Pseudocrenilabrus philander*, *Sarotherodon galilaeus*, *Serranochromis angusticeps*, *Tilapia sparrmanii*

Trichodina equatorialis Kazubski, 1986 on the gills of ***Tilapia* sp**. (Kenya)

Trichodina fahaka Al-Rasheid, Ali, Sakran, Abdel-Baki et Abdel Ghaffar, 2000 on the gills of ***Tetraodon lineatus*** [syn. *Tetraodon fahaka*] (Egypt)

Trichodina frenata Van As et Basson, 1992 on the gills of *Lates niloticus*, ***Mastacembelus frenatus*** (Namibia), *Sarotherodon galilaeus*

Trichodina heterodentata Duncan, 1977 on the skin, fins and gills of *Chetia flaviventris*, *Coptodon rendalli*, *Cyprinus carpio*, *Enteromius eutaenia*, *E. paludinosus*, *E. trimaculatus*, *Glossogobius giuris*, *Hydrocynus forskahlii*, *Labeo cylindricus*, *Labeobarbus marequensis*, *Marcusenius macrolepidotus*, *Mesobola brevianalis*, *Micralestes acutidens*, *Micropanchax johnstoni*, *Micropterus salmoides*, *Oreochromis* *mossambicus*, *O. niloticus, Petrocephalus catostoma*, *Pseudocrenilabrus philander*, *Synodontis zambezensis*, *Tilapia sparrmanii*

Trichodina kalimbeza Van As et Basson, 1992 on the skin and fins of ***Enteromius fasciolatus*** (Namibia)

Trichodina kazubski Van As et Basson, 1989 on the skin, fins and gills of ***Enteromius paludinosus*** (South Africa), *E. trimaculatus*

Trichodina kwando Van As et Basson, 1992 on the gills, rarely skin and fins of ***Brycinus lateralis*** (Namibia), *Micralestes acutidens*

Trichodina labyrinthipicis Basson et Van As, 2002 on the gills, rarely skin of *Ctenopoma multispine*, ***Microctenopoma intermedium*** (Botswana)

Trichodina lepsii Lom, 1962 from *Lates niloticus*

Trichodina linyanta Van As et Basson, 1992 on the skin and gills of *Hemichromis elongatus*, ***Oreochromis andersonii*** (Namibia)

Trichodina magna Van As et Basson, 1989 on the skin and fins, occasionally gills of *Clarias gariepinus*, ***Coptodon rendalli*** (South Africa), *C. zillii*, *Hepsetus cuvieri*, *Lates niloticus*, *Marcusenius macrolepidotus*, *Mesobola brevianalis*, *Micropanchax johnstoni*, *Micropterus salmoides*, *Oreochromis andersonii*, *O. mossambicus*, *O. niloticus*, *Petrocephalus catostoma*, *Pseudocrenilabrus philander*, *Sarotherodon galilaeus*, *Schilbe mystus*, *Serranochromis angusticeps*, *Tilapia sparrmanii* [Fig. 4.2.6C]

Trichodina maritinkae Basson et Van As, 1991 on the gills of *Clarias stappersii*, ***C. gariepinus*** (South Africa), *Heterobranchus longifilis*

Trichodina matsu Basson et Van As, 1994 on the gills of *Clarias gariepinus*

Trichodina microspina Van As et Basson, 1992 on the skin and fins, occasionally gills of ***Ctenopoma multispine*** (Namibia), *Microctenopoma intermedium*

Trichodina minuta Basson, Van As et Paperna, 1983 from the skin, fins and gills of *Enteromius trimaculatus*, *Hemichromis elongatus*, ***Oreochromis mossambicus*** (South Africa), *Pseudocrenilabrus philander*, *Tilapia sparrmanii*

Trichodina mutabilis Kazubski et Migala, 1968 on the skin and gills of *Carassius auratus*, *Oreochromis niloticus*

Trichodina ngoma Van As et Basson, 1992 on the skin, fins and gills of ***Nannocharax multifasciatus*** (Namibia)

Trichodina nigra Lom, 1961 on the skin, fins and gills of *Enteromius paludinosus*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii*

Trichodina nkasa Van As et Basson, 1992 on the gills of *Synodontis leopardinus*, ***S. macrostigma*** (Namibia)

Trichodina nobilis Chen, 1963 on the skin and gills of *Cyprinus carpio*

Trichodina rectuncinata Raabe, 1958 from *Clarias gariepinus*, *Lates niloticus*, *Oreochromis niloticus*

Note: this species was probably misidentified because it is a marine trichodinid, described from various parts of the world from the gills of various marine fish hosts. The identification of this record cannot be verified as none of the authors provided any morphological data or micrographs.

Trichodina reticulata Hirschmann et Partsch, 1955 on the skin and gills of *Carassius auratus*, *Cyprinus carpio*, *Oreochromis niloticus*

Trichodina sangwala Van As et Basson, 1992 on the gills of *Clarias gariepinus*, ***Schilbe mystus*** (South Africa)

Trichodina uniforma Van As et Basson, 1989 on the skin, fins and gills of ***Carassius auratus*** (South Africa)

Trichodina uretra Basson, 1989 in the urinary bladder and ureters of ***Enteromius trimaculatus*** (South Africa)

Trichodina sp. on the skin and gills of *Clarias gariepinus*, *Coptodon zillii*, *Cyprinus carpio*, *Enteromius paludinosus*, *Heterobranchus bidorsalis*, *H. longifilis*, *Oncorhynchus mykiss*, *Oreochromis leucostictus*, *O. niloticus*, *Protopterus annectens*, *Salmo trutta*, *Synodontis schall*

Trichodinella Raabe, 1950

Trichodinella crennulata Basson et Van As, 1987 on the gills of ***Micralestes acutidens*** (South Africa)

Trichodinella epizootica (Raabe, 1950) on the gills of *Anguilla anguilla*, *Coptodon rendalli*, *C. zillii*, *Ctenopharyngodon idella*, *Cyprinus carpio*, *Enteromius paludinosus*, *E. trimaculatus*, *Mesobola brevianalis*, *Mormyrus kannume*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander* [Fig. 4.2.6D]

Trichodinella sp. on the skin and gills of *Coptodon zillii*, *Cyprinus carpio*, *Enteromius paludinosus*, *Oreochromis leucostictus*

Tripartiella Lom, 1959

Tripartiella clavodonta Basson et Van As, 1987 on the gills of *Mesobola brevianalis*, ***Oreochromis mossambicus*** (South Africa), *Pseudocrenilabrus philander*

Tripartiella ctenopomae Basson et Van As, 2002 on the gills of ***Ctenopoma multispine*** (Botswana) [Fig. 4.2.6E]

Tripartiella dactylodentata Al-Rasheid, Ali, Sakran, Abdel-Baki et Ghaffar, 2000 on the gills of ***Mormyrus kannume*** (Egypt)

Tripartiella lechridens Basson et Van As, 1987 on the gills of *Cyprinus carpio*, *Enteromius paludinosus*, *E. trimaculatus*, ***Labeo cylindricus*** (South Africa), *Mesobola brevianalis*, *Micralestes acutidens*, *Oreochromis mossambicus*

Tripartiella leptospina Basson et Van As, 1987 on the gills of ***Oreochromis mossambicus*** (South Africa)

Tripartiella macrosoma Basson et Van As, 1987 on the gills of ***Enteromius eutaenia*** (South Africa)

Tripartiella microctenopomae Basson et Van As, 2002 on the gills of ***Microctenopoma intermedium*** (Botswana)

Tripartiella nana Basson et Van As, 1987 on the gills of ***Oreochromis mossambicus*** (South Africa)

Tripartiella orthodens Basson et Van As, 1987 on the gills of ***Coptodon rendalli*** (South Africa), *Sarotherodon galilaeus*

EXCAVATA Cavalier-Smith, 2002

Diplomonadida Wenyon, 1926 – **Hexamitinae** Kent, 1880

Diplomonadids – basic characteristics

- with a pair of kinetids and two nuclei, each kinetid usually with four kinetosomes and flagella (sometimes three or two), or uncommonly, one kinetid and nucleus
- with a pair of kinetids and two nuclei, each kinetid usually with four kinetosomes and flagella (sometimes three or two), or uncommonly, one kinetid and nucleus
- at least one flagellum per kinetid directed posteriorly, associated with a cytopharyngeal tube or groove, or lying axially within the cell
- various non-microtubular fibres supporting nucleus and cytopharyngeal apparatus
- free-living or endobiotic, often parasitic
- with functional feeding apparatuses
- with an alternate genetic code (TAR codon for glutamine)

List of diplomonadids from African freshwater fishes

Hexamita Dujardin, 1838

Hexamita africanus Imam, Ramadan et Derahli, 1987 from ***Synodontis schall*** (Egypt)

Hexamita sp. in the stomach and intestine of *Clarias gariepinus*, *Coptodon rendalli*, *Heterobranchus longifilis*, *Oreochromis niloticus*, *Sarotherodon galilaeus*, *Synodontis schall* [Fig. 4.2.5F]

Euglenozoa Cavalier-Smith, 1981 – **Prokinetoplastina** Vickerman in Moreira *et al.*, 2004

Prokinetoplastins – basic characteristics

- cells with two (occasionally one, rarely more) flagella, inserted into an apical/subapical flagellar pocket

- with rare exception, emergent flagella with paraxonemal rods
- usually with tubular feeding apparatus associated with flagellar apparatus
- basic flagellar apparatus pattern consisting of two functional kinetosomes and three asymmetrical arranged microtubular roots
- mostly with discoidal cristae

List of prokinetoplastins from African freshwater fishes

Ichthyobodo Pinto, 1928

Ichthyobodo necator (Henneguy, 1883) on the skin and gills of *Cyprinus carpio*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii* [Fig. 4.2.5G]

Ichthyobodo sp. on the skin and gills of *Clarias gariepinus*, *Cyprinus carpio*, *Heterobranchus longifilis*, *Labeobarbus* sp., *Oreochromis niloticus*, *Synodontis schall*

Euglenozoa Cavalier-Smith, 1981 **– Metakinetoplastina** Vickerman in Moreira *et al.*, 2004

List of metakinetoplastins from African freshwater fishes

Cryptobia Leidy, 1846

Cryptobia iubilans Nohynková, 1948 in the stomach and intestine of *Clarias gariepinus*, *Heterobrachus longifilis*

Cryptobia sp. in the intestine, liver, gills and blood of *Clarias gariepinus*, *Coptodon rendalli*, *Enteromius paludinosus*, *Oreochromis niloticus*, *Sarotherodon galilaeus*, *Synodontis schall* [Fig. 4.2.5H]

Trypanosoma Gruby, 1843

Trypanosoma alhussaini Mohamed, 1978 from ***Clarias gariepinus*** (Egypt)

Trypanosoma cyanophilum Mohamed, 1978 from ***Coptodon zillii*** (Egypt)

Trypanosoma mansouri Mohamed, 1978 from ***Coptodon zillii*** (Egypt)

Trypanosoma cf. *mugilicola* Becker et Overstreet, 1979 from *Mugil cephalus*

Note: this fish inhabits marine, freshwater and brackish water systems.

Trypanosoma mukasai Hoare, 1932 from *Astatoreochromis alluaudi*, *Bagrus docmak*, *Clarias gariepinus*, *C. theodorae*, *Haplochromis cinereus*, *H. humilior*, *H. nubilus*, *H. serranus*, ***Haplochromis* spp**. (Uganda), *Mormyrus kannume*, *Oreochromis andersonii*, *O. esculentus*, *O. mossambicus*, *O. niloticus*, *O. variabilis*, *Parauchenoglanis ngamensis*, *Schilbe intermedius*, *Serranochromis angusticeps*, *S. macrocephalus*, *S. robustus*, *Synodontis nigromaculatus*, *S. vanderwaali*, *Tilapia sparrmanii* [Fig. 4.2.3D]

Trypanosoma tobeyi Dias, 1952 from ***Clarias angolensis*** (Mozambique)

Trypanosoma toddi Bouet, 1909 from *Clarias angolensis*, ***C. anguillaris*** (French West Africa)

Trypanosoma sp. from *Coptodon zillii* (Egypt)

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Chapter 4.3.

MYXOZOA

Ivan FIALA & Pavla BARTOŠOVÁ-SOJKOVÁ

Myxozoa – basic characteristics, life cycles, classification and principal diagnostic features

- parasitic cnidarians (Cnidaria: Myxozoa) with about 2,400 species classified in 67 genera with a worldwide distribution
- endoparasites of fish, annelids and bryozoans, less frequently in amphibians and reptiles; exceptionally in birds, mammals and sipunculids
- two-host life cycle: an invertebrate (annelid or bryozoan) definitive host produces actinospores or malacospores and a vertebrate (mostly fish) intermediate host produces myxospores or fish malacospores (Fig. 4.3.1)
- a vertebrate-derived spore consists mostly of two valves, 1-4 polar capsules and an infectious sporoplasm
- spores microscopic (typically 10-20 μm in size)
- spores develop in vegetative stages (trophozoites, plasmodia) which are histozoic (between tissue cells, *e.g.*, muscles, gills, skin, brain, kidney, liver and spleen) or coelozoic (in cavities, *e.g.*, gall bladder, urinary bladder, lumen of renal tubules and renal corpuscles)
- vegetative stages vary greatly in dimensions, histozoic ones may be macroscopic (up to several cm in diameter)
- trophozoites characterised by cell-within-cell organisation
- species belonging to *Myxobolus* are the most common freshwater myxozoans
- myxozoans from freshwater fishes cluster within the malacosporean clade (*Buddenbrockia*, *Tetracapsuloides*), *Sphaerospora* sensu stricto clade (*Sphaerospora*) and the freshwater (oligochaete) myxosporean lineage (all other genera, *e.g.*, *Myxobolus*).
- causative agents of serious fish diseases, *e.g.*, whirling disease and proliferative kidney disease (PKD)

The classification of the Myxozoa is based on myxospore morphology. The shape of the spore, number of shell valves and polar capsules and the position of the polar capsules within the spore are the most important features for the definition of myxozoan genera (Fiala *et al.* 2015; Fig. 4.3.2). Classification at the species level is based on other spore characteristics such as spore and polar capsule dimensions, spore surface structures, the number of polar filament coils, etc.

The subphylum Myxozoa consists of two classes, the Malacosporea (*Buddenbrockia* and *Tetracapsuloides*) and the Myxosporea with two orders, Bivalvulida and Multivalvulida (mostly marine species, *e.g.*, *Kudoa*). Bivalvulida includes two suborders: Variisporina (*e.g.*, *Myxidium*, *Zschokkella*, *Sphaerospora*, *Hoferellus*, *Chloromyxum* and *Myxobilatus*) and Platysporina (*e.g.*, *Myxobolus*, *Henneguya*, *Thelohanellus* and *Unicauda*) (Fig. 4.3.2).

Key to the genera of the Myxozoa from freshwater fishes (modified from Fiala *et al.* 2015)

The taxonomic key includes myxozoan genera reported from freshwater fishes from all continents (Lom & Dyková 2006) with the genera previously reported in Africa in bold.

1 (2) Spores with soft (unhardened) shell valves (Malacosporea)……………….3

2 (1) Spores with hardened shell valves (Myxosporea)…………………………. 5

3 (4) Fish malacospores with two shell valves, two spherical polar capsules, one sporoplasm; bryozoan-related trophozoites sac- or worm-like; sacs irregularly shaped, elongate, ellipsoid or constricted; myxoworms with triploblast organisation………………………………………..*Buddenbrockia*

4 (3) Fish malacospores with two shell valves, two spherical polar capsules, one sporoplasm; bryozoan-related stages mostly sac-like of regular spherical shape…………………………………………………*Tetracapsuloides*

5 (6) Mature spore contains only one polar capsule………………………………7

6 (5) Mature spore contains two or more polar capsules…………………………11

7 (8) Spore with a bifurcate caudal process………………………….*Phlogospora*

8 (7) Spore without a caudal process……………………………………………9

9 (10) Spores with polar capsule discharging apically and axially [Fig. 4.3.2B] …………………………………………………………….***Thelohanellus***

10 (9) Spores with polar capsule discharging subapically and to the side ………………………………………………………….*Neothelohanellus*

11 (12) Mature spore contains two polar capsules…………………………………13

12 (11) Mature spore contains four or more polar capsules…………………………51

13 (14) Polar capsules set apart from each other…………………………………15

14 (13) Polar capsules located close to each other……………………………….25

15 (16) Polar capsules each located separately at spore ends…………………...17

16 (15) Polar capsules located not terminally and set widely apart in the sutural plane…………………………………………………………………………...19

17 (18) Spores fusiform, straight or slightly crescent or sigmoid-shaped with more or less pointed ends, usually pyriform polar capsules; mostly coelozoic [Fig. 4.3.2K]………………………………………………………… ***Myxidium***

18 (17) Spores usually ellipsoidal, slightly bent or semicircular, with rounded or bluntly pointed ends and almost spherical polar capsules; mostly coelozoic [Fig. 4.3.2L]……………………………………………………… ***Zschokkella***

19 (20) Spores spherical or subspherical [Fig. 4.3.2F]…………………***Ortholinea***

20 (19) Spores ovoid or triangular………………………………………………... 21

21 (22) Spores triangular with rounded corners, flattened parallel to sutural plane, without projections [Fig. 4.3.2G]…………………………………***Triangula***

22 (21) Spores ovoid…………………………………………………………………..23

23 (24) Spores flattened parallel to the sutural plane without sutural markings ……………………………………………………………….. *Neomyxobolus*

24 (23) Spores spindle-shaped in sutural view with sutural markings along the posterior border……………………………………………*Cardimyxobolus*

25 (26) Spores asymmetrical with two caudal projections……………*Hennegoides*

26 (25) Spores bilaterally symmetrical…………………………………………….27

27 (28) Polar capsules at distance from the spore apex…………………………. 29

28 (27) Polar capsules in the apex of the spore………………………………….31

29 (30) Spindle-shaped spores with two spherical polar capsules positioned in tandem at a distance from the anterior end and two projections at both spore ends…………………………………………………..*Neohenneguya*

30 (29) Large spherical polar capsules in the centre of an oval spore in valvular view and with triangular shape in sutural view………………………*Wardia*

31 (32) Polar capsules set in a plane perpendicular to the sutural line……………33

32 (31) Polar capsules set in the sutural plane……………………………………..39

33 (34) Spores without projections…………………………………………………..35

34 (33) Spores with projections……………………………………………………37

35 (36) Spores spherical, subspherical or slightly elongate in the direction perpendicular to the sutural plane; mostly coelozoic in the excretory system [Fig 4.3.2E]……………………………………………………..***Sphaerospora***

36 (35) Spores pyriform or mitre-like with ridged valves…………………….*Acauda*

37 (38) Spores spindle-shaped, with a pair of long posterior projections [Fig. 4.3.2I] ……………………………………………………………….***Myxobilatus***

38 (37) Spores pointed, mitre-like or rounded in valvular view with numerous stiff filaments at the posterior end [Fig. 4.3.2J]……………………….***Hoferellus***

39 (40) Spores without projections…………………………………………………...41

40 (39) Spores with projections……………………………………………………43

41 (42) Sutural line strongly sinuous…………………………………..*Spirosuturia*

42 (41) Sutural line straight, spores ellipsoidal, ovoid or rounded [Fig. 4.3.2A] ……………………………………………………………… ***Myxobolus***

43 (44) Spores with a single caudal projection [Fig. 4.3.2C]…………….***Unicauda***

44 (43) Spores with more than one caudal projection……………………………45

45 (46) Spores with two caudal projections………………………………………47

46 (45) Spores with four posterolateral projections…………………..*Tetrauronema*

47 (48) Spores with two laterally extending projections…………………………49

48 (47) Spores with two slightly divergent projections [Fig. 4.3.2D]…..***Henneguya***

49 (50) Lateral projections extend from one side of the posterior spore end ……………………………………………………………*Laterocaudata*

50 (49) Lateral projections extend in opposite directions…………………...*Dicauda*

51 (52) Spores with two shell valves……………………………………………53

52 (51) Spores with four shell valves………………………………………….. 55

53 (54) Spores spherical [Fig. 4.3.2H]……………………………….***Chloromyxum***

54 (53) Spores almost spherical with one or two caudal projections………………………………………………………… *Caudomyxum*

55 (56) Spores stout spindle-shaped with the sutural ridge extending both spore ends as a spine, coelozoic……………………………………… *Octospina*

56 (55) Spores stellate, quadrate, subspherical to ovoid in apical view; histozoic [Fig. 4.3.2M]…………………………………………………………..***Kudoa***

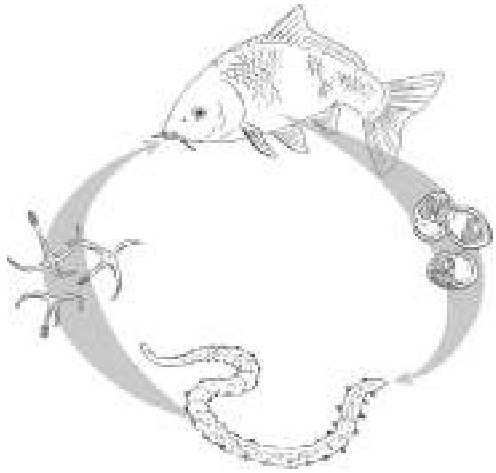

Fig. 4.3.1. Myxozoan life cycle with an annelid definitive host releasing actinospores and a fish intermediate host releasing myxospores. (Illustration by M. Luo.)

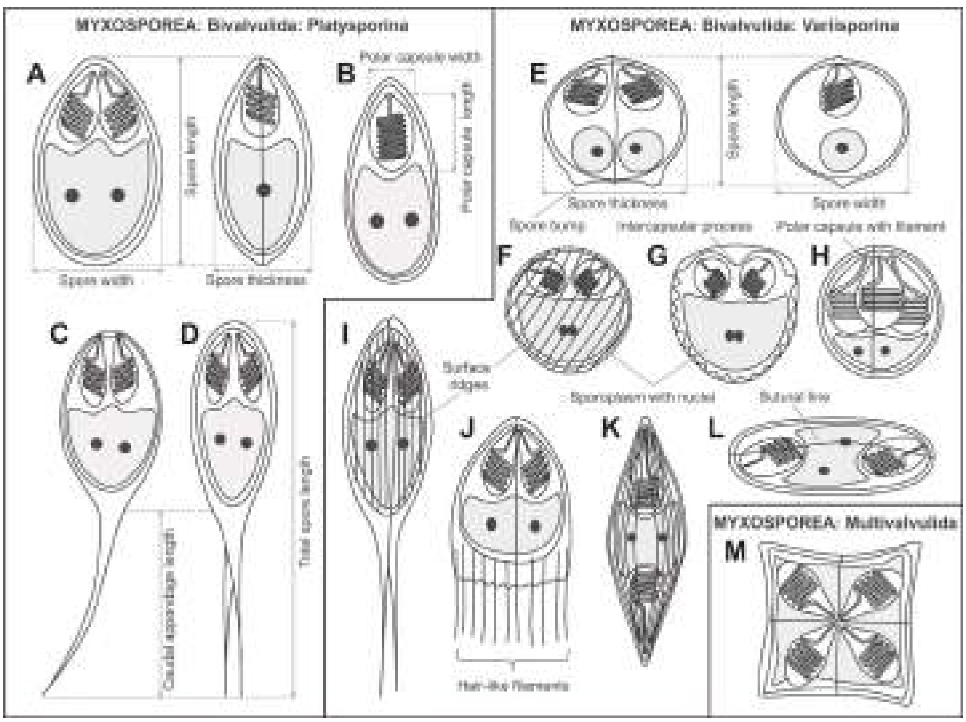

Fig. 4.3.2. Schematic line drawings of myxospores representing myxosporean genera reported from African freshwater fishes with instructions on spore measurements and indicating the most important spore features. **A.** *Myxobolus*, frontal and sutural view; **B.** *Thelohanellus*; **C.** *Unicauda*; **D.** *Henneguya*; **E.** *Sphaerospora*, sutural and lateral view; **F.** *Ortholinea*; **G.** *Triangula*; **H.** *Chloromyxum*; **I.** *Myxobilatus*; **J.** *Hoferellus*; **K.** *Myxidium*; **L.** *Zschokkella*; **M.** *Kudoa*.

List of the Myxozoa in African freshwater fishes

Species are listed alphabetically according to individual myxozoan genera with information about infection site and type host species (in bold) and country of origin if described from Africa. The systematic survey is based on Fomena & Bouix (1997), Eiras (2002), Eiras *et al.* (2005, 2011, 2012, 2014), Abdel-Ghaffar *et al.* (2008), Eiras & Adriano (2012), Zhang *et al.* (2013), and Alama-Bermejo *et al.* (2016).

Cnidaria Hatschek, 1888

MYXOZOA Grassé, 1970

Myxosporea Bütschli, 1881

Bivalvulida Shulman, 1959

PLATYSPORINA Kudo, 1919

Henneguya Thélohan, 1892

Henneguya auchenoglanii Kostoïngué, Diebakate, Faye et Toguebaye, 2001 in the base of primary gill lamellae of ***Auchenoglanis occidentalis*** (Chad)

Henneguya bopeleti Fomena et Bouix, 1987 in the gills of ***Chrysichthys nigrodigitatus*** (Cameroon)

Henneguya branchialis Ashmawy, Abu-Elwafa, Imam et El-Otifi, 1989 in the gills and intestine of *Clarias anguillaris*, ***C. gariepinus*** (Egypt), *Coptodon zillii*, *Sarotherodon galilaeus*

Henneguya camerounensis Fomena et Bouix, 1987 in the gills of *Schilbe multitaeniatus*, ***Synodontis batesii*** (Cameroon)

Henneguya chrysichthyi Obiekezie et Enyenihi, 1988 in the gills of ***Chrysichthys nigrodigitatus*** (Nigeria)

Henneguya clariae Abolarin, 1971 in the gills of ***Clarias gariepinus*** (Nigeria)

Henneguya ctenopomae Fomena et Bouix, 1996 in the gills of ***Microctenopoma nanum*** (Cameroon)

Henneguya dini Kabre, Sakiti, Marquès et Sawadogo, 1997 in the gills of ***Heterotis niloticus*** (Burkina Faso)

Henneguya fusiformis Kostoïngué, Fall, Faye et Toguebaye, 1999 in the gills of ***Clarias anguillaris*** (Chad), *C. gariepinus*.

Henneguya ghaffari Ali, 1999 in the intestine, pyloric caeca and gills of ***Lates niloticus*** (Egypt)

Henneguya laterocapsulata Landsberg, 1987 in the skin of *Clarias gariepinus*, ***C. gariepinus* × *Heterobranchus bidorsalis***

Henneguya logonensis Kostoïngué, Diebakate, Faye et Toguebaye, 2001 in the primary gill lamellae of ***Citharinus citharus*** (Chad)

Henneguya mailaoensis Kostoïngué, Diebakate, Faye et Toguebaye, 2001 in the primary gill lamellae of ***Mormyrus caschive*** (Chad)

Henneguya malapteruri Fomena et Bouix, 1996 in the skin and muscles of ***Malapterurus electricus*** (Cameroon)

Henneguya mandouri Rabie, Mohammed, Hussein et Hussein, 2009 in the middle and base of gill filaments of ***Lates niloticus*** (Egypt)

Henneguya maraensis Kostoïngué, 1997 in the gills and intestine of ***Lates niloticus*** (Chad)

Henneguya massii Kostoïngué, Diebakate, Faye et Toguebaye, 2001 in the primary gill lamellae of ***Lates niloticus*** (Chad)

Henneguya mbakaouensis Fomena et Bouix, 2000 in the gills of ***Lates niloticus*** (Cameroon)

Henneguya mormyri Kostoïngué, Diebakate, Faye et Toguebaye, 2001 in the primary gill lamellae of ***Mormyrus caschive*** (Chad)

Henneguya nkamensis Fomena, Folefack et Bouix, 2008 in the secondary gill lamellae of ***Hepsetus odoe*** (Cameroon)

Henneguya ntemensis Fomena et Bouix, 1996 in the gall bladder, kidney and spleen of ***Brienomyrus brachyistius*** (Cameroon)

Henneguya ntondei Fomena, Folefack et Bouix, 2008 in the secondary gill lamellae of ***Schilbe mystus*** (Cameroon)

Henneguya nyongensis Fomena et Bouix, 1996 in the gills and muscles of ***Marcusenius moorii*** (Cameroon)

Henneguya odzai Fomena et Bouix, 1996 in the gills of *Brevimyrus niger*, *Hyperopisus bebe*, ***Marcusenius moorii*** (Cameroon), *M. senegalensis*, *Mormyrus rume*

Henneguya pethericii Fomena, Folefack et Bouix, 2008 in the secondary gill lamellae of ***Ctenopoma petherici*** (Cameroon)

Henneguya samochimensis Reed, Basson et Van As, 2003 in the primary gill filaments of ***Clarias gariepinus*** (Botswana)

Henneguya sarotherodoni Fall, Fomena, Kostoïngué, Diebakate, Faye et Toguebaye, 2000 in the intestine of ***Sarotherodon galilaeus*** (Chad)

Henneguya somahiensis Sakiti, 1997 in the gills of ***Ctenopoma kingsleyae*** (Benin)

Henneguya suprabranchiae Landsberg, 1987 in the accessory breathing organ of *Clarias anguillaris*, ***C. gariepinus***, *Oreochromis niloticus*

Myxobolus Bütschli, 1882

Myxobolus africanus Fomena, Bouix et Birgi, 1989 in the brain, gill adductor muscle, muscles of the operculum, bile duct and gall bladder wall of ***Hepsetus odoe*** (Cameroon)

Myxobolus agolus Landsberg, 1985 [syn. *Myxobolus melenensis* Fomena, Bouix et Birgi, 1985] in the kidney, spleen and gills of *Coptodon guineensis*, *Hemichromis fasciatus*, *Oreochromis niloticus*, *Sarotherodon galilaeus*

Myxobolus amieti Fomena, Bouix et Birgi, 1989 in the gills, eye, superficial mandibular muscles, muscles of the operculum and pharyngeal wall and connective tissue covering the gill arches of ***Microctenopoma nanum*** (Cameroon)

Myxobolus bagri Negm-Eldin, Govedich et Davies, 1999 in the gills of ***Bagrus bajad*** (Egypt)

Myxobolus beninensis Sakiti, Blanc, Marquès et Bouix, 1991 in the gills of ***Sarotherodon melanotheron*** (Benin)

Myxobolus bilongi Fomena, Marquès, Bouix et Njiné, 1994 in the gills and fins of ***Labeo* sp.** (Cameroon)

Myxobolus bizerti Bahri et Marquès, 1996 in the gills of ***Mugil cephalus*** (Tunisia)

Myxobolus bouixi Fomena, Folefack et Tang, 2007 in the gills of ***Chrysichthys nigrodigitatus*** (Cameroon)

Myxobolus brachysporus (Baker, 1963) in the spleen and kidney of *Coptodon guineensis*, ***Oreochromis esculentus*** (Uganda), *O. niloticus*, *O. niloticus* × *S. galilaeus*, *O. variabilis*, *Sarotherodon galilaeus*

Myxobolus branchiophilus Abdel-Ghaffar, El-Toukhy, Al-Quarishy, Al-Rashid, Abdel-Baki, Hegazy et Bashtar, 2008 in the gill filaments of ***Oreochromis niloticus*** (Egypt)

Myxobolus burkinei Kabré, 1995 in the gills and fins of ***Labeo coubie*** (Burkina Faso)

Myxobolus camerounensis Fomena, Marquès et Bouix, 1993 in the gills, eyes and muscles of ***Oreochromis niloticus*** (Cameroon)

Myxobolus caudatus Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002 in the tail and fins of ***Labeobarbus bynni*** (Egypt)

Myxobolus charii Fomena, 2004 in the skin of ***Citharinus citharus*** (Chad)

Myxobolus chrysichthyi Negm-Eldin, Govedich et Davies, 1999 in the gills of ***Chrysichthys auratus*** (Egypt)

Myxobolus clarias Negm-Eldin, Govedich et Davies, 1999 in the gills of ***Chrysichthys auratus*** (Egypt)

Myxobolus comoei Kabre, Sakiti, Marquès et Sawadogo, 1995 in the fins and gills of ***Clarias anguillaris*** (Burkina Faso)

Myxobolus dahomeyensis (Siau, 1971) in the ovaries of *Coptodon zillii*, *Oreochromis niloticus*, *O. mossambicus* × *O. niloticus*, *Sarotherodon melanotheron*, ***Synodontis ansorgii*** (Benin)

Myxobolus diamaensis Diamanka, Faye, Fall et Toguebaye, 2007 in the gill filaments of ***Sarotherodon melanotheron*** (Senegal)

Myxobolus distichodi Kostoingué et Toguebaye, 1994 in the gills, intestine and liver of ***Distichodus engycephalus*** (Chad)

Myxobolus djoudjensis Diamanka, Faye, Fall et Toguebaye, 2007 in the ovaries of ***Coptodon guineensis*** (Senegal)

Myxobolus dossoui Sakiti, Blanc, Marquès et Bouix, 1991 in the gill arches and cartilage of ***Coptodon zillii*** (Benin), *Hemichromis fasciatus*, *Oreochromis mossambicus* x *O. niloticus*

Myxobolus egypticus (Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002) [syn. *M. intestinalis* Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002] in the intestine of ***Labeobarbus bynni*** (Egypt)

Myxobolus equatorialis (Landsberg, 1985) in the spleen and kidney of *Coptodon guineensis, Oreochromis niloticus*, *Sarotherodon galilaeus*

Myxobolus etsataensis Reed, Basson et Van As, 2002 in the gills of ***Enteromius thamalakanensis*** (Botswana)

Myxobolus exiguous Thélohan, 1895 in the scales of *Chelon aurata*, *Mugil cephalus*

Myxobolus fahmii Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002 in the gills of ***Labeobarbus bynni*** (Egypt)

Myxobolus fobobi (Fomena, 1985) [syn. *Myxobolus barbi* Fomena, 1985] in the gills of ***Enteromius aspilus*** (Cameroon), *E. camptacanthus*, *E. jae*, *E. guirali*, *E. martorelli*

Myxobolus fomenai Abdel-Ghaffar, El-Toukhy, Al-Quarishy, Al-Rashid, Abdel-Baki, Hegazy et Bashtar, 2008 in the muscles, intestine and kidney of ***Oreochromis niloticus*** (Egypt)

Myxobolus fotoi Fomena, Marquès and Bouix, 1993 in the gills of ***Oreochromis niloticus*** (Cameroon)

Myxobolus galilaeus Landsberg, 1985 in the kidney, spleen, eyes, gills and intestine of *Coptodon guineensis*, *Oreochromis niloticus*, *O. niloticus* × *S. galilaeus*, ***Sarotherodon galilaeus***

Myxobolus gandiolensis Fall, Fomena, Kostoïngué, Diebakate, Faye et Toguebaye, 2000 in the kidney of ***Coptodon guineensis*** (Senegal)

Myxobolus gariepinus Reed, Basson et Van As, 2003 in the ovaries of ***Clarias gariepinus*** (Botswana)

Myxobolus heterosporus (Baker, 1963) [syn. *Myxosoma heterospora* Baker, 1963] in the kidney, liver, spleen, gills, intestine and gall bladder of *Coptodon zillii*, *Hemichromis fasciatus, Oreochromis niloticus*, *Sarotherodon melanotheron*

Myxobolus heterotisi Bongou, Kabré, Sakiti, Marquès et Sawadogo, 2006 in the primary gill filaments of ***Heterotis niloticus*** (Burkina Faso)

Myxobolus homeosporus (Baker, 1963) in the muscles and cornea of *Coptodon zillii*, ***Oreochromis esculentus*** (Uganda), *O. niloticus*, *O. variabilis*, *Sarotherodon galilaeus*

Myxobolus hydrocyni Kostoïngué et Toguebaye, 1994 in the gills of ***Hydrocynus forskahlii*** (Chad)

Myxobolus imami Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002 in the kidney of ***Labeo niloticus*** (Egypt), *Labeobarbus bynni*

Myxobolus israelensis Landsberg, 1985 in the kidney, spleen and gills of *Coptodon cameronensis*, *C. guineensis*, *Oreochromis niloticus*, ***O. niloticus*** × ***O. aureus***, *Sarotherodon galilaeus*, *S. mvogoi*

Myxobolus kainjiae (Obiekezie et Okaeme, 1990) [syn. *M. ovariae* Paperna, 1973] in the ovaries and urinary bladder of *Coptodon nyongana*, ***Haplochromis angustifrons*** (Uganda), *H. elegans*, *Oreochromis niloticus*, *Sarotherodon galilaeus*

Myxobolus kouoptamoensis Nchoutpouen et Fomena, 2011 in the gills, spleen and kidney of ***Labeo parvus*** (Cameroon)

Myxobolus kribiensis Fomena et Bouix, 1994 in the skin, eye sclera and kidney of ***Brycinus longipinnis*** (Cameroon)

Myxobolus labeoi Boungou, Kabré, Sakiti, Marquès et Sawadogo, 2006 in the fin rays of ***Labeo coubie*** (Burkina Faso)

Myxobolus labiae Negm-Eldin, Govedich et Davies, 1999 in the gills of ***Labeo niloticus*** (Egypt)

Myxobolus latesi Kostoïngué et Toguebaye, 1994 in the gills and intestine of ***Lates niloticus*** (Chad)

Myxobolus latis Negm-Eldin, Govedich et Davies, 1999 in the gills of ***Lates niloticus*** (Egypt)

Myxobolus lazera (Mandour, Galal et Abed, 1993) [syn. *M. clarii* Mandour, Galal et Abed, 1993] in the testes of ***Clarias gariepinus*** (Egypt)

Myxobolus mbailaoi Fomena, 2004 in the operculum, skin and intestine of ***Citharinus citharus*** (Chad)

Myxobolus naffari Ghaffar, Ibrahiem, Bashtar et Ali, 1998 in the gills of ***Labeo niloticus*** (Egypt) and *Labeobarbus bynni*

Myxobolus nchoutnounensis Nchoutpouen et Fomena, 2011 in the gills, scales, liver, fins, spleen, kidney and eyes of ***Labeo parvus*** (Cameroon)

Myxobolus negmgoda (Negm-Eldin, Govedich et Davies, 1999) [syn. *M. synodontis* Negm-Eldin, Govedich et Davies, 1999] in the gills of ***Synodontis schall*** (Egypt)

Myxobolus ngassami Lekeufack Folefack, Defoueng et Fomena 2017 in the fins, operculum, skin and sclera of the eye of ***Enteromius callipterus*** (Cameroon)

Myxobolus nilei (Faisal et Shalaby, 1987) [syn. *Myxosoma tilapiae* Faisal et Shalaby, 1987] in the gills, skin, eyes, kidney and pancreas of ***Oreochromis niloticus*** (Egypt)

Myxobolus niloticus Fahmy, Mandour et El-Naffar, 1971 in the tail fin rays and operculum of ***Labeo niloticus*** (Egypt)

Myxobolus njinei Fomena, Bouix et Birgi, 1985 in the gill arch of ***Enteromius camptacanthus*** (Cameroon), *E. guirali*, *E. martorelli*

Myxobolus njoyai Nchoutpouen et Fomena, 2011 in the gills, scales, fins, spleen and kidney of ***Labeo parvus*** (Cameroon)

Myxobolus nkolyaensis Fomena et Bouix, 1994 in the gills and caudal muscles of ***Enteromius jae*** (Cameroon)

Myxobolus nokoueensis Sakiti, 1991 in the gills of ***Sarotherodon melanotheron*** (Benin)

Myxobolus nounensis Fomena et Bouix, 2000 in the kidney and spleen of ***Sarotherodon galilaeus*** (Cameroon)

Myxobolus nyongana (Fomena, Bouix et Birgi, 1985) [syn. *Myxobolus barbi* Fomena, Bouix et Birgi, 1985] in the gills and eyes of *Alestes dentex*, *Enteromius aspilus*, *E. camptacanthus*, *E. guirali*, ***E. jae*** (Cameroon), *E. martorelli*, *Labeo parvus*, *Sarotherodon melanotheron*

Myxobolus occularis Abu-El-Wafa, 1988 in the eyes of ***Tilapia* sp.** (Egypt)

Myxobolus oloi Fomena et Bouix, 1994 in the gill arch epithelium, kidney and heart of ***Enteromius aspilus*** (Cameroon), *E. camptacanthus*, *E. guirali, E. martorelli*

Myxobolus ovoidalis Fantham, 1930 in the subcutaneous tissue of *Barbus* sp., ***Cyprinus carpio*** (South Africa), *Salvelinus fontinalis*

Myxobolus paludinosus Reed, Basson et Van As, 2002 in the gills of ***Enteromius paludinosus*** (Botswana)

Myxobolus perforata Ali, Al-Rasheid, Sakran, Abdel-Baki et Abdel-Ghaffar, 2002 in the internal surface of operculum of ***Hydrocynus forskahlii*** (Egypt)

Myxobolus pethericii Fomena, Folefack et Tang, 2007 in the gills, fins, stomach wall, liver, small intestine, operculum and kidney of ***Ctenopoma petherici*** (Cameroon)

Myxobolus polycentropsi Fomena, Bouix et Birgi 1985 [syn. *M. microcapsularis* Sakiti, Blanc, Marquès et Bouix, 1991] in the gills arch cartilage of ***Polycentropsis abbreviata*** (Cameroon), *Coptodon zillii*

Myxobolus saintlouisiensis Diamanka, Faye, Fall et Toguebaye, 2007 in the gill filaments of ***Oreochromis niloticus*** (Senegal)

Myxobolus sanagaensis Lekeufack Folefack, Defoueng et Fomena 2017 in the heart auricles of ***Enteromius callipterus*** (Cameroon)

Myxobolus sangei Fomena, Folefack et Tang, 2007 in the gills, skin, kidney of ***Brycinus macrolepidotus*** (Cameroon)

Myxobolus sarigi (Landsberg, 1985) in the kidney and spleen of *Coptodon margaritacea*, *C. guineensis*, *Oreochromis niloticus*, *O. niloticus* × *Sarotherodon galilaeus*, *S. galilaeus*

Myxobolus sarotherodoni Sakiti, Blanc, Marquès et Bouix, 1991 in the gills of ***Sarotherodon melanotheron*** (Benin)

Myxobolus sessabai Lekeufack Folefack, Defoueng et Fomena, 2017 in the skin of ***Enteromius callipterus*** (Cameroon)

Myxobolus sheroidalis Abu-El-Wafa, 1988 in the viscera of *Clarias* sp., ***Tilapia* sp.** (Egypt)

Myxobolus sourouensis Bongou, Kabré, Sakiti, Marquès et Sawadogo, 2006 in the primary gill filaments of ***Heterotis niloticus*** (Burkina Faso)

Myxobolus stenosus Paperna, 1973 in the gills and kidney of *Synodontis clarias*, ***S. schall*** (Uganda)

Myxobolus synodonti Fomena, Bouix et Birgi, 1985 in the stomach wall of ***Synodontis batesii*** (Cameroon)

Myxobolus tilapiae Abolarin, 1974 in the buccal cavity, gills, fins, kidney and spleen of *Coptodon margaritacea*, *C. rendalli*, *C. zillii*, ***Oreochromis niloticus*** (Nigeria), *Sarotherodon galilaeus*, *S. mvogoi*

Myxobolus tingrelaensi Bongou, Kabré, Sakiti, Marquès et Sawadogo, 2006 in the fin rays of ***Sarotherodon galilaeus*** (Burkina Faso)

Myxobolus zillii Sakiti, Blanc, Marquès et Bouix, 1991 [syn. *Myxobolus latesi* Kostoïngué et Toguebaye 1994] in the gills and intestine of ***C. zillii*** (Benin), *Lates niloticus*

Thelohanellus Kudo, 1933

Thelohanellus assambai Fomena, Marquès, Bouix et Njine, 1994 in the gills and fins of ***Labeo* sp.** (Cameroon)

Thelohanellus bicornei Kabre, Sakiti, Marquès et Sawadogo, 2002 in the gills of ***Labeo coubie*** (Burkina Faso)

Thelohanellus citharini Kostoïngué, Fall, Faye et Toguebaye, 1999 in the heart of ***Citharinus citharus*** (Chad)

Thelohanellus costeae Sakiti, 1997 in the gills of ***Labeo senegalensis*** (Benin)

Thelohanellus lagdoensis Fomena, Farikou-Oumarou, Tang et Bouix, 2007 in from the intestine of ***Citharinus citharus*** (Cameroon)

Thelohanellus ndjamenaensis Kostoïngué, Fall, Faye et Toguebaye, 1999 in the gills of ***Labeo parvus*** (Chad)

Thelohanellus niloticus Abdel-Ghaffar, Morsy, Bashtar, El-Ganainy et Gamal, 2013 [syn. *Myxobolus unicapsulatus* Gurley, 1893] in the gills of ***Labeo niloticus*** (Egypt)

Thelohanellus njinei Fomena, Farikou-Oumarou, Tang et Bouix, 2007 in the intestine of ***Schilbe mystus*** (Cameroon)

Thelohanellus rhabdalestus Azevedo, Samuel, Saveia, Delgado et Casal, 2011 in the liver and heart of ***Rhabdalestes maunensis*** (Angola)

Thelohanellus sanagaensis Fomena, Marquès, Bouix et Njine, 1994 in the gills and fins of ***Labeo* sp.** (Cameroon)

Thelohanellus taguii Fomena, Abakar-Ousman, Ngassam et Bouix, 2004 in the gills, liver, opercular muscles and intestine of ***Citharinus citharus*** (Chad)

Thelohanellus valeti Fomena et Bouix, 1987 in the stomach wall, gill filaments, muscles and operculum of *Enteromius aspilus*, ***E. jae*** (Cameroon), *Oreochromis niloticus*

Unicauda Davis, 1944

Unicauda strongylura (Gurley, 1893) [syn. *Henneguya strongylura* (Gurey, 1893) Labbé, 1899] in the tissues of ***Synodontis schall*** (Egypt)

VARIISPORINA Lom et Noble, 1984

Chloromyxum Mingazzini, 1890

Chloromyxum alii Abdel-Baki, 2007 in the gall bladder of ***Schilbe mystus*** (Egypt)

Chloromyxum birgii Fomena et Bouix, 1994 in the gall bladder of *Amphilius longirostris*, *Enteromius aspilus*, ***E. martorelli*** (Cameroon)

Chloromyxum vanasi Ali, 1998 in the gall bladder of ***Bagrus bajad*** (Egypt)

Hoferellus Berg, 1898

Hoferellus gnathonemi Alama-Bermejo, Jirků, Kodádková, Pecková, Fiala et Holzer, 2016 in the kidney of ***Gnathonemus petersii*** (Nigeria)

Myxidium Bütschli, 1882

Myxidium beninensis Sakiti, 1997 in the gall bladder of *Chrysichthys auratus*, ***C. nigrodigitatus*** (Benin)

Myxidium birgii Fomena et Bouix, 1986 in the gall bladder of ***Aphyosemion bivittatum*** (Cameroon)

Myxidium bouixi Siau, 1971 in the gall bladder of ***Synodontis ansorgii*** (Benin)

Myxidium brienomyri Fomena et Bouix, 1986 in the gall bladder of ***Brienomyrus brachyistius*** (Cameroon)

Myxidium camerounense Fomena et Bouix, 1986 in the gall bladder of ***Neolebias ansorgii*** (Cameroon)

Myxidium distichodi Kostoïngué, Faye et Toguebaye, 1998 in the gall bladder of ***Distichodus engycephalus*** (Chad), *Parachanna obscura*

Myxidium latesi Kostoïngué, Faye et Toguebaye, 1998 in the gall bladder of ***Lates niloticus*** (Chad)

Myxidium mendehi Fomena et Bouix, 1994 in the kidney of ***Enteromius guirali*** (Cameroon), *E. martorelli*

Myxidium nkamense Fomena, Folefack et Bouix, 2010 in the gall bladder of ***Clarias pachynema*** (Cameroon)

Myxidium nyongense Fomena et Bouix, 1986 in the gall bladder of *Enteromius aspilus*, *E. camptacanthus*, *E. guirali*, ***E. jae*** (Cameroon), *E. martorelli*

Myxidium parachannae Sakiti, 1997 in the gall bladder of ***Parachanna obscura*** (Benin)

Myxidium petrocephali Fomena et Bouix, 1986 in the gall bladder of *Ctenopoma petherici*, ***Petrocephalus simus*** (Cameroon)

Myxidium sangei Fomena, Folefack et Bouix, 2010 in the gall bladder of ***Parachanna obscura*** (Cameroon)

Myxidium schalli Abdel Ghaffar, El-Shahawi et Naas, 1995 in the gall bladder of ***Synodontis schall*** (Egypt)

Myxidium schilba Ali, Sakran et Abdel-Baki, 1999 in the gall bladder of ***Schilbe mystus*** (Egypt)

Myxidium shamama Ali, Sakran et Abdel-Baki, 1999 in the kidney of ***Labeo niloticus*** (Egypt)

Myxobilatus Davis, 1944

Myxobilatus accessobranchialis Obiekezie et Okaeme, 1987 in the accessory breathing organs of ***Heterobranchus bidorsalis*** (Nigeria)

Myxobilatus synodontis Siau, 1971 in the gills of ***Synodontis ansorgii*** (Benin)

Ortholinea Shulman, 1962

Ortholinea africanus Abdel-Ghaffar, El-Toukhy, Al-Quraishy, Al-Rasheid, Abdel-Baki, Hegazy et Bashtar, 2008 in the urinary bladder of ***Oreochromis niloticus*** (Egypt)

Sphaerospora Thélohan, 1892

Sphaerospora melenensis Fomena, Marquès et Bouix, 1993 in the kidney of ***Oreochromis niloticus***

Sphaerospora sangmelimaensis Fomena, Marquès et Bouix, 1993 in the kidney of ***Brienomyrus brachyistius*** (Cameroon), *Hepsetus odoe*, *Petrocephalus simus*

Sphaerospora tilapiae Fomena, Marquès et Bouix, 1993 in the kidney and spleen of ***Oreochromis niloticus*** (Cameroon)

Triangula Chen et Hsieh, 1984

Triangula egyptica Abdel-Ghaffar, El-Toukhy, Al-Quraishy, Al-Rasheid, Abdel-Baki, Hegazy et Bashtar, 2008 in the kidney of ***Oreochromis niloticus*** (Egypt)

Zschokkella Auerbach, 1910

Zschokkella nilei Abdel-Ghaffar, El-Toukhy, Al-Quraishy, Al-Rasheid, Abdel-Baki, Hegazy et Bashtar, 2008 in the kidney of ***Oreochromis niloticus*** (Egypt)

MULTIVALVULIDA Shulman, 1959

Kudoa Meglitsch, 1947

Kudoa eleotrici Siau, 1971 in the gills of ***Kribia kribensis*** (Benin). Note: this is a very exceptional finding as species of *Kudoa* are typically parasites of marine fishes.

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Chapter 4.4.

MONOGENEA

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Monogenea – basic characteristics, life cycles, classification and principal diagnostic features

- parasitic flatworms with a syncytial tegument (Platyhelminthes: Neodermata)
- over 5,500 species allocated to more than 750 genera
- mostly parasites of freshwater, brackish water and marine fishes; a number of species parasitise crustaceans, cephalopods, amphibians, reptiles and a mammal
- majority of African species found on external surfaces (gills, skin, fins, rarely mouth cavity and nostrils); a few species are endoparasitic (*Enterogyrus* – foregut and stomach, *Urogyrus* – urinary bladder)
- body dorsoventrally flattened, varying in size from *ca.* 100 μm up to 4 cm long (typically 0.3-10 mm)
- main attachment organ on the posterior end called haptor (or opisthaptor) houses a variable array of sclerotised (hard) structures; number, shape and configuration of the haptoral structures are key to species identification and classification
- simple digestive system consisting of mouth, pharynx and intestine with no terminal opening (anus)
- intestine usually with two simple or branched stems often fusing (anastomosing) posteriorly
- hermaphroditic (commonly protandrous), usually with cross-fertilisation
- distal parts of the male and female reproductive system (male copulatory organ, vagina) may contain sclerotised elements (*e.g.*, copulatory tube, accessory piece) that help in species identification
- direct life cycles (no intermediate host required) (Fig. 4.4.1)
- oviparous (oncomiracidium larva), viviparous (sequential polyembryony)
- a high degree of host and site (microhabitat) specificity

The classification of monogeneans is still under discussion. Even the name of the class, Monogenea (used by the majority of workers) or Monogenoidea, is controversial (Wheeler & Chisholm 1995). There are several classifications of monogeneans that are based on morphology, ontogeny and spermatology (Bychowsky 1957; Yamaguti 1963; Lebedev 1988; Malmberg 1990; Justine 1991; Boeger & Kritsky 1993, 2001). The system of Boeger and Kritsky (1993,

Fig. 4.4.1. Life cycles of monogeneans (no intermediate host required). **A.** Oviparous life cycle (*Dactylogyrus* sp.); **B.** Viviparous life cycle (*Macrogyrodactylus* sp.). (Illustrations by M. Luo.)

2001) is followed here for the higher taxonomical levels, based on a variety of anatomical and ultrastructural characters. The subclasses as listed below are now well accepted, as is the division of Heteronchoinea into two infrasubclasses (*i.e.*, Oligonchoinea and Polystomatoinea).

Generic classification of monogeneans is based mainly on characters associated with the attachment structures. However, information on the internal anatomy and sclerotised distal parts of the male and female reproductive system is also important, as an integral part of the generic definition.

Species identification of monogeneans (especially so-called lower monogeneans – Polyonchoinea) is based on the morphology of the sclerotised structures of the haptor and distal parts of the reproductive systems (*i.e.*, male copulatory organ and vagina). However, details on the arrangement of internal structures may also supplement the taxonomical evaluation, and should ideally be a part of the species description.

The unique and characteristic morphological feature of the Monogenea is the presence of the posterior attachment organ called the haptor (or opisthaptor). It is a complex organ composed of the attachment disc and various sclerotised structures (Fig. 4.4.2). The terminology of the haptoral structures is not unified and some researchers use these terms differently. Here, the following terms are used in keying out these parasites:

Anchors (hamuli, grypi, large hooks, central hooks, Mittelhakens) are paired trifid structures situated on the ventral and/or dorsal surface of the central part of the haptor. One or two pairs of anchors may be present, but in some monogeneans (*e.g.*, species of *Heteronchocleidus*, *Trianchoratus*, *Urogyrus*) one of the anchors may not be fully developed, *i.e.*, it is much reduced in size and shape. In a typical case, the anchor consists of inner root (superficial root, ventral root, guard), outer root (deep root, dorsal root, shaft), base, shaft (blade) and point; the membranous structures arising from the convex surface of the shaft are termed anchor filaments (wings, filament loops). The anchors of some genera may possess accessory sclerites (patch, cuneus) associated with the tip of the inner root (*e.g.*, species of *Birgiellus*, *Paraquadriacanthus*, *Quadriacanthus*).

Bars (connecting bars, transverse bars) are one or two-piece structures connecting the basis of individual members of a pair of anchors. Generally, each pair of anchors has a bar and together they form the so-called ventral and/or dorsal anchor-bar complex. In a number of genera with two pairs of anchors, only one pair has a bar (*e.g.*, species of *Enterogyrus*, *Eutrianchoratus*). Conversely, two bars (ventral and dorsal) may be present in some monogeneans with one pair of anchors (*e.g.*, species of *Dactylogyrus*, *Gyrodactylus*).

Needles (4A hooks) are paired delicate (usually poorly detectable) splinter-like structures of which the nature and origin have not yet been clearly documented. Mostly they are considered to be vestigial anchors or vestigial hooks (*e.g.*, species of *Dactylogyrus*, *Dogielius*, *Schilbetrematoides*).

Hooks (marginal hooks, uncinuli) are bilaterally arranged pairs of small sickle-shaped structures. In a typical case, each hook consists of a sickle (with sickle-filament loop or FH loop) and a handle. There are two types of hooks called unhinged and hinged hooks. The sickle of a hinged hook is movable in relation to the handle. The number of hooks is 14 in species with unhinged hooks (Dactylogyridae, Diplectanidae) and 16 in species with hinged hooks (Gyrodactylidae). There are several types of numbering systems for hook pairs; here the system of Mizelle (1936) is adopted, because it is the only currently used method that considers both the anteroposterior and dorsoventral positions of the respective hook pairs in the adult haptor.

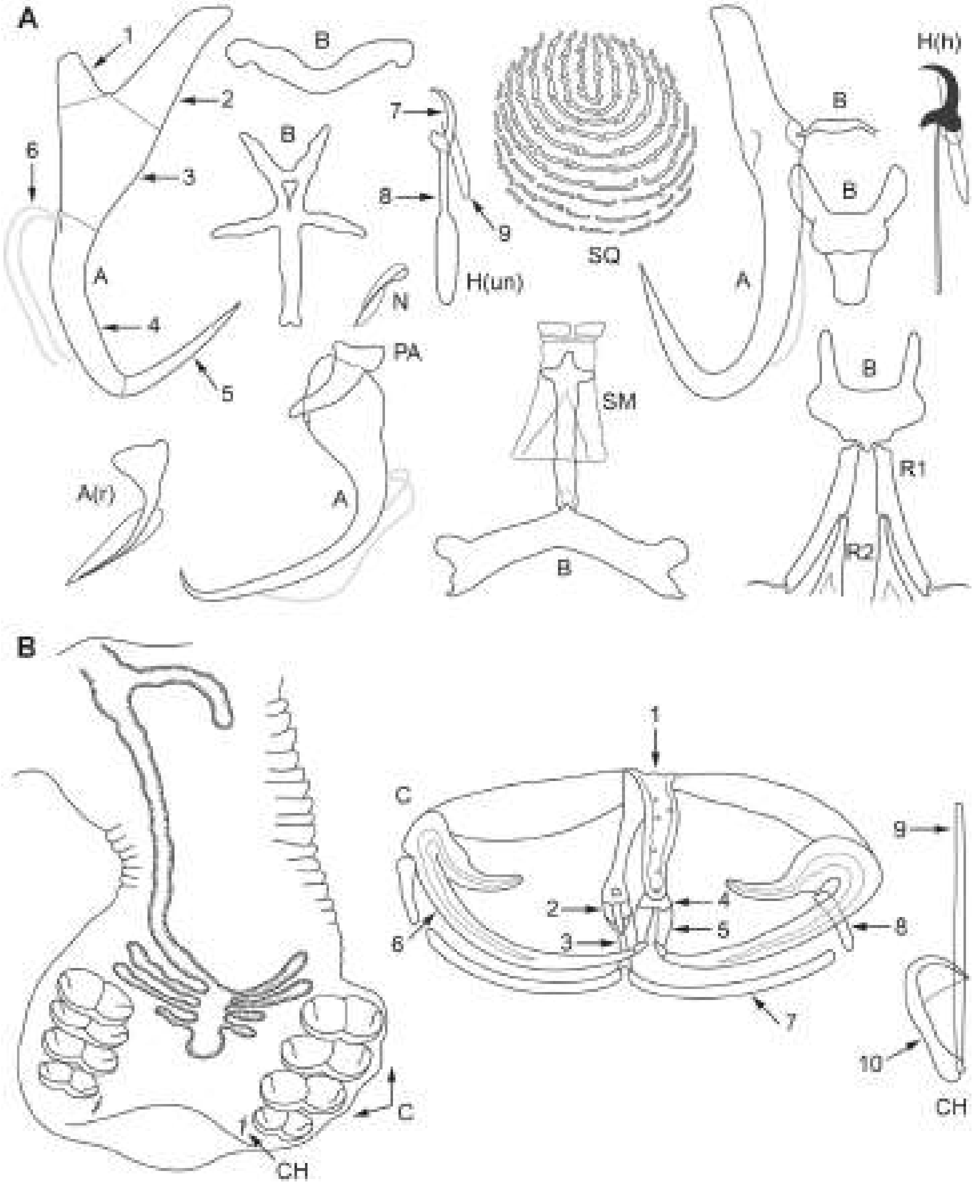


Fig. 4.4.2. Examples of haptoral attachment structures, as typically seen in papers describing new species. **A.** Polyonchoinea. A = anchor, A(r) = anchor (rudimentary), B = bar, H(h) = hook (hinged), H(un) = hook (unhinged), N = needle, Pa = patch, R = rods, SM = supporting membrane, SQ = squamodisc, 1 = outer root, 2 = inner root, 3 = base, 4 = shaft, 5 = point, 6 = filament, 7 = sickle, 8 = handle, 9 = FH loop; **B.** Oligonchoinea. (Modified after Khotenovsky 1985.) C = clamps, CH = central hook, 1 = median plate, 2 = proximal additional sclerite, 3 = distal additional sclerite, 4 = trapeze spur, 5 = anterior joining sclerites, 6 = anterior jaw, 7 = posterior jaw (median sclerite), 8 = posterior jaw (lateral sclerite), 9 = handle, 10 = sickle.

Squamodiscs are circular or oval plate-like formations, which are found only in certain monogeneans of the Diplectanidae. There are, typically, two squamodiscs (one ventral and one dorsal) located anteriorly to the anchor-bar complexes. Each disc possesses scales embedded in the tegument, which appear under the microscope as rootlets arranged in rows.

Clamps are metamorphosed suckers characteristic of higher monogeneans (Oligonchoinea). They are highly specialised structures, often armed with sclerotised elements. The number of clamps varies from two to many; they are distributed symmetrically or asymmetrically. The number and arrangement of clamps as well as the number, shape and size of clamp sclerites are of taxonomic importance in differentiating taxa.

4.4.1. Identification keys for monogeneans (adults)

The guide to the monogeneans parasitising African freshwater fishes is presented here as keys to individual genera and higher-level taxa. Every step in the keys refers to a corresponding figure for a better understanding of identifying feature(s). Figures are labelled to illustrate the used terminology of the sclerotised structures; taxonomically important characters indicated by arrows. In case of hooks, only half of them is depicted in the key to the Dactylogyridae. The genus *Ancyrocephalus* is not included in the keys. Following the emendation of the generic diagnosis for *Ancyrocephalus* of Bychowsky and Nagibina (1970), *A. barilli*, *A. claveaui*, *A. limnotrissae* and *A. pellonulae* do not belong to the genus *sensu stricto*. Nevertheless, we retain them in *Ancyrocephalus* until their generic status is formally resolved. Thus, these species are listed below under *Ancyrocephalus sensu lato*.

Key to the subclasses/infrasubclasses of monogeneans

1 (2) Hook-like sclerites with various connecting and supporting sclerites (*e.g.*, bars, squamodiscs) are main attachment structures of haptor [Fig. 4.4.2A]……………………………………………………**Polyonchoinea**

2 (1) Main attachment structures of haptor are morphologically and functionally changed suckers – clamps [Fig. 4.4.2B]……………………**Oligonchoinea**

Key to the families of the Oligonchoinea Bychowsky, 1937

1 (2) Haptor with 4 + 4 clamps; male copulatory organ armed with a circle of spines, present in anterior part of the body (just behind pharynx); no fused (concrescent) individuals occur [Fig. 4.4.3]……………….**Diclidophoridae**

2 (1) Haptor with 4 + 4 and more clamps; one pair of posteriorly situated hooks (central hooks) usually present; male copulatory organ absent; already in juvenile stage, two individuals (diporpae) permanently fused forming an X-shape [Fig. 4.4.4]……………………………………………**Diplozoidae**

Key to the genera of the Diplozoidae Tripathi, 1959

1 (2) Haptor with 4 + 4 laterally situated clamps and one pair of small central hooks; gill parasites of *Brycinus macrolepidotus* (Alestidae) and Cyprinidae [Fig. 4.4.4A]……………………………………………….***Paradiplozoon***

2 (1) Haptor with more than 4 + 4 laterally situated clamps (up to 15 pairs) and one pair of small central hooks; gill parasites of *Alestes baremoze* (Alestidae) and Cyprinidae [Fig. 4.4.4B]…………………………….***Afrodiplozoon***

Key to the families of the Polyonchoinea Bychowsky, 1937

1 (2) Oviparous, usually with two pairs of eye spots; haptor with unhinged (dactylogyrid) hooks [Fig. 4.4.5A]…………………………………………….……3

2 (1) Viviparous or oviparous, eye spots are lacking; haptor with hinged (gyrodactylid) 8 + 8 hooks [Fig. 4.4.5B]…………………………..**Gyrodactylidae**

3 (4) Haptor with 7 + 7 unhinged hooks, two pairs of anchors (one or two anchors may be rudimentary), two bars (one may be rudimentary or absent) [Fig. 4.4.6A]……………………………………………...**Dactylogyridae**

4 (3) Haptor with 7 + 7 unhinged hooks, two pairs of anchors, two bars (dorsal bar two-pieced); accessory adhesive organs (squamodiscs) present [Fig. 4.4.6B] .... **Diplectanidae**

Key to the genera of the Dactylogyridae Bychowsky, 1933

1 (2) Dactylogyrids parasitising internal organs……………………………………3

2 (1) Dactylogyrids parasitising external organs………………………………..5

3 (4) In stomach; haptor with two pairs of anchors; ventral anchors associated with the ventral bar (*i.e.*, ventral anchor-bar complex present); dorsal anchors with recurved inner root, elongate outer root; dorsal bar absent; male copulatory organ a spirally coiled tube; in Cichlidae [Fig. 4.4.7A]…………………………………………………… ***Enterogyrus***

4 (3) In urinary bladder; haptor with one pair of ventral anchors associated with the ventral bar; right anchor rudimentary (reduced in size and shape); dorsal anchor-bar complex absent; in Cichlidae [Fig. 4.4.7B]………***Urogyrus***

5 (6) Haptor with two developed anchors (ventral or dorsal pair) and two anchors reduced or replaced by needles (poorly defined)……………………………7

6 (5) Haptor with more than two developed anchors…………………………….13

7 (8) Ventral anchor-bar complex developed; dorsal anchors modified into spike-like sclerites; dorsal bar absent; gill parasites of *Citharinus citharus*

(Citharinidae) [Fig. 4.4.8A]……….…………………………….Nanotrema

8 (7) One pair of delicate splinter-like structures (needles) located near hook pair V present………………………………………………………………………...9

9 (10) Only one bar present; anchors with short roots (inner root often with basal fold) of similar size, poorly differentiated shafts and points (with subterminal curvature and strongly recurved tip) directing towards each other (like a pair of pincers); gill parasites of Cyprinidae [Fig. 4.4.8B]………………… ……………………………………………………………………..***Dogielius***

10 (9) One or two bars present…………………………………………………11

11 (12) Dorsal bar present; ventral bar usually smaller than the dorsal one, rudimentary or absent; dorsal anchors often with roots of unequal size, well-differentiated shaft and point; mostly on gills of Cyprinidae [Fig. 4.4.9A] ……………………………………………………………***Dactylogyrus***

12 (11) Ventral bar simple, rod-shaped; dorsal bar complex, comprising bar proper and massive shield-like structure posteriorly serving as a guide for anchor points; dorsal anchors with elongate inner root having a superficial protuberance near mid-length; gill parasites of *Schilbe* (Schilbeidae) [Fig. 4.4.9B]…………………………………………..***Schilbetrematoides***

13 (14) Three developed and one (ventral) rudimentary (*i.e.*, markedly reduced in size and shape) anchors present…………………………………………15

14 (13) Four (two pairs) developed anchors present……………………………..17

15 (16) Two bars present; developed anchors (two dorsal, one ventral) with well-differentiated roots; dorsal anchors usually differ from each other in size and shape; gill parasites of *Ctenopoma* (Anabantidae) [Fig. 4.4.10A] ……………………………………………………….***Heteronchocleidus***

16 (15) One bar present; developed anchors (two left, one right) in a claw-like position, each with stout inner root and poorly developed outer root; a circular muscle attached terminally to the inner root of the left ventral anchor detectable; gill parasites of *Parachanna obscura* (Channidae) [Fig.4.4.10B]…………………………………………………***Eutrianchoratus***

17 (18) One or both pairs of anchors associated with a two-piece bar……………19

18 (17) Each anchor pair associated with a one-piece (solid) bar…………………25

19 (20) Both bars two-pieced; ventral and dorsal anchors similar in shape and size; base of copulatory tube delicate, usually with finger-like processes; gill parasites of Mormyridae [Fig. 4.4.11A]………………………. ***Bouixella***

20 (19) Ventral bar two-pieced; dorsal bar solid……………………………………21

21 (22) Ventral bar comprising two well-separated components; ventral anchors

markedly smaller than dorsal anchors; dorsal bar straight, broadly V- or M-shaped; gill parasites of Cichlidae [Fig. 4.4.11B]…………***Onchobdella***

22 (21) Ventral bar comprising two components articulating medially……………23

23 (24) Anchors with patches, rootless; dorsal bar T-shaped, with bilateral arms and expanded mid-region with posterior process; gill parasites of Clariidae, *Bagrus* (Bagridae), and *Papyrocranus afer* (Notopteridae) [Fig. 4.4.12A] ……………………………………………………………..***Quadriacanthus***

24 (23) Anchors without patches; dorsal anchors with inner root terminally curled; ventral anchors with thickened ridge extending from outer root across base to shaft; pouch-like structure (onchium), through which dorsal extrinsic muscles extend, present in the anterior region of haptor; gill parasites of *Chrysichthys* (Claroteidae) and *Malapterurus electricus* (Malapteruridae) [Fig. 4.4.13]……………………………………***Protoancylodiscoides***

25 (26) Anchors with patches……………………………………………………27

26 (25) Anchors without patches…………………………………………………..31

27 (28) Patches on ventral anchors only; anchors large, with poorly developed outer root; bars simple in shape; gill parasites of *Heterotis niloticus* (Arapaimidae) [Fig. 4.4.12B]…………………………………………..***Heterotesia***

28 (27) Patches on both dorsal and ventral anchors……………………………….29

29 (30) Ventral and dorsal anchors rootless; patches small; dorsal anchors with shaft sharply (at about 90º) bent proximally; dorsal bar cross-shaped; ventral bar triangular or three-armed; gill parasites of *Clarias* (Clariidae) [Fig. 4.4.14A]……………………………………………………***Birgiellus***

30 (29) Dorsal anchors robust, with flange on superficial surface of base, large patch (wings unequal); ventral anchors small, with delicate patch; dorsal bar complex, with anterior shield and posterior arrow- or T-shaped process; ventral bar broadly U-shaped; in the nasal cavity of *Clarias gariepinus* (Clariidae) [Fig. 4.4.14B]………………………….***Paraquadriacanthus***

31 (32) Dorsal bar with two submedial auricles …………………………………33

32 (31) Dorsal bar lacking auricles………………………………………………35

33 (34) Dorsal bar with long auricles and lateral wing-shaped enlargements; ventral bar associated with supporting membrane marked by fan-shaped median thickenings; gill parasites of Cichlidae [Fig. 4.4.15A]………………… ……………………………………………………………………***Scutogyrus***

34 (33) Dorsal bar with auricles variable in length; ventral bar not associated with supporting membrane, V-shaped, usually with a medial portion reduced in diameter; an auxiliary plate lying in close proximity of the male copulato-

ry organ sometimes present; gill parasites of *Aphyosemion cameronense* (Nothobranchiidae), Cichlidae and *Polycentropsis abbreviata* (Nandidae) [Fig. 4.4.15B]……………………………………………….***..Cichlidogyrus***

35 (36) Both ventral and dorsal bar associated with lightly sclerotised (sometimes poorly defined) supporting membrane; ventral bar often with median process; anchors with inner roots having recurved (erect) terminal half, elongate shaft and short point; gill parasites of Alestidae [Fig. 4.4.16A]……………………………………………………***Annulotrema***

36 (35) Supporting membrane absent or associated with only one bar……………37

37 (38) Bar(s) with a median projection and/or two bilateral anterior arms (*i.e.*, bar ends bent at about 90° anteriorly)……………………………………………39

38 (37) Bars lacking median projection and such bilateral anterior arms…………47

39 (40) Median projection articulated to the ventral bar and associated with lightly sclerotised skirt-like supporting membrane; dorsal bar yoke-shaped with a posterior shield; gill parasites of *Auchenoglanis occidentalis* (Claroteidae) [Fig. 4.4.16B]…………………………………………………….***Bagrobdella***

40 (39) Median projection arising (not articulated) from the ventral bar……………41

41 (42) Ventral anchors modified in shape……………………………………………43

42 (41) Ventral anchors with basal surface protuberance…………………………45

43 (44) Ventral anchors with recurved inner root, elongate (erected) outer root, and diagonally truncate point; ventral bar with two bilateral anterior arms, small/delicate posteromedial projection usually present; dorsal bar simple, rod-shaped; gill parasites of Alestidae [Fig. 4.4.17A]……………………… ……………………………………………………………***..Characidotrema***

44 (43) Ventral anchors with prominent superficial knob on base near its union with the shaft, shaft sharply (usually at about 90º) bent proximally, roots variable in shape; bars usually with lateral, subterminal (often horn-shaped) and medial anterior projections; accessory sclerite associated with anteromedial projection of the ventral bar may be present; gill parasites of Schilbeidae [Fig. 4.4.17B]…………………………………………***Schilbetrema***

45 (46) Ventral anchors with pestle-shaped protuberance diagonally extending from outer root to inner side of proximal part of the shaft; ventral bar with lobed ends and medial projection; dorsal anchors with small to reduced outer root; dorsal bar simple, with indistinct supporting membrane; gill parasites of *Synodontis* (Mochokidae) [Fig. 4.4.18A]………………………… ……………………………………………………………… ***Synodontella***

46 (45) Ventral anchors robust, with leaf-shaped protuberance extending along medial part of base; ventral bar with lobed ends and medial projection;

dorsal anchors with shaft slightly swollen at its union with base; dorsal bar with two subterminal joint-like thickenings; gill parasites of *Gnathonemus petersii* (Mormyridae) [Fig. 4.4.18B]……………………***Archidiplectanum***

47 (48) Bars (primarily ventral bar) with recurved ends, subterminal constrictions; dorsal bar with indistinct supporting membrane; ventral anchors with wide base; gill parasites of Cichlidae in Madagascar [Fig. 4.4.19A]……………… ………………………………………………………………***Insulacleidus***

48 (47) Ventral bar saddle-shaped, with enlarged (bulbous) terminations and rectangular enlargement of anteromedial margin; dorsal bar rod- or broadly U-shaped; anchors with enlarged roots and relatively delicate shaft; gill parasites of *Distichodus* (Distichodontidae) [Fig. 4.4.19B]………………… …………………………………………………………***Afrocleidodiscus***

Key to the genera of the Gyrodactylidae van Beneden et Hesse, 1863

1 (2) Haptor with 16 hooks of the same type; ventral and dorsal bar present……3

2 (1) Haptor with 16 hooks of two different types (ten hooks with large falculate sickles, six smaller hooks with well-articulated sickles), a pair of muscular adhesive discs situated on the side of anchors; no dorsal bar present; gill parasites of *Polypterus senegalus* (Polypteridae) [Fig. 4.4.20A]…………… ………………………………………………………..***Diplogyrodactylus***

3 (4) Hooks evenly distributed along the edge of haptor……………………………5

4 (3) Hooks distributed unevenly; 14 hooks arranged in a row along the posterior margin of haptor; two hooks located on anterolateral lobes, reflected forwards; peg-like tegumental extensions (supporting struts) present on lateral and anterior margins of haptor; inner roots of anchors associated with accessory bars; ventral bar associated with two pairs of supporting rods (not incorporated in the bar); gill, skin/fin parasites of Clariidae, *Ctenopoma muriei* (Anabantidae), *Lates niloticus* (Latidae) and Polypteridae [Fig. 4.4.20B]…………………………………………***Macrogyrodactylus***

5 (6) Haptor with four pairs of accessory bars; three (two lateral, one central) supporting rods incorporated in ventral bar present; skin/fin parasites of *Marcusenius macrolepidotus* (Mormyridae) [Fig. 4.4.21A]………………………………………***Mormyrogyrodactylus***

6 (5) Haptor lacking accessory bars…………………………………………………7

7 (8) Anchors with two developed roots; outer root conspicuous, approximately half-length of inner root; ventral bar without membrane and anterolateral processes; gill, skin/fin parasites of Alestidae [Fig. 4.4.21B] ………………………………………………………….***Afrogyrodactylus***

8 (7) Anchors with only one (inner) developed root………………………………9

9 (10) Ventral bar with membrane, anterolateral processes may be present; male copulatory organ bulbous, equipped with one apical spine and row(s) of small spines; gill, skin/fin parasites of various host families [Fig. 4.4.22A] ……………………………………………………………………***Gyrodactylus***

10 (9) Anchors with a constriction between shaft and point; ventral bar with membrane, anterolateral processes lacking; male copulatory organ muscular, consists of a central curved cone and a muscular pouch armed with numerous small spines; gill parasites of *Citharinus citharus* (Citharinidae) [Fig. 4.4.22B]………………………………………………***Citharodactylus***

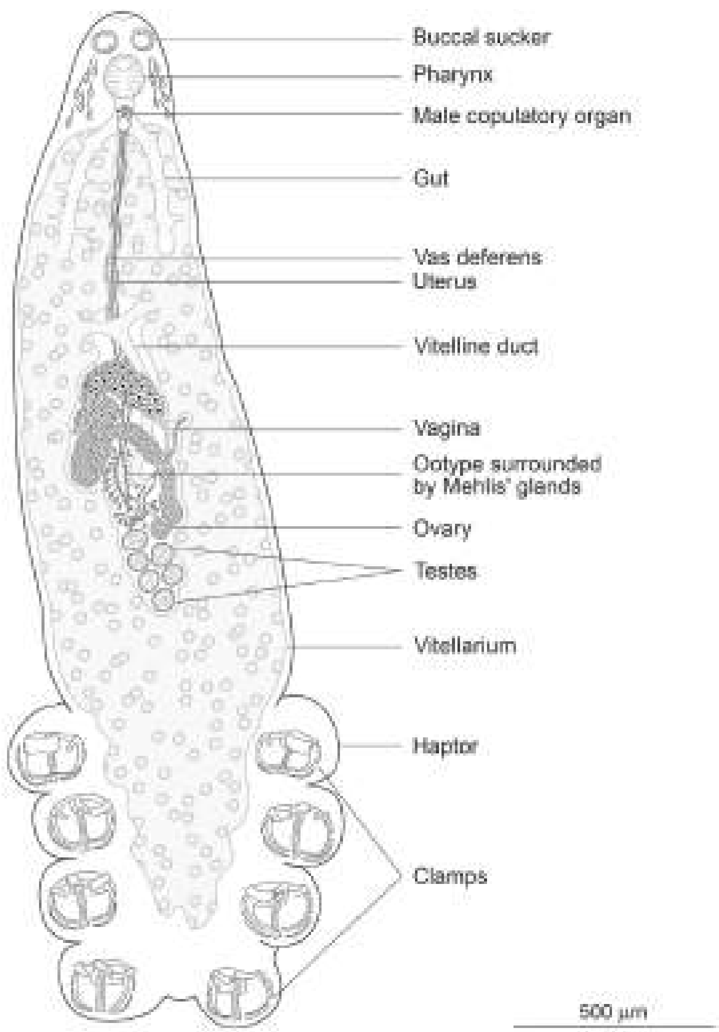


Fig. 4.4.3. Monogenea (Diclidophoridae). *Heterobothrium fluviatilis* Euzet et Birgi, 1975 from *Tetraodon lineatus*. (Modified from Euzet & Birgi 1975.)

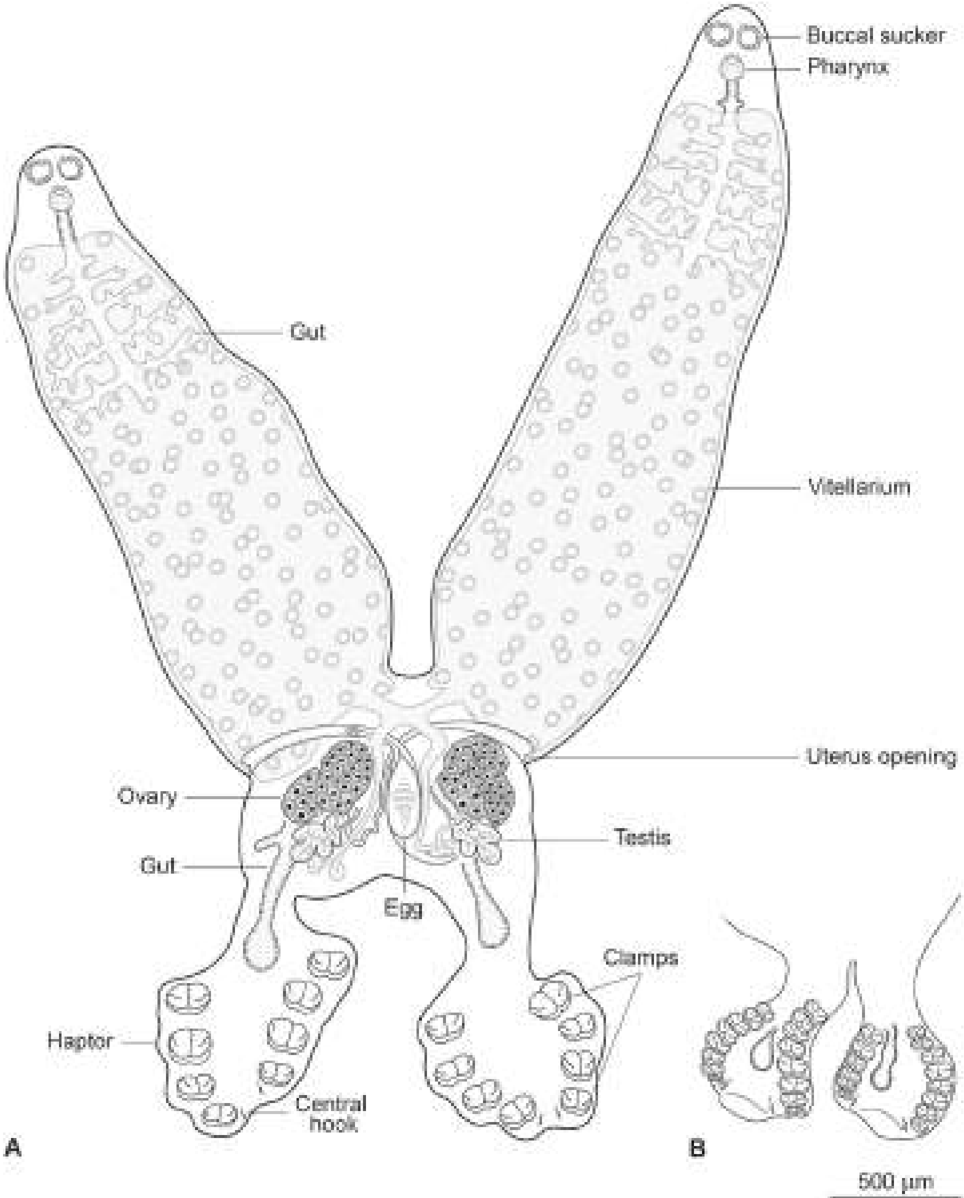


Fig. 4.4.4. Monogenea (Diplozoidae). **A.** *Paradiplozoon ghanense* (Thomas, 1957) from *Brycinus macrolepidotus*; **B.** *Afrodiplozoon polycotyleus* (Paperna, 1973) from *Enteromius cercops*. (Modified from Khotenovsky 1985.)

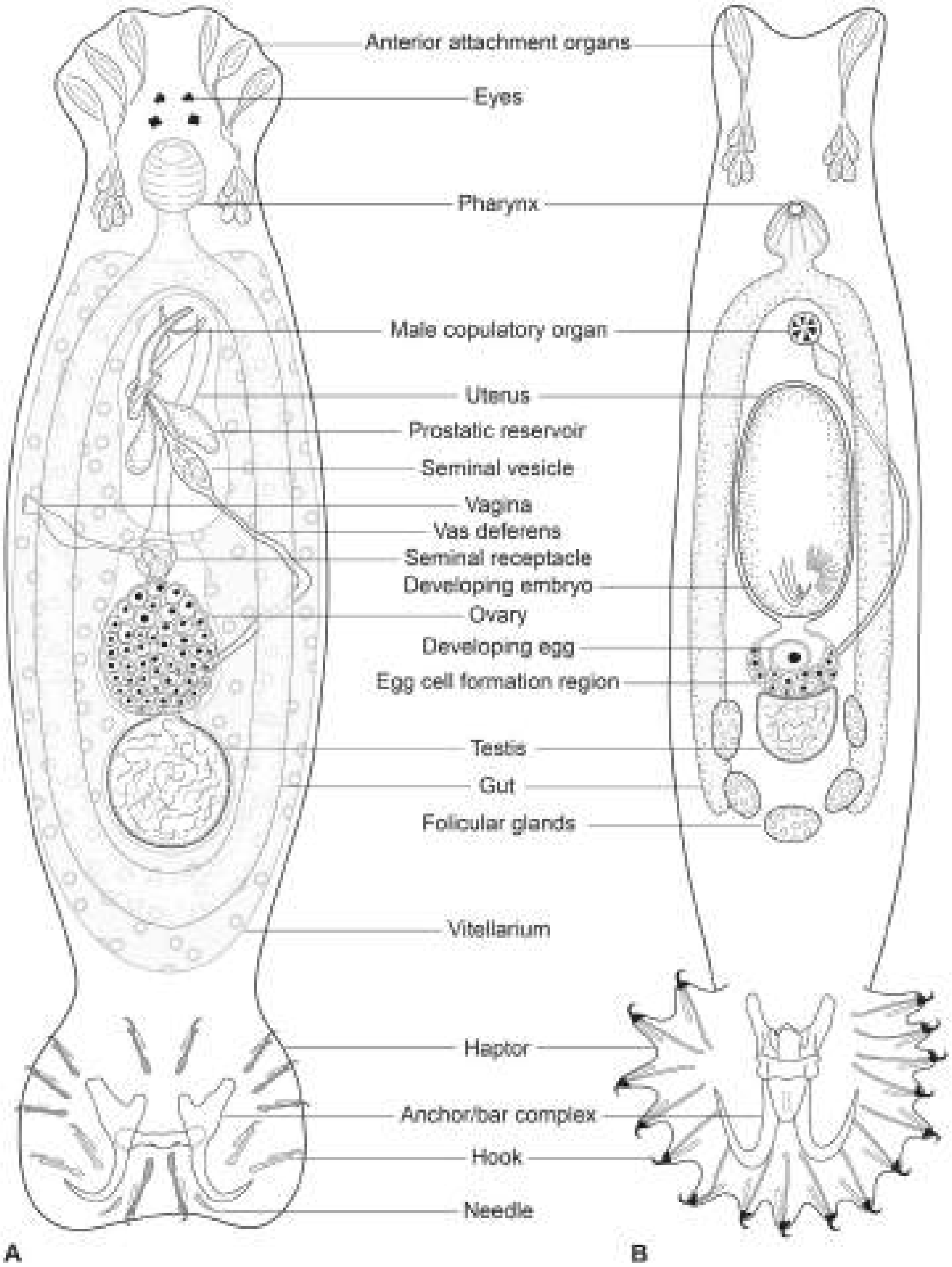


Fig. 4.4.5. Monogenea (Polyonchoinea). **A.** Generalised anatomy of oviparous *Dactylogyrus* sp., ventral view; **B.** Generalised anatomy of viviparous *Gyrodactylus* sp., ventral view. (Modified from Roberts *et al.* 2013.)

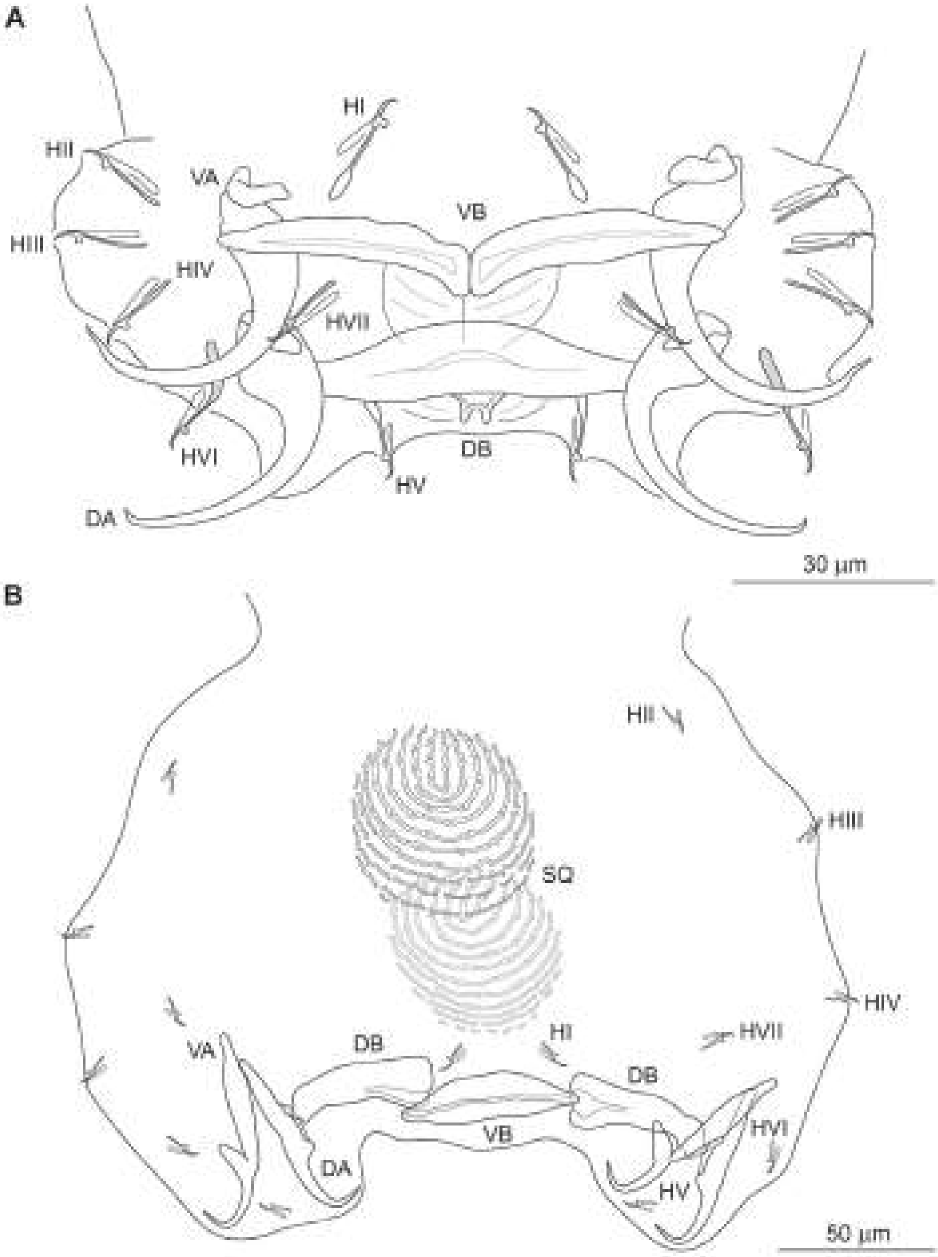
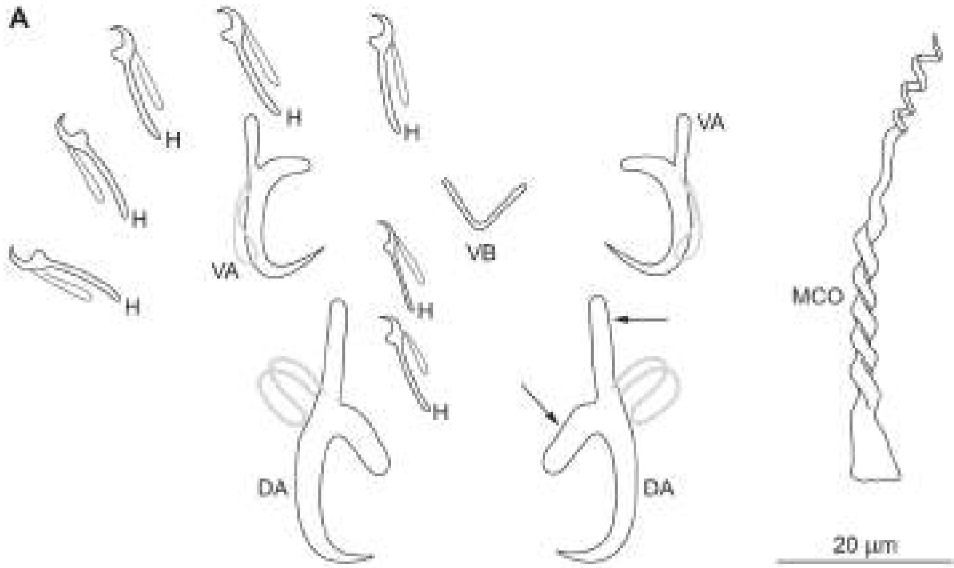


Fig. 4.4.6. Monogenea (Polyonchoinea). **A.** Haptor of *Quadriacanthus ashuri* Kritsky et Kulo, 1988 (Dactylogyridae) from *Clarias gariepinus*. **B.** Haptor of *Diplectanum lacustre* Thurston et Paperna, 1969 (Diplectanidae) from *Lates niloticus*. (Modified from Kritsky & Kulo 1988.) VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; HI-VII = hooks; SQ = squamodiscs.



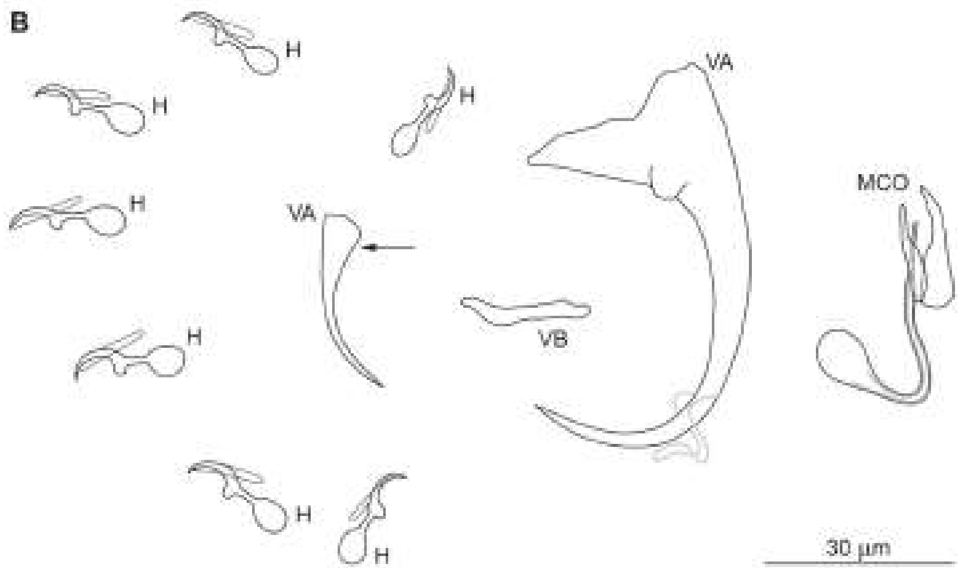


Fig. 4.4.7. Monogenea (Dactylogyridae). **A.** *Enterogyrus amieti* Bilong Bilong, Euzet et Birgi, 1996 from *Sarotherodon galilaeus*; **B.** *Urogyrus cichlidarum* Bilong Bilong, Birgi et Euzet, 1994 from *Benitochromis batesii*. (Modified from Bilong Bilong *et al.* 1994, 1996.) VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; H = hook; MCO = male copulatory organ.

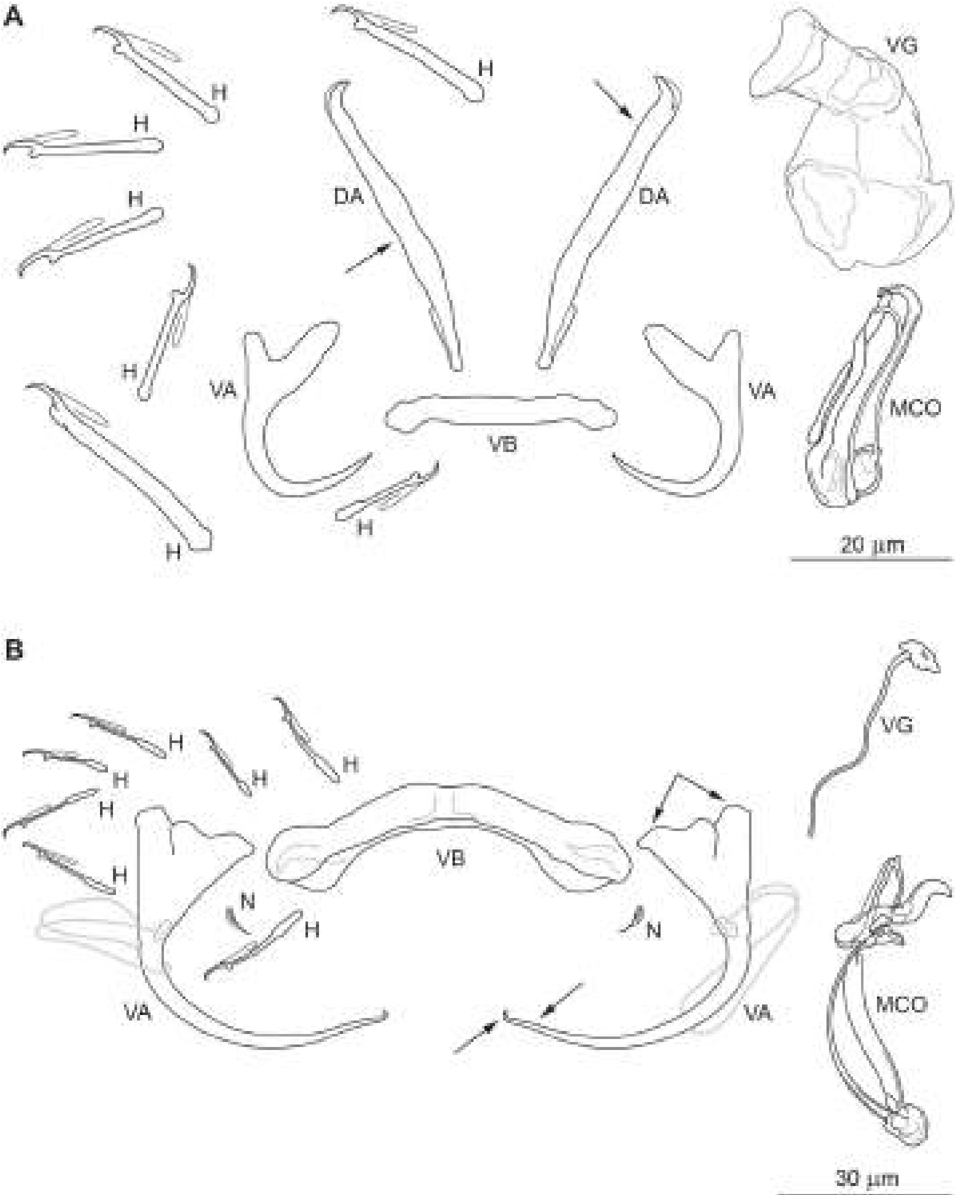


Fig. 4.4.8. Monogenea (Dactylogyridae). **A.** *Nanotrema citharini* Paperna, 1969 from *Citharinus citharus*; **B.** *Dogielius anthocolpos* Guégan, Lambert et Euzet, 1989 from *Labeo coubie*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; N = needle; H = hook; VG = vagina; MCO = male copulatory organ.

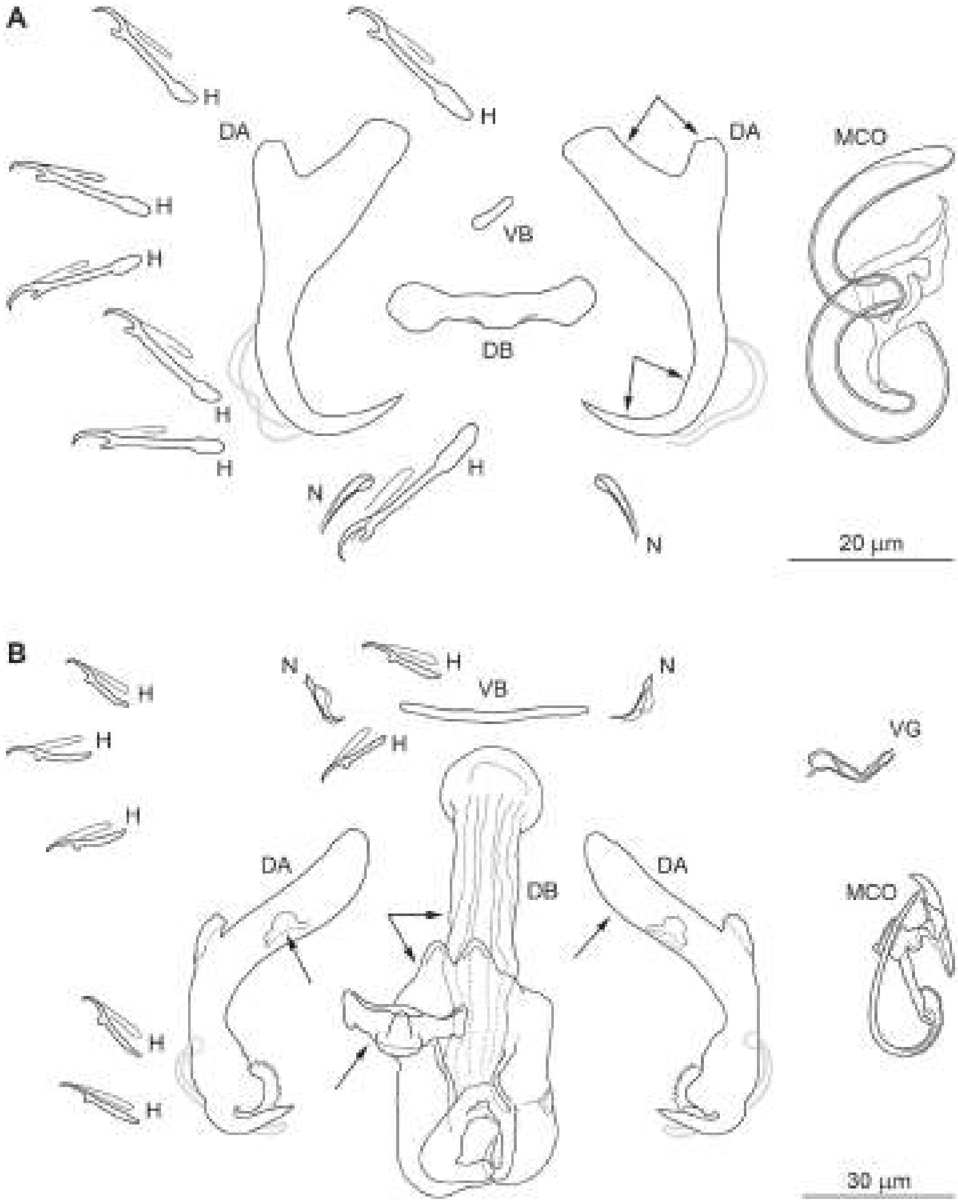


Fig. 4.4.9. Monogenea (Dactylogyridae). **A.** *Dactylogyrus yassensis* Musilová, Řehulková et Gelnar, 2009 from *Labeo coubie*. **B.** *Schilbetrematoides manizani* N'Douba, Lambert, Pariselle et Euzet, 2000 from *Schilbe intermedius*. (Modified from Musilová *et al.* 2009.) VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; N = needle; H = hook; VG = vagina; MCO = male copulatory organ.

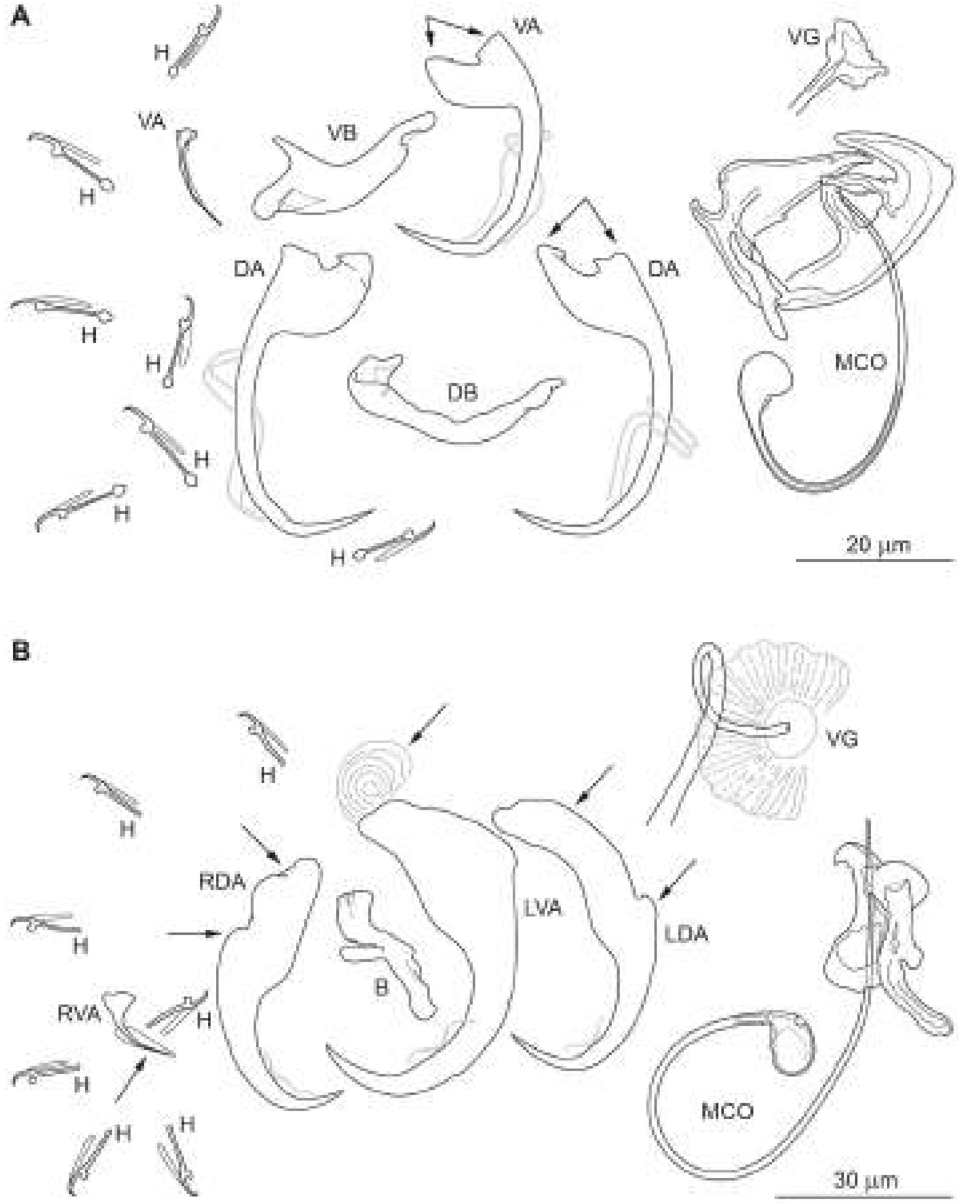


Fig. 4.4.10. Monogenea (Dactylogyridae). **A.** *Heteronchocleidus adjanohouni* Euzet et Dossou, 1975 from *Ctenopoma petherici*; **B.** *Eutrianchoratus malleus* Bilong Bilong, Euzet et Birgi, 1994 from *Parachanna obscura*. VA = ventral anchor; LVA = left ventral anchor; RVA = right ventral anchor; VB = ventral bar; DA = dorsal anchor; LDA = left dorsal anchor; RDA = right dorsal anchor; DB = dorsal bar; B = bar; H = hook; VG = vagina; MCO = male copulatory organ.

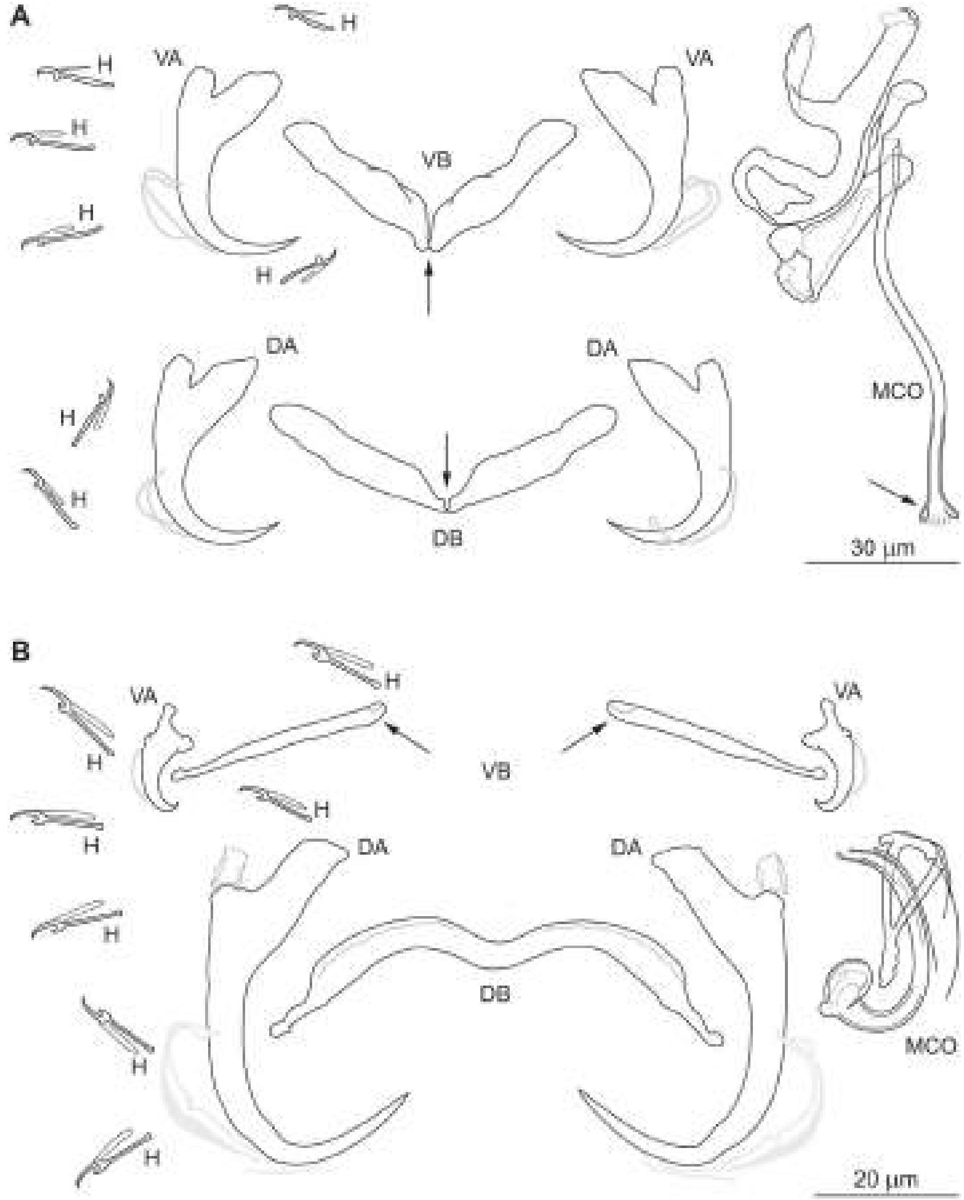


Fig. 4.4.11. Monogenea (Dactylogyridae). **A.** *Bouixella mormyrume* Euzet et Dossou, 1976 from *Mormyrus kannume*; **B.** *Onchobdella voltensis* Paperna, 1968 from *Hemichromis fasciatus*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; MCO = male copulatory organ.

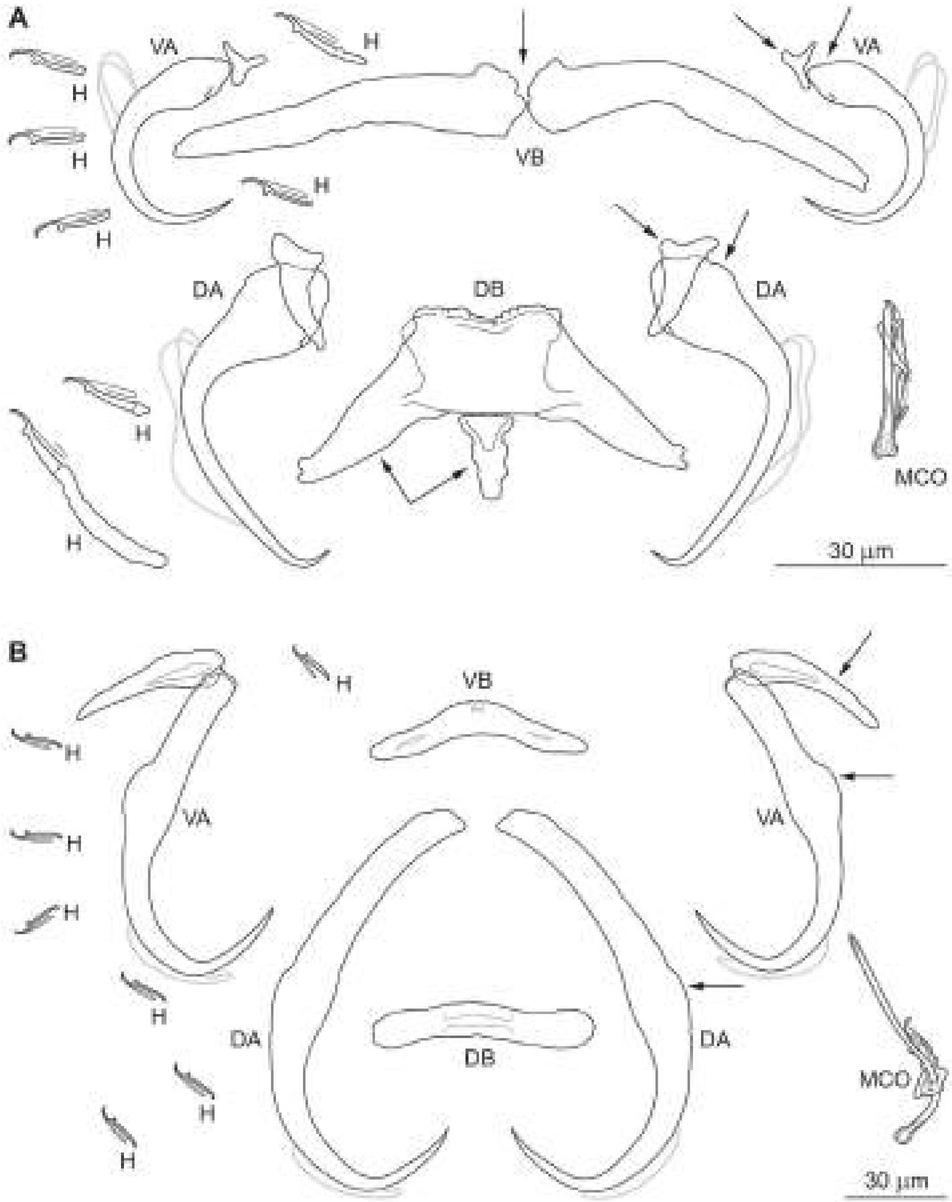


Fig. 4.4.12. Monogenea (Dactylogyridae). **A.** *Quadriacanthus clariadis* Paperna, 1961 from *Clarias gariepinus*; **B.** *Heterotesia voltae* Paperna, 1969 from *Heterotis niloticus*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; MCO = male copulatory organ.

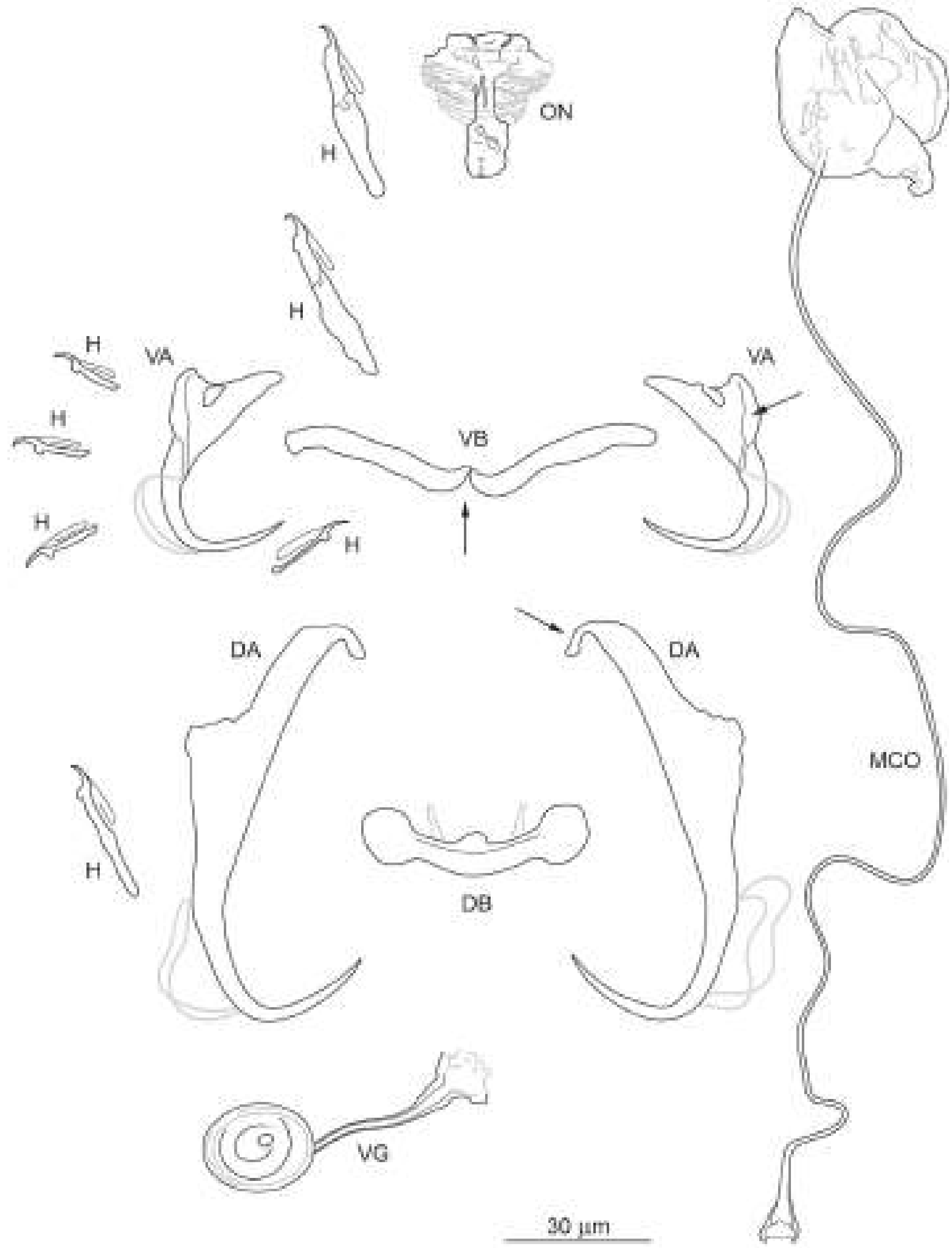


Fig. 4.4.13. Monogenea (Dactylogyridae). *Protoancylodiscoides mansourensis* El-Naggar, 1987 from *Chrysichthys auratus*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; ON = onchium; VG = vagina; MCO = male copulatory organ.

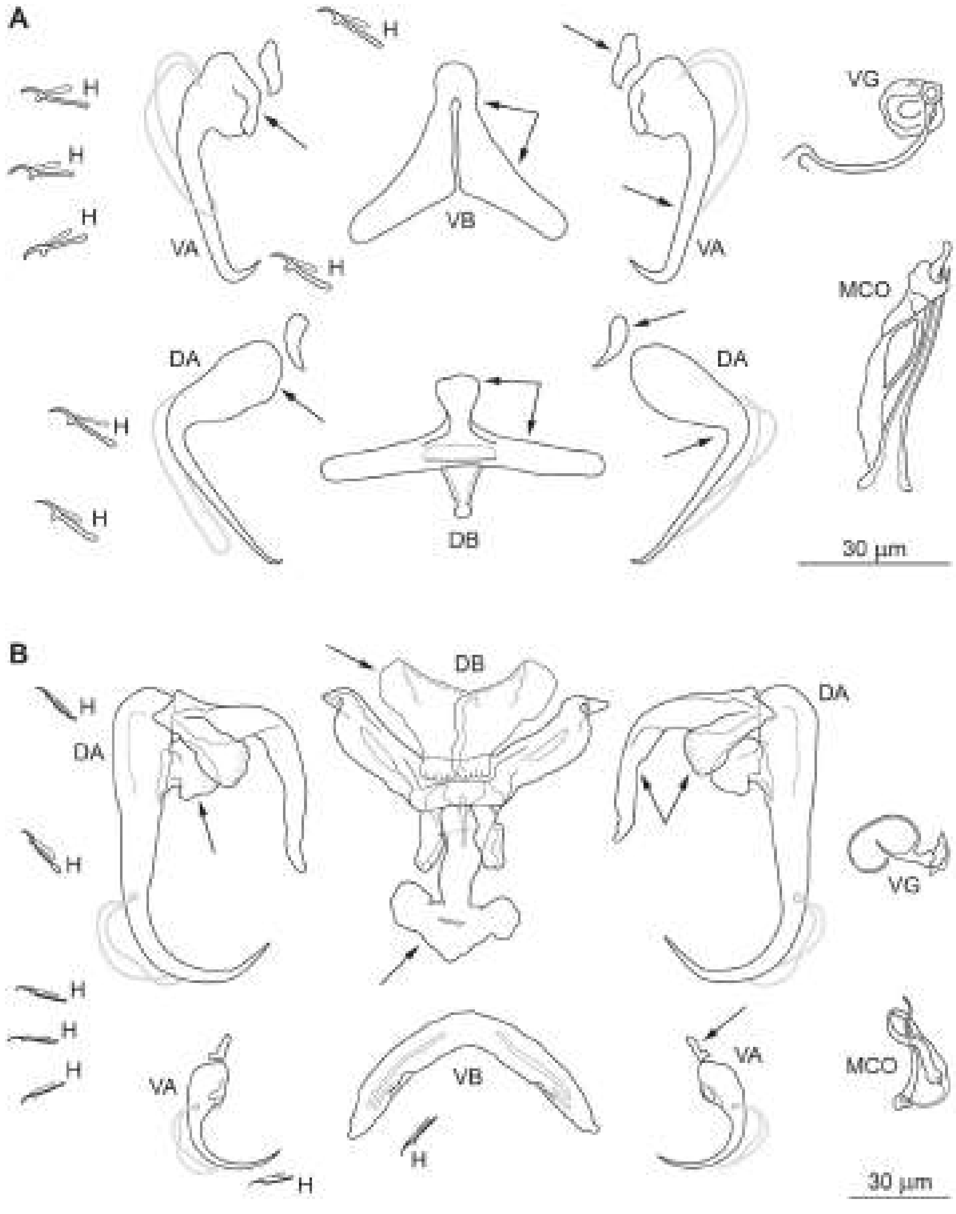


Fig. 4.4.14. Monogenea (Dactylogyridae). **A.** *Birgiellus mutatus* Bilong Bilong, Nack et Euzet, 2007 from *Clarias pachynema*. **B.** *Paraquadriacanthus nasalis* Ergens, 1988 from *Clarias gariepinus*. (Modified from Bilong Bilong *et al.* 2007.) VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ.

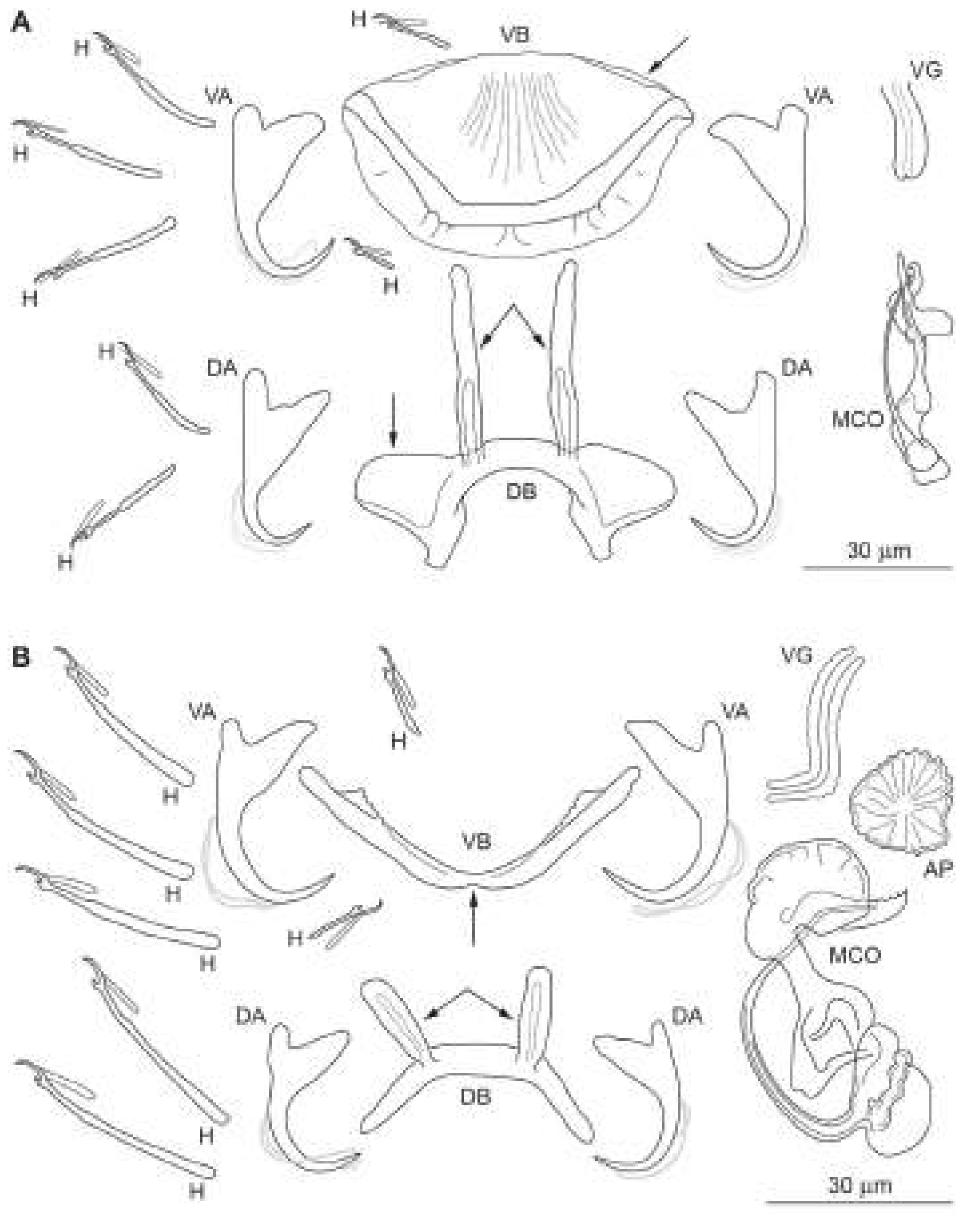


Fig. 4.4.15. Monogenea (Dactylogyridae). **A.** *Scutogyrus minus* (Dossou, 1982) from *Oreochromis niloticus*; **B.** *Cichlidogyrus gallus* Pariselle et Euzet, 1995 from *Coptodon guineensis*. (Modified from Pariselle & Euzet, 1995a,b.) VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ; AP = accessory plate.

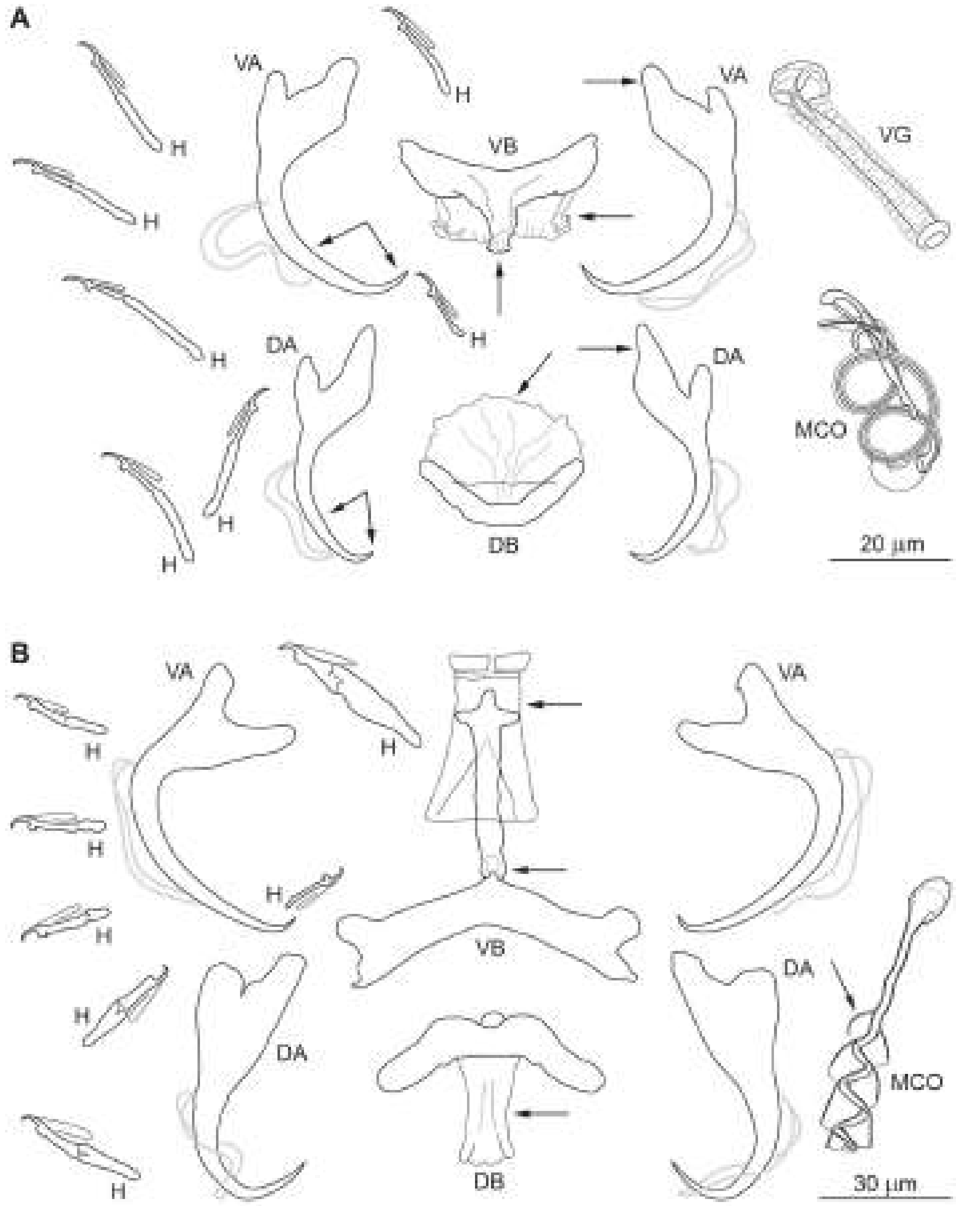


Fig. 4.4.16. Monogenea (Dactylogyridae). **A.** *Annulotrema uncata* Řehulková, Musilová et Gelnar, 2014 from *Hydrocynus brevis*. **B.** *Bagrobdella auchenoglanii* Paperna, 1969 from *Auchenoglanis occidentalis*. (Modified from Řehulková *et al.* 2014.) VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ.

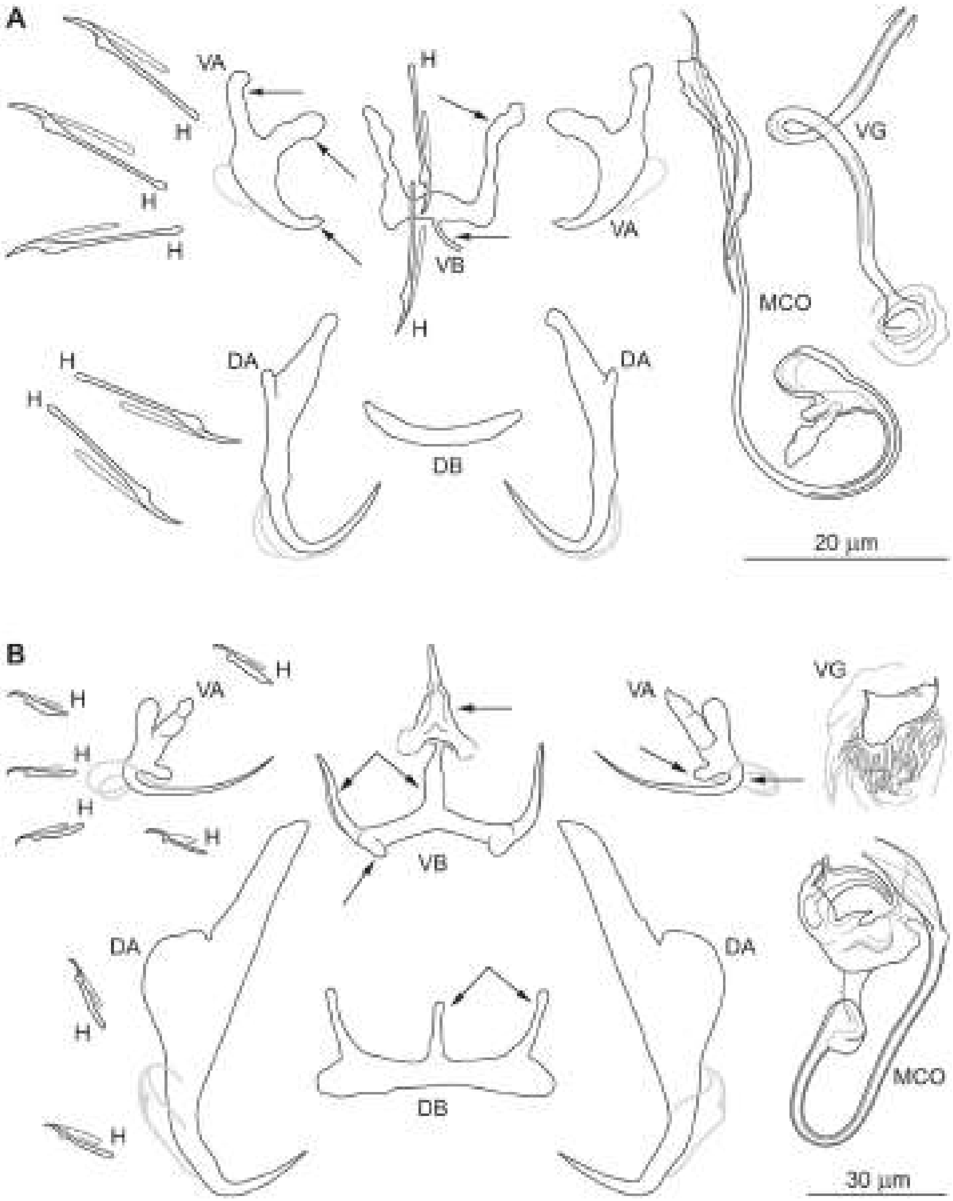


Fig. 4.4.17. Monogenea (Dactylogyridae). **A.** *Characidotrema nursei* Ergens, 1973 from *Brycinus nurse*; **B.** *Schilbetrema hexacornis* Paperna, 1969 from *Schilbe mystus*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ.

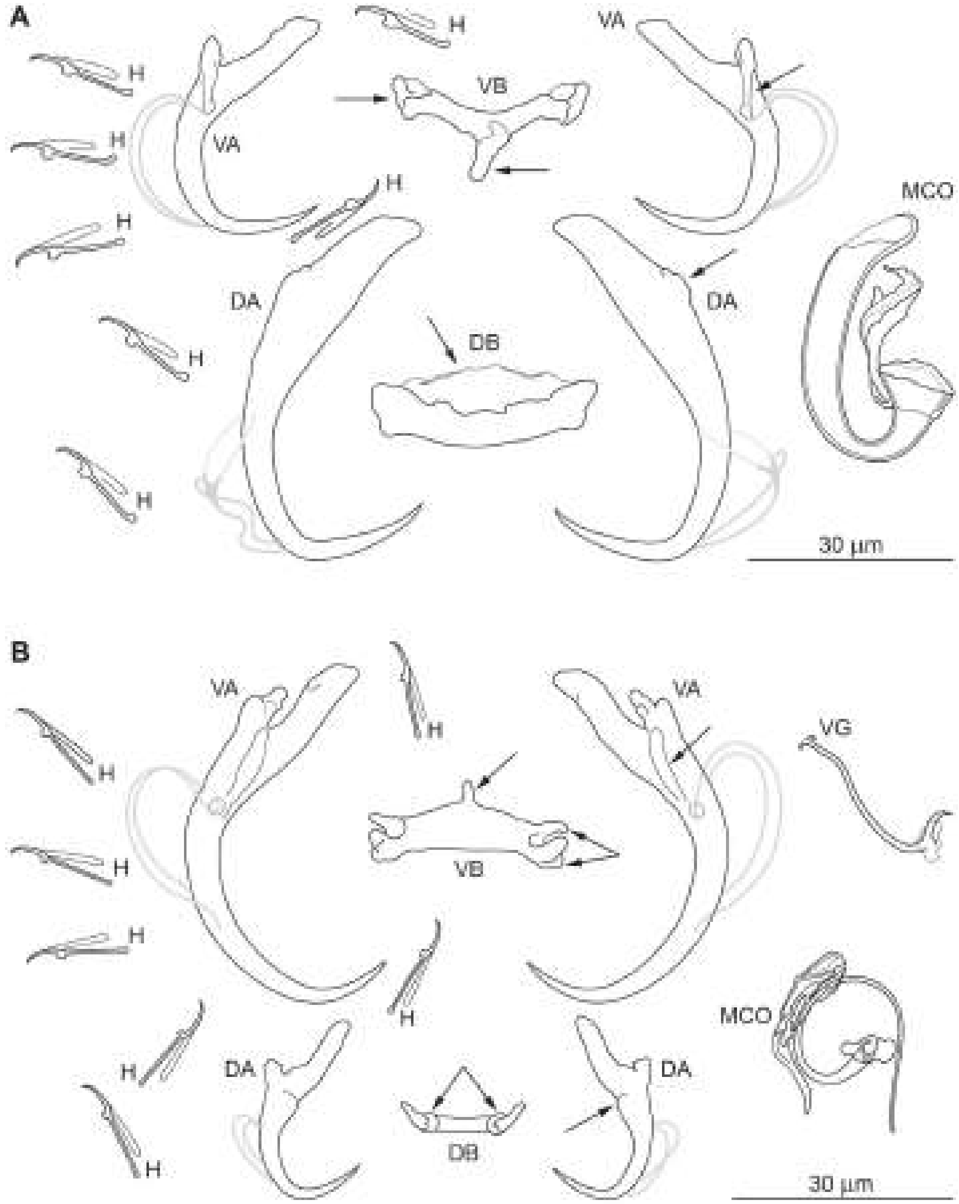


Fig. 4.4.18. Monogenea (Dactylogyridae). **A.** *Synodontella melanoptera* Dossou et Euzet, 1993 from *Synodontis melanopterus*; **B.** *Archidiplectanum archidiplectanum* Mizelle et Kritsky, 1969 from *Gnathonemus petersii*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ.

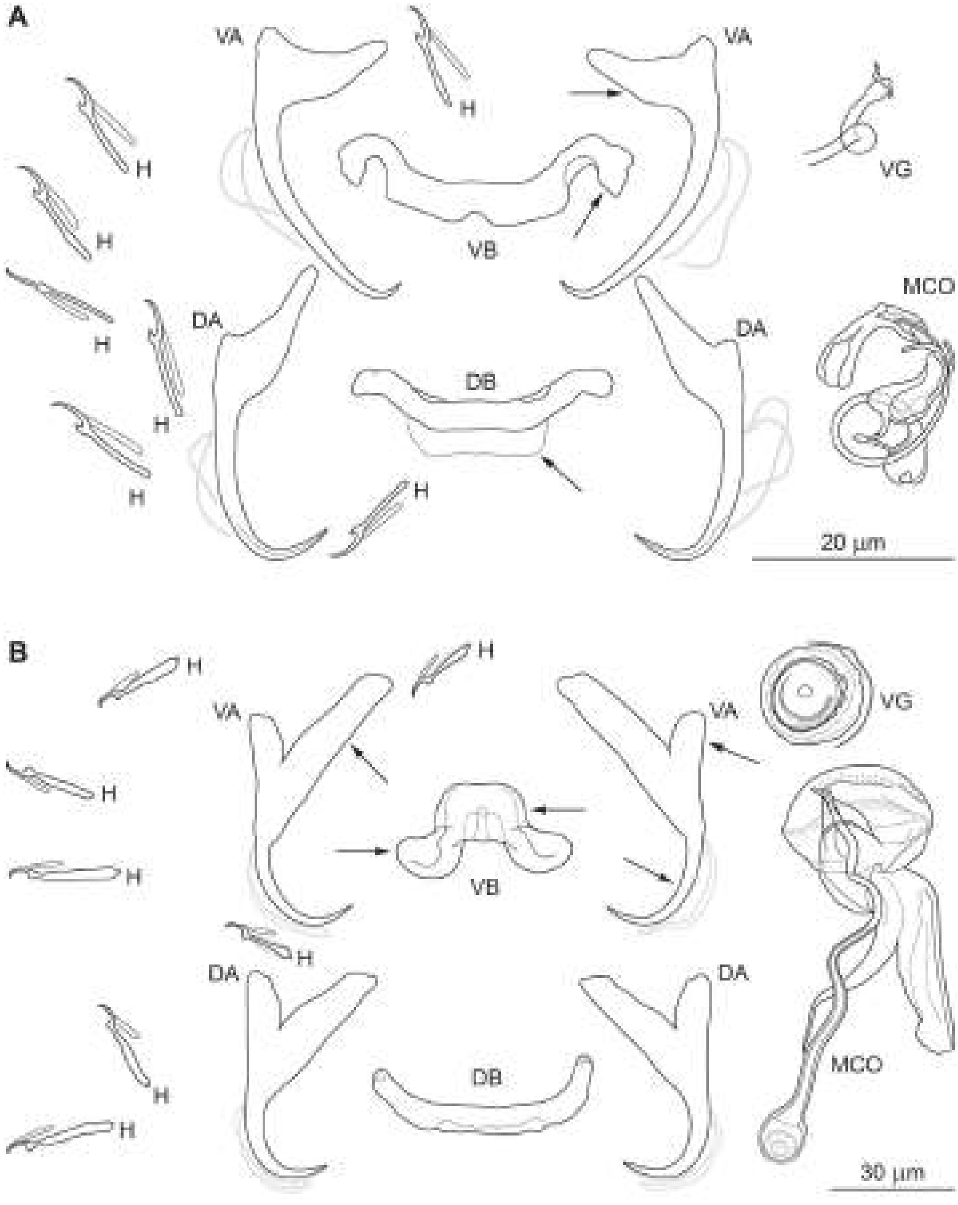


Fig. 4.4.19. Monogenea (Dactylogyridae). **A.** *Insulacleidus paratilapiae* Rakotofiringa et Euzet, 1983 from *Paratilapia polleni*; **B.** *Afrocleidodiscus paracleidodiscus* Paperna, 1973 from *Distichodus nefasch*. VA = ventral anchor; VB = ventral bar; DA = dorsal anchor; DB = dorsal bar; H = hook; VG = vagina; MCO = male copulatory organ.

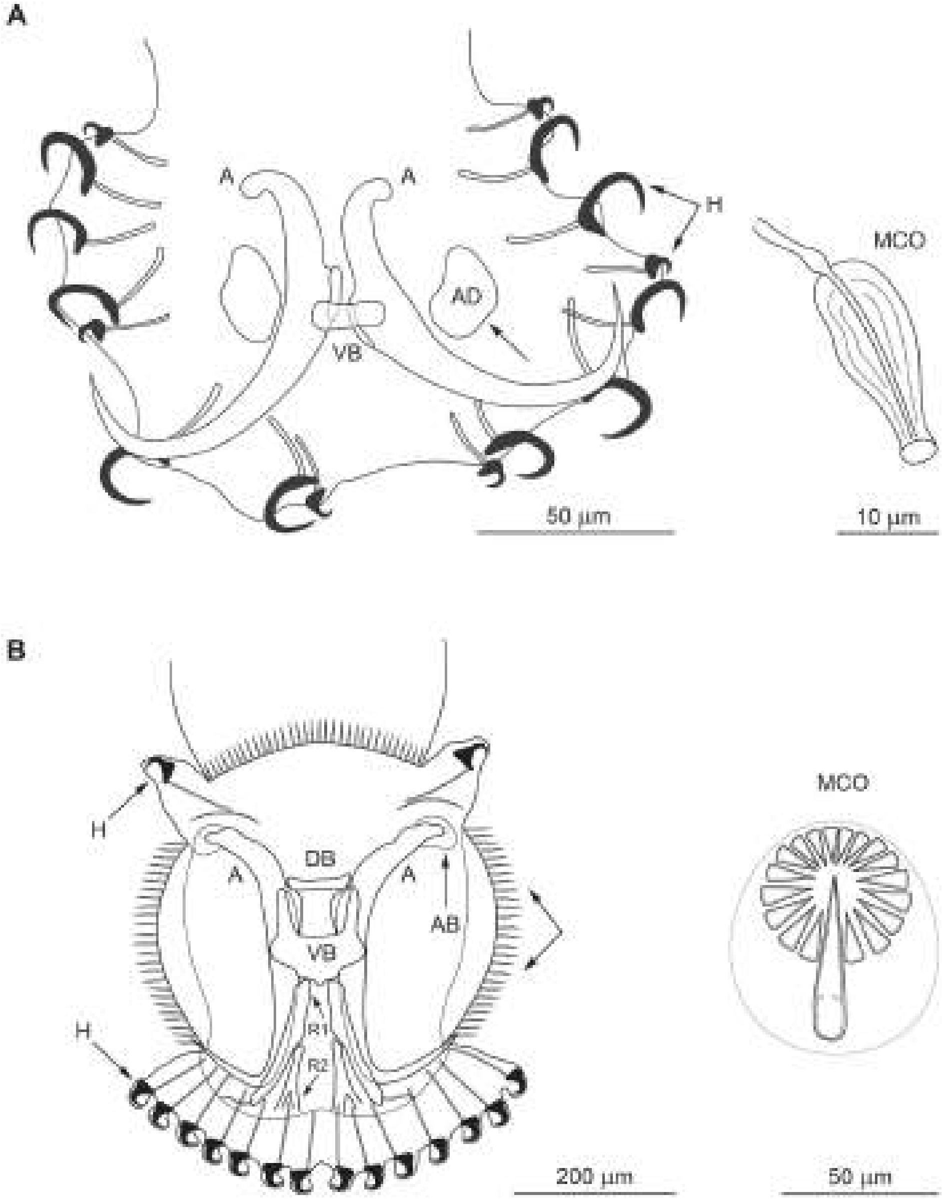


Fig. 4.4.20. Monogenea (Gyrodactylidae). **A.** *Diplogyrodactylus martini* Přikrylová, Matějusová, Musilová, Gelnar et Harris, 2009 from *Polypterus senegalus*; **B.** *Macrogyrodactylus congolensis* (Prudhoe, 1957) from *Clarias gariepinus*. (Modified from El-Naggar *et al.* 1999; Přikrylová *et al.* 2009.) A = anchor; AB = accessory bar, VB = ventral bar; R1, R2 = ventral bar rods; DB = dorsal bar; H = hook; AD = adhesive disc, MCO = male copulatory organ.

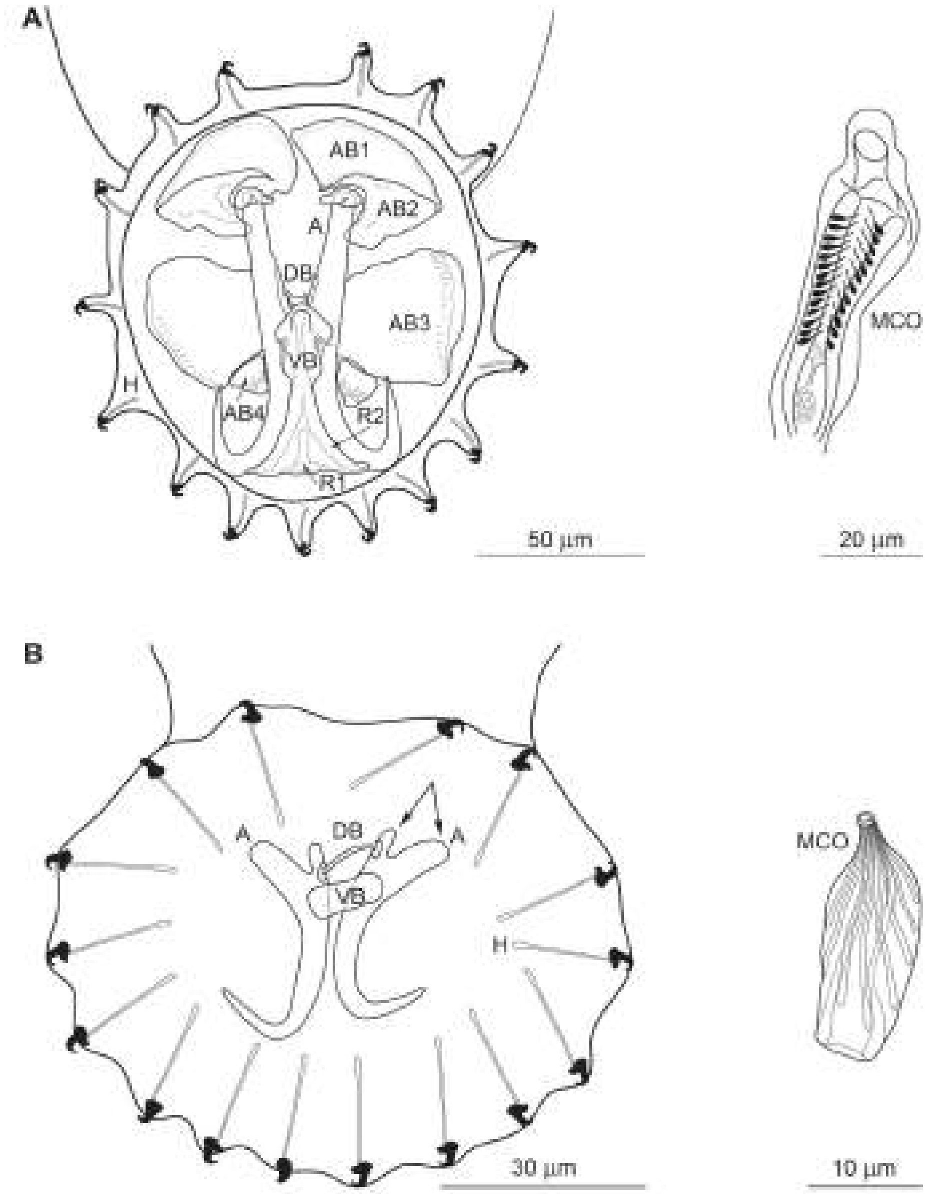


Fig. 4.4.21. Monogenea (Gyrodactylidae). **A.** *Mormyrogyrodactylus gemini* Luus-Powell, Mashego et Khalil, 2003 from *Marcusenius macrolepidotus*; **B.** *Afrogyrodactylus kingi* Přikrylová et Luus-Powell, 2014 from *Micralestes acutidens*. (Modified from Luus-Powell *et al.* 2003; Vianna *et al.* 2007; Přikrylová & Luus-Powell 2014.) A = anchor; VB = ventral bar; R1, R2 = ventral bar rods; DB = dorsal bar; H = hook; AB1–AB4 = accessory bars; MCO = male copulatory organ.

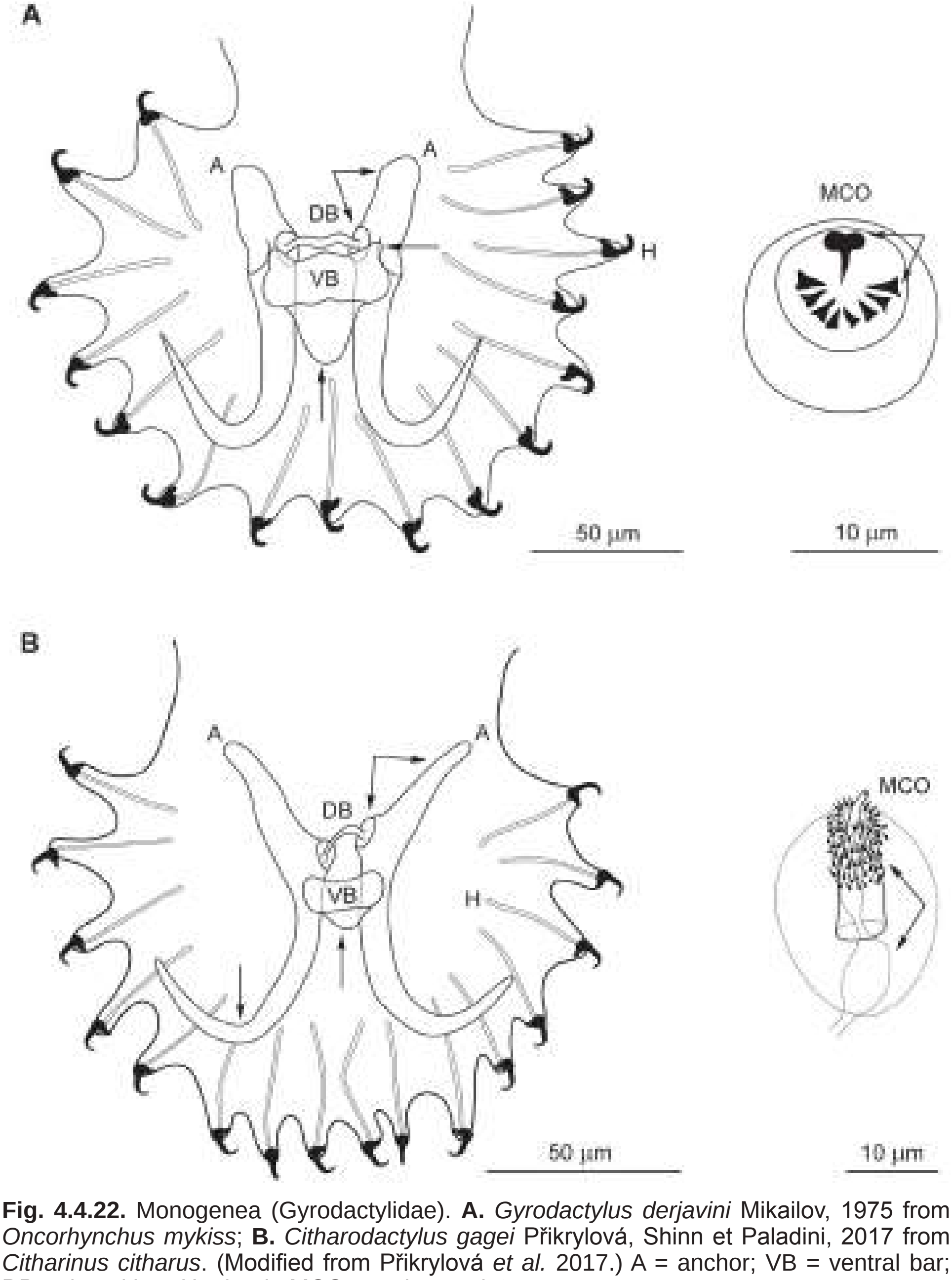


Fig. 4.4.22. Monogenea (Gyrodactylidae). **A.** *Gyrodactylus derjavini* Mikailov, 1975 from *Oncorhynchus mykiss*; **B.** *Citharodactylus gagei* Přikrylová, Shinn et Paladini, 2017 from *Citharinus citharus*. (Modified from Přikrylová *et al.* 2017.) A = anchor; VB = ventral bar; DB = dorsal bar; H = hook; MCO = male copulatory organ.

4.4.2. A systematic survey of monogeneans on/in African freshwater fishes

Knowledge of monogeneans in Africa is incomplete and the present numbers of these parasites on/in fishes in the region are likely to be an underestimate, as relatively few species of fishes have been examined. Today, more than twenty years since the last compilation (Khalil & Polling 1997), a total of 475 species of polyonchoineans (33 genera in 3 families) and 7 species of oligonchoineans (3 genera in 2 families) have been described. Given the high diversity of freshwater fish species in Africa (more than 3,200 spp.), monogeneans are therefore still poorly known. At the family level, the Dactylogyridae are represented currently by the greatest number of species (423 spp.) belonging to 26 genera: *Afrocleidodiscus* (3), *Ancyrocephalus* (s.l.) (4), *Annulotrema* (48), *Archidiplectanum* (1), *Bagrobdella* (4), *Birgiellus* (3), *Bouixella* (10), *Characidotrema* (10), *Cichlidogyrus* (112), *Dactylogyrus* (100), *Dogielius* (21), *Enterogyrus* (8), *Eutrianchoratus* (5), *Heteronchocleidus* (5), *Heterotesia* (1), *Insulacleidus* (3), *Nanotrema* (2), *Onchobdella* (8), *Paraquadriacanthus* (1), *Protoancylodiscoides* (9), *Quadriacanthus* (34), *Schilbetrema* (14), *Schilbetrematoides* (2), *Scutogyrus* (7), *Synodontella* (7), and *Urogyrus* (1). Only a single species of diplectanids, *Diplectanum lacustre*, has been reported from *Lates niloticus*. The viviparous gyrodactylids are the second-largest family, with 51 species attributed to 6 genera, *i.e.*, *Afrogyrodactylus* (4), *Citharodactylus* (1), *Diplogyrodactylus* (1), *Gyrodactylus* (35), *Macrogyrodactylus* (9), and *Mormyrogyrodactylus* (1). The only representatives of the Oligonchoinea are 7 species belonging to *Paradiplozoon* (5), *Afrodiplozoon* (1) (Diplozoidae) and one species of *Heterobothrium* (Diclidophoridae).

The catalogue of monogenean species described from freshwater fishes in Africa has been compiled from various sources (*e.g.*, Paperna 1979; Khalil & Polling 1997; Lim *et al.* 2001; Pariselle & Euzet 2009). Mention of the monogenean species in the list does not imply that the authors agree with their validity or taxonomy. Species are listed alphabetically according to individual monogenean genera. The type species of genera and type host of parasite species are highlighted in bold. The country where the type locality lies is also provided if known. Names of hosts recorded here are those provided in FishBase (Froese & Pauly 2017); names used in the original description are retained in square brackets as synonyms.

HETERONCHOINEA: OLIGONCHOINEA Bychowsky, 1937

DICLIDOPHORIDAE Cerfontaine, 1895

Heterobothrium Cerfontaine, 1895

Heterobothrium fluviatilis Euzet et Birgi, 1975 from ***Tetraodon lineatus*** [syn. *Tetraodon fahaka*] (Chad) [Fig. 4.4.3]

DIPLOZOIDAE Tripathi, 1959

Afrodiplozoon Khotenovsky, 1981

Afrodiplozoon polycotyleus (Paperna, 1973) [syn. *Neodiplozoon polycotyleus* Paperna, 1973] from *Alestes baremoze*, *Enteromius cercops*, *E. kerstenii*, ***E. neefi***, *E. neumayeri*, *E. paludinosus*, *E. trimaculatus*, ***Labeo victorianus*** (Kenya), *Labeobarbus macrolepis*, *L. marequensis* [Fig. 4.4.4B]

Paradiplozoon Akhmerov, 1974

Paradiplozoon aegyptense (Fischthal et Kuntz, 1963) [syn. *Diplozoon aegyptensis* Fischthal et Kuntz, 1963] from *Brycinus macrolepidotus*, *Enteromius paludinosus*, *Labeo coubie*, *L. cylindricus*, ***L. forskalii*** (Egypt), *L. victorianus*, *Raiamas senegalensis* [syn. *Barilius loati*]

Paradiplozoon ghanense (Thomas, 1957) [syn. *Diplozoon ghanense* Thomas, 1957] from *Alestes baremoze*, ***Brycinus macrolepidotus*** (Ghana) [Fig. 4.4.4A]

Paradiplozoon ichthyoxanthon Avenant-Oldewage 2014 from ***Labeobarbus aeneus*** (South Africa)

Paradiplozoon krugerense Dos Santos et Avenant-Oldewage, 2016 from ***Labeo congoro*** (South Africa), *L. rosae*

Paradiplozoon vaalense Dos Santos, Jansen van Vuuren et Avenant-Oldewage, 2015 from *Labeo capensis*, ***L. umbratus*** (South Africa)

POLYONCHOINEA Bychowsky, 1937

DACTYLOGYRIDAE Bychowsky, 1933

Afrocleidodiscus Paperna, 1969

Afrocleidodiscus distichodis Paperna, 1969 from ***Distichodus rostratus*** (Ghana)

Afrocleidodiscus hydrocynuous Paperna, 1969 from ***Hydrocynus*** sp. (Ghana)

Afrocleidodiscus paracleidodiscus Paperna, 1973 from ***Distichodus nefasch*** [syn. *Distichodus niloticus*] (Uganda) [Fig. 4.4.19B]

Ancyrocephalus Creplin, 1839 *sensu lato*

Ancyrocephalus barilii Paperna, 1973 from ***Raiamas senegalensis*** [syn. *Barilius loati*] (Uganda), *Barilius* sp.

Ancyrocephalus claveaui Birgi, 1988 from ***Poropanchax luxophthalmus*** [syn. *Aplocheilichthys macrophthalmus*] (Cameroon)

Ancyrocephalus limnotrissae Paperna, 1973 from ***Limnothrissa miodon*** (Tanzania)

Ancyrocephalus pellonulae Paperna, 1969 from ***Pellonula leonensis*** [syn. *Pellonula afzeliusi*] (Ghana)

Annulotrema Paperna et Thurston, 1969

Annulotrema alberti Paperna, 1973 from ***Brycinus macrolepidotus*** (Uganda)

Annulotrema alestesimberi Paperna, 1973 from ***Brycinus imberi*** Tanzania)

Annulotrema alestesnursi Paperna, 1973 from ***Brycinus nurse*** (Uganda)

Annulotrema allogravis Paperna, 1973 from ***Brycinus imberi*** (Tanzania)

Annulotrema amieti Birgi, 1988 from *Hemigrammopetersius pulcher*, ***Phenacogrammus major*** (Cameroon)

Annulotrema ansatum Kičinjaová et Řehulková, 2015 from ***Hydrocynus forskahlii*** (Kenya)

Annulotrema besalis Řehulková, Musilová et Gelnar, 2014 from ***Hydrocynus brevis*** (Senegal), *H. forskahlii*

Annulotrema biaensis N'Douba, Pariselle et Euzet, 1997 from ***Hepsetus odoe*** (Ivory Coast)

Annulotrema bilongi Birgi, 1988 from ***Neolebias trewavasae*** (Cameroon)

Annulotrema bipatens Kičinjaová, Řehulková et Blažek, 2015 from ***Hydrocynus forskahlii*** (Kenya)

Annulotrema bouixi Birgi, 1988 from ***Brycinus kingsleyae*** (Cameroon)

Annulotrema bracteatum Kinčinjaová et Řehulková, 2018 from ***Hydrocynus vittatus*** (Zimbabwe)

Annulotrema combesi Birgi, 1988 from ***Brycinus kingsleyae*** (Cameroon)

Annulotrema cryptophallus Paperna, 1973 from ***Hydrocynus forskahlii*** (Uganda)

Annulotrema cucullatum Kičinjaová, Řehulková et Blažek, 2015 from ***Hydrocynus forskahlii*** (Kenya)

Annulotrema curvipenis Paperna, 1969 from ***Alestes baremoze*** (Ghana), *Hydrocynus forskahlii*

Annulotrema delta Paperna, 1973 from ***Brycinus nurse*** (Uganda)

Annulotrema edeensis Birgi, 1988 from ***Micralestes*** sp. (Cameroon)

Annulotrema elongata Paperna et Thurston, 1969 from ***Alestes baremoze*** (Uganda), *A. dentex*, *Brycinus macrolepidotus*

Annulotrema endjami Birgi, 1988 from ***Neolebias trewavasae*** (Cameroon)

Annulotrema fomenai Birgi, 1988 from ***Neolebias trewavasae*** (Cameroon)

Annulotrema gabrioni Birgi, 1988 from *Hemigrammopetersius pulcher*, ***Phenacogrammus major*** (Cameroon)

Annulotrema gracilis (Wedl, 1861) [syns *Dactylogyrus gracilis* Wedl, 1861; *Neodactylogyrus gracilis* (Wedl, 1861)] from ***Hydrocynus forskahlii*** [syn. *Hydrocyon dentex*] (Egypt)

Annulotrema gravis Paperna et Thurston, 1969 from *Brycinus jacksonii*, ***B. nurse*** (Uganda)

Annulotrema helicocirra Paperna, 1973 from ***Brycinus macrolepidotus*** (Uganda)

Annulotrema hepseti Paperna et Thurston, 1969 from *Hepsetus cuvieri*, ***H. odoe*** (Ghana)

Annulotrema hydrocynusi Paperna, 1973 from ***Hydrocynus forskahlii*** (Uganda)

Annulotrema kribiensis Birgi, 1988 from ***Brycinus longipinnis*** (Cameroon)

Annulotrema lamberti Birgi, 1988 from ***Brycinus longipinnis*** (Cameroon)

Annulotrema longipenis Paperna, 1969 from *Alestes baremoze*, ***Brycinus macrolepidotus*** (Ghana), *Hydrocynus brevis*, *H. forskahlii*, *H. vittatus*

Annulotrema macropenis N'Douba, Pariselle et Euzet, 1997 from ***Hepsetus odoe*** (Ivory Coast)

Annulotrema magna Paperna, 1973 from ***Hydrocynus vittatus*** (Tanzania)

Annulotrema magnihamula Paperna, 1973 from ***Hydrocynus forskahlii*** (Uganda)

Annulotrema maillardi Birgi, 1988 from ***Brycinus kingsleyae*** (Cameroon)

Annulotrema moanko Birgi, 1988 from ***Brycinus longipinnis*** (Cameroon)

Annulotrema nannaethiopis Birgi, 1988 from ***Nannaethiops unitaeniatus*** (Cameroon)

Annulotrema nili Paperna, 1973 from *Hydrocynus brevis*, ***H. forskahlii*** (Uganda), *H. vittatus*

Annulotrema noyongensis Birgi, 1988 from ***Brycinus kingsleyae*** (Cameroon)

Annulotrema pikei (Price, Peebles et Bamford, 1969) [syns *Cleidodiscus pikei* Price, Peebles et Bamford, 1969; *Annulotrema armorata* Paperna, 1969] from *Hydrocynus forskahlii*, ***H. vittatus*** (South Africa)

Annulotrema pikoides Guégan, Lambert et Birgi, 1988 from ***Hydrocynus vittatus*** (Mali)

Annulotrema pontile Kičinjaová et Řehulková, 2015 [syn. *Annulotrema pikei* of Paperna (1979)] from ***Hydrocynus forskahlii*** (Kenya)

Annulotrema pseudonili Kičinjaová et Řehulková, 2018 from ***Hydrocynus vittatus*** (Zimbabwe)

Annulotrema robusta Paperna, 1969 from ***Brycinus leuciscus*** (Ghana)

Annulotrema ruahae Paperna, 1973 from ***Hydrocynus vittatus*** (Tanzania)

Annulotrema sangmelinensis Birgi, 1988 from ***Micralestes humilis*** (Cameroon)

Annulotrema spiropenis Paperna, 1969 from ***Brycinus nurse*** (Ghana), *Hydrocynus forskahlii*

Annulotrema tenuicirra Paperna, 1973 from ***Brycinus macrolepidotus*** (Uganda)

Annulotrema uncata Řehulková, Musilová et Gelnar, 2014 from ***Hydrocynus brevis*** (Senegal) [Fig. 4.4.16A]

Archidiplectanum Mizelle et Kritsky, 1969

Archidiplectanum archidiplectanum Mizelle et Kritsky, 1969 from ***Gnathonemus petersii*** (Western Africa) [Fig. 4.4.18B]

Bagrobdella Paperna, 1969

Bagrobdella anthopenis Euzet et Le Brun, 1990 from ***Auchenoglanis occidentalis*** (Mali)

Bagrobdella auchenoglanii Paperna, 1969 from ***Auchenoglanis occidentalis*** (Ghana) [Fig. 4.4.16B]

Bagrobdella fraudulenta Euzet et Le Brun, 1990 [syn. *Bagrobdella auchenoglanii* of Paperna, 1971] from ***Auchenoglanis occidentalis*** (Mali or Uganda; type locality not indicated)

Bagrobdella parauchenoglanii Akoumba, Pariselle et Tombi, 2017 from ***Parauchenoglanis monkei*** (Cameroon)

Birgiellus Bilong Bilong, Nack et Euzet 2007

Birgiellus calaris Bilong Bilong, Nack et Euzet, 2007 from ***Clarias jaensis*** (Cameroon)

Birgiellus kellensis Bilong Bilong, Nack et Euzet, 2007 from ***Clarias camerunensis*** (Cameroon)

Birgiellus mutatus Bilong Bilong, Nack et Euzet, 2007 from ***Clarias pachynema*** (Cameroon) [Fig. 4.4.14A]

Bouixella Euzet et Dossou, 1976

Bouixella beninensis Euzet et Dossou, 1976 from ***Mormyrus rume*** (Benin)

Bouixella deliciosa Dossou et Euzet, 1984 from ***Mormyrops anguilloides*** [syn. *Mormyrops deliciosus*] (Benin)

Bouixella furcillata Dossou et Euzet, 1984 from ***Marcusenius senegalensis*** (Benin)

Bouixella gorei Blahoua, Pariselle, N'Douba, Kone et Kouassi, 2009 from ***Mormyrus rume*** (Ivory Coast)

Bouixella koutouani Blahoua, Pariselle, N'Douba, Kone et Kouassi, 2009 from ***Mormyrus rume*** (Ivory Coast)

Bouixella mormyris (Paperna, 1973) [syn. *Ancyrocephalus mormyris* Paperna, 1973] from ***Mormyrus niloticus*** (Uganda)

Bouixella mormyrume Euzet et Dossou, 1976 from *Mormyrus kannume* (new host record), ***M. rume*** (Benin) [Fig. 4.4.11A]

Bouixella pusilla Dossou et Euzet, 1984 from ***Brienomyrus brachyistius*** (Benin)

Bouixella torta Dossou et Euzet, 1984 from ***Mormyrops anguilloides*** [syn. *Mormyrops deliciosus*] (Benin)

Bouixella yaoi Blahoua, Pariselle, N'Douba, Kone et Kouassi, 2009 from ***Mormyrus rume*** (Ivory Coast)

Characidotrema Paperna et Thurston, 1968

Characidotrema brevipenis Paperna, 1969 [syn. *Jainus brevipenis* (Paperna, 1969) Paperna, 1979] from *Alestes baremoze*, ***Brycinus nurse*** (Ghana)

Characidotrema elongata Paperna et Thurston, 1968 [syn. *Jainus elongatus* (Paperna et Thurston, 1968) Paperna, 1979] from ***Brycinus jacksonii*** (Uganda), *Brycinus leuciscus*

Characidotrema nursei Ergens, 1973 [syns *Jainus longipenis* Paperna, 1973; *Jainus nursei* (Ergens, 1973) Paperna, 1979] from *Alestes dentex*, *Brycinus leuciscus*, ***Brycinus nurse*** (Egypt) [Fig. 4.4.17A]

Characidotrema nzoiae (Paperna, 1979) [syn. *Jainus brevipenis nzoiae* Paperna, 1979] from ***Brycinus jacksonii*** (Kenya)

Characidotrema regia Birgi, 1988 from ***Brycinus kingsleyae*** (Cameroon)

Characidotrema ruahae (Paperna, 1979) [syn. *Jainus brevipenis ruahae* Paperna, 1979] from ***Brycinus imberi*** (Tanzania)

Characidotrema spinivaginus (Paperna, 1973) [syn. *Jainus spinivaginus* Paperna, 1973] from ***Brycinus nurse*** (Uganda)

Characidotrema spiropenis Birgi, 1988 from *Phenacogrammus major*, *P. urotaenia, Hemigrammopetersius pulcher* (Cameroon; type host not indicated)

Characidotrema undifera Kritsky, Kulo et Boeger, 1987 from ***Brycinus* cf. *nurse*** (Togo)

Characidotrema zelotes Kritsky, Kulo et Boeger, 1987 from ***Brycinus* cf. *nurse*** (Togo)

Cichlidogyrus Paperna, 1960

Cichlidogyrus acerbus Dossou, 1982 from *Sarotherodon galilaeus*, ***S. melanotheron*** (Benin)

Cichlidogyrus aegypticus Ergens, 1981 from *Coptodon camerunensis*, *C. dageti*, *C. guineensis*, *C. gutturosa*, *C. kottae*, *C. louka*, *C. walteri*, ***C. zillii*** (Egypt), *Oreochromis niloticus*, *Sarotherodon galilaeus*

Cichlidogyrus agnesi Pariselle et Euzet, 1995 from ***Coptodon guineensis*** (Ivory Coast)

Cichlidogyrus albareti Pariselle et Euzet, 1998 from ***Tilapia brevimanus*** (Guinea)

Cichlidogyrus amieti Birgi et Euzet, 1983 from ***Aphyosemion cameronense*** [syn. *Aphyosemion obscurum*] (Cameroon), *A. exiguum*

Cichlidogyrus amphoratus Pariselle et Euzet, 1996 from *Coptodon guineensis*, ***C. louka*** (Guinea)

Cichlidogyrus anthemocolpos Dossou, 1982 from *Coptodon camerunensis*, *C. guineensis*, ***C. zillii*** (Benin)

Cichlidogyrus arfii Pariselle et Euzet, 1995 [syn. *Cichlidogyrus dionchus* Paperna, 1968 (*partim*)] from ***Pelmatochromis buettikoferi*** (Guinea)

Cichlidogyrus arthracanthus Paperna, 1960 from *Coptodon camerunensis*, *C. coffea*, *C. dageti*, *C. deckerti*, *C. guineensis*, *C. gutturosa*, *C. kottae*, *C. walteri*, ***C. zillii***, *Oreochromis niloticus*, *Sarotherodon galilaeus*, *Tilapia* sp.

Cichlidogyrus aspiralis Rahmouni, Vanhove et Šimková, 2017 from ***Ophthalmotilapia nasuta*** (Burundi)

Cichlidogyrus attenboroughi Kmentová, Gelnar, Koblmüller et Vanhove, 2016 from ***Benthochromis horii*** (Burundi)

Cichlidogyrus banyankimbonai Pariselle et Vanhove, 2015 from ***Simochromis diagramma*** (Democratic Republic of the Congo)

Cichlidogyrus berminensis Pariselle, Bitja Nyom et Bilong Bilong, 2013 from *Coptodon bakossiorum*, ***C. bemini*** (Cameroon), *C. gutturosa*, *C. thysi*

Cichlidogyrus berradae Pariselle et Euzet, 2003 from *Coptodon camerunensis*, *C. guineensis*, ***Pelmatolapia cabrae*** (Cabinda, Angola)

Cichlidogyrus berrebii Pariselle et Euzet, 1994 from ***Tylochromis jentinki*** (Ivory Coast)

Cichlidogyrus bifurcatus Paperna, 1960 from *Haplochromis aeneocolor*, *H. elegans*, *H. limax*, *H. squamipinnis*, *Oreochromis aureus*, *Pseudocrenilabrus multicolor*

Cichlidogyrus bilongi Pariselle et Euzet, 1995 from *Coptodon camerunensis*, ***C. guineensis*** (Guinea)

Cichlidogyrus bonhommei Pariselle et Euzet, 1998 from ***Heterotilapia buttikoferi*** (Sierra Leone)

Cichlidogyrus bouvii Pariselle et Euzet, 1997 from ***Sarotherodon occidentalis*** (Guinea)

Cichlidogyrus brunnensis Kmentová, Gelnar, Koblmüller et Vanhove, 2016 from ***Trematocara unimaculatum*** (Burundi)

Cichlidogyrus buescheri Pariselle et Vanhove, 2015 from ***Interochromis loocki*** (Zambia)

Cichlidogyrus bychowskii (Markevich, 1934) [syn. *Ancyrocephalus bychowskii* Markevitch, 1934] from ***Hemichromis bimaculatus***, *H. fasciatus*, *Sarotherodon galilaeus*

Cichlidogyrus casuarinus Pariselle, Muterezi Bukinga et Vanhove, 2015 from *Bathybates fasciatus*, *B. hornii*, *B. leo*, ***B. minor*** (Democratic Republic of the Congo), *B. vittatus*, *Hemibates stenosoma*

Cichlidogyrus centesimus Vanhove, Volckaert et Pariselle, 2011 from *Ophthalmotilapia boops*, *O. nasuta*, ***O. ventralis*** (Zambia)

Cichlidogyrus chrysopiformis Pariselle, Bitja Nyom et Bilong Bilong, 2014 from ***Tylochromis sudanensis*** (Cameroon)

Cichlidogyrus cirratus Paperna, 1964 [syn. *Cichlidogyrus nematocirrus* Paperna, 1969] from *Coptodon zillii*, *Oreochromis esculentus*, *O. mweruensis*, *O. niloticus*, *O. variabilis*, ***Sarotherodon galilaeus***

Cichlidogyrus consobrini Jorissen, Pariselle et Vanhove, 2018 from *Orthochromis katumbii*, ***Sargochromis mellandi*** (Democratic Republic of the Congo)

Cichlidogyrus cubitus Dossou, 1982 from *Coptodon camerunensis*, *C. dageti*, *C. guineensis*, *C. louka*, *C. walteri*, ***C. zillii*** (Benin)

Cichlidogyrus dageti Dossou et Birgi, 1984 from ***Hemichromis fasciatus*** (Benin or Cameroon; type locality not indicated)

Cichlidogyrus digitatus Dossou, 1982 [syns *Cichlidogyrus* cf. *brevicirrus* of Paperna (1969); *Cichlidogyrus halinus* Paperna, 1969 (*partim*)] from *Coptodon camerunensis*, *C. dageti*, *C. discolor*, *C. guineensis*, *C. louka*, *C. walteri*, ***C. zillii*** (Benin), *Pelmatolapia mariae*, *Tilapia brevimanus*

Cichlidogyrus dionchus Paperna, 1968 [syn. *Cichlidogyrus brevicirrus* Paperna et Thurston, 1969] from *Chromidotilapia guntheri*, *Coptodon discolor*, *C. zillii*, *Haplochromis guiarti*, *H. longirostris*, *H. obesus*, *H. obliquidens*, *H. retrodens*, *Hemichromis fasciatus*, ***Sarotherodon galilaeus*** (Ghana)

Cichlidogyrus discophonum Rahmouni, Vanhove et Šimková, 2017 from ***Aulonocranus dewindti*** (Burundi)

Cichlidogyrus djietoi Pariselle, Bitja Nyom et Bilong Bilong, 2014 from ***Tylochromis sudanensis*** (Cameroon)

Cichlidogyrus dossoui Douëllou, 1993 from *Coptodon camerunensis*, *C. guineensis*, ***C. rendalli*** (Zimbabwe), *Oreochromis mortimeri*, *O. mossambicus*, *O. mweruensis*, *Serranochromis macrocephalus*, *Tilapia sparrmanii*

Cichlidogyrus douellouae Pariselle, Bilong Bilong et Euzet, 2003 from ***Sarotherodon galilaeus*** (Nigeria)

Cichlidogyrus dracolemma Řehulková, Mendlová et Šimková, 2013 from ***Hemichromis letourneuxi*** (Senegal)

Cichlidogyrus ergensi Dossou, 1982 from *Coptodon camerunensis*, *C. guineensis*, ***C. zillii*** (Benin), *Pelmatolapia mariae*

Cichlidogyrus euzeti Dossou et Birgi, 1984 from ***Hemichromis fasciatus*** (Benin or Cameroon; type locality not indicated)

Cichlidogyrus evikae Rahmouni, Vanhove et Šimková, 2017 from ***Tanganicodus irsacae*** (Burundi)

Cichlidogyrus falcifer Dossou et Birgi, 1984 [syn. *Cichlidogyrus dionchus* Paperna, 1968 (*partim*)] from ***Hemichromis fasciatus*** (Benin or Cameroon; type locality not indicated)

Cichlidogyrus flexicolpos Pariselle et Euzet, 1995 from *Coptodon dageti*, ***C. guineensis*** (Ivory Coast), *Pelmatolapia mariae*

Cichlidogyrus fontanai Pariselle et Euzet, 1997 from ***Sarotherodon occidentalis*** (Guinea)

Cichlidogyrus frankwillemsi Pariselle et Vanhove, 2015 from ***Pseudosimochromis curvifrons*** (Zambia)

Cichlidogyrus franswittei Pariselle et Vanhove, 2015 from *Pseudosimochromis curvifrons*, ***P. marginatus*** (Democratic Republic of the Congo)

Cichlidogyrus gallus Pariselle et Euzet, 1995 [syn. *Cichlidogyrus aegypticus* Ergens, 1981 (*partim*)] from ***Coptodon guineensis*** (Ivory Coast), *C. walteri*, *C. zillii* [Fig. 4.4.15B]

Cichlidogyrus georgesmertensi Pariselle et Vanhove, 2015 from ***Pseudosimochromis babaulti*** (Democratic Republic of the Congo)

Cichlidogyrus gillardinae Muterezi Bukinga, Vanhove, Van Steenberge et Pariselle, 2012 from ***Astatotilapia burtoni*** (Democratic Republic of the Congo)

Cichlidogyrus gillesi Pariselle, Bitja Nyom et Bilong Bilong, 2013 from ***Coptodon guineensis*** (Cameroon)

Cichlidogyrus giostrai Pariselle, Bilong Bilong et Euzet, 2003 from ***Sarotherodon caudomarginatus*** (Guinea)

Cichlidogyrus gistelincki Gillardin, Vanhove, Pariselle, Huyse et Volckaert, 2012 from ***Ctenochromis horei*** (Zambia)

Cichlidogyrus glacicremoratus Rahmouni, Vanhove et Šimková, 2017 from ***Ophthalmotilapia nasuta*** (Burundi)

Cichlidogyrus guirali Pariselle et Euzet, 1997 from ***Sarotherodon occidentalis*** (Guinea)

Cichlidogyrus halinus Paperna, 1969 [syn. *Cichlidogyrus erectus* Dossou, 1982] from *Coptodon guineensis*, ***Sarotherodon melanotheron*** [syn. *Tilapia heudeloti*] (Ghana)

Cichlidogyrus halli (Price et Kirk, 1967) [syns *Cleidodiscus halli* Price et Kirk, 1967; *Cichlidogyrus tubicirrus magnus* Paperna et Thurston, 1969; *Cichlidogyrus magnus* Paperna et Thurston, 1969; *Cichlidogyrus halli typicus* Paperna, 1979; *Cichlidogyrus halli victorianus* Paperna, 1979] from *Oreochromis esculentus*, *O. leucostictus*, *O. mortimeri*, *O. mweruensis*, *O. niloticus* x *mweruensis*, ***O. shiranus*** (Malawi), *O. spilurus*, *O. tanganicae*, *O. variabilis*, *Sarotherodon galilaeus*, *S. melanotheron*, *S. occidentalis*, *Serranochromis macrocephalus*

Cichlidogyrus haplochromii Paperna et Thurston, 1969 [*Cichlidogyrus dionchus* Paperna, 1968 (*partim*); *Cichlidogyrus tubicirrus minutus* Paperna et Thurston, 1969 (*partim*); *Cichlidogyrus tubicirrus* Paperna et Thurston, 1969 (*partim*)] from *Haplochromis aeneocolor*, *H. angustifrons*, *H. bicolor*, *H. degeni*, *H. elegans*, ***H. guiarti*** (Uganda), *H. limax*, *H. longirostris*, *H. macrognathus*, *H. macrops*, *H. nigripinnis*, *H. nubilus*, *H. obesus*, *H. obliquidens*, *H. petronius*, *H. retrodens*, *H. schubotzi*, *H. squamipinnis*, *Oreochromis leucostictus*, *Pharyngochromis darlingi*, *Thoracochromis wingatii*

Cichlidogyrus hemi Pariselle et Euzet, 1998 from ***Tilapia brevimanus*** (Guinea)

Cichlidogyrus inconsultans Birgi et Lambert, 1987 [syn. *Cichlidogyrus euzeti* Birgi et Lambert, 1986 renamed] from ***Polycentropsis abbreviata*** (Cameroon)

Cichlidogyrus irenae Gillardin, Vanhove, Pariselle, Huyse et Volckaert, 2012 from ***Gnathochromis pfefferi*** (Zambia)

Chichlidogyrus jeanloujustinei Rahmouni, Vanhove et Šimková, 2017 from ***Eretmodus marksmithi*** (Burundi)

Cichlidogyrus karibae Douëllou, 1993 from *Oreochromis mortimeri*, ***Sargochromis codringtonii*** (Zimbabwe)

Cichlidogyrus kothiasi Pariselle et Euzet, 1994 from ***Tylochromis jentinki*** (Ivory Coast)

Cichlidogyrus kouassii N'Douba, Thys van den Audenaerde et Pariselle, 1997 from ***Coptodon guineensis*** (Ivory Coast)

Cichlidogyrus lagoonaris Paperna, 1969 [syn. *Cichlidogyrus gibbus* Dossou, 1982] from *Coptodon guineensis*, ***Sarotherodon melanotheron*** (Ghana)

Cichlidogyrus legendrei Pariselle et Euzet, 2003 from ***Pelmatolapia cabrae*** (Cabinda, Angola)

Cichlidogyrus lemoallei Pariselle et Euzet, 2003 from ***Pelmatolapia cabrae*** (Republic of the Congo), *P. mariae*

Cichlidogyrus levequei Pariselle et Euzet, 1996 from ***Coptodon coffea*** (Guinea)

Cichlidogyrus longicirrus Paperna, 1965 from *Chromidotilapia guntheri*, ***Hemichromis fasciatus*** (Ghana)

Cichlidogyrus longipenis Paperna et Thurston, 1969 [syn. *Cichlidogyrus tubicirrus longipenis* Paperna et Thurston, 1969] from ***Astatoreochromis alluaudi*** (Uganda)

Cichlidogyrus louipaysani Pariselle et Euzet, 1995 from ***Coptodon guineensis*** (Guinea)

Cichlidogyrus makasai Vanhove, Volckaert et Pariselle, 2011 from *Ophthalmotilapia boops*, *O. nasuta*, ***O. ventralis*** (Zambia)

Cichlidogyrus mbirizei Muterezi Bukinga, Vanhove, Van Steenberge et Pariselle, 2012 from *Oreochromis niloticus*, *O. niloticus* × *mossambicus*, *O. niloticus* × *mweruensis*, ***O. tanganicae*** (Democratic Republic of the Congo)

Cichlidogyrus microscutus Pariselle et Euzet, 1996 from *Coptodon camerunensis*, *C. dageti*, ***C. guineensis*** (Republic of the Congo)

Cichlidogyrus milangelnari Rahmouni, Vanhove et Šimková, 2017 from ***Cyprichromis microlepidotus*** (Burundi)

Cichlidogyrus mulimbwai Muterezi Bukinga, Vanhove, Van Steenberge et Pariselle, 2012 from ***Tylochromis polylepis*** (Democratic Republic of the Congo)

Cichlidogyrus muterezii Pariselle et Vanhove, 2015 from ***Simochromis diagramma*** (Democratic Republic of the Congo)

Cichlidogyrus muzumanii Muterezi Bukinga, Vanhove, Van Steenberge et Pariselle, 2012 from ***Tylochromis polylepis*** (Democratic Republic of the Congo)

Cichlidogyrus mvogoi Pariselle, Bitja Nyom et Bilong Bilong, 2014 from ***Sarotherodon mvogoi*** (Cameroon)

Cichlidogyrus nageus Řehulková, Mendlová et Šimková, 2013 from *Coptodon guineensis*, ***Sarotherodon galilaeus*** (Senegal)

Cichlidogyrus nandidae Birgi et Lambert, 1986 from ***Polycentropsis abbreviata*** (Cameroon)

Cichlidogyrus njinei Pariselle, Bilong Bilong et Euzet, 2003 from ***Sarotherodon galilaeus*** (Cameroon)

Cichlidogyrus nshomboi Muterezi Bukinga, Vanhove, Van Steenberge et Pariselle, 2012 from ***Boulengerochromis microlepis*** (Democratic Republic of the Congo)

Cichlidogyrus nuniezi Pariselle et Euzet, 1998 from *Heterotilapia buttikoferi*, ***H. cessiana*** (Ivory Coast)

Cichlidogyrus ornatus Pariselle et Euzet, 1996 from *Coptodon camerunensis*, *C. dageti*, ***C. zillii*** (Ivory Coast)

Cichlidogyrus ouedraogoi Pariselle et Euzet, 1996 from ***Coptodon coffea*** (Guinea), *C. guineensis*, *C. kottae*, *Pelmatolapia mariae*

Cichlidogyrus paganoi Pariselle et Euzet, 1997 from ***Sarotherodon occidentalis*** (Guinea)

Cichlidogyrus papernastrema Price, Peebles et Bamford, 1969 from *Coptodon rendalli*, *Oreochromis mweruensis*, ***Tilapia sparrmanii*** (South Africa)

Cichlidogyrus philander Douëllou, 1993 from ***Pseudocrenilabrus philander*** (Zimbabwe)

Cichlidogyrus pouyaudi Pariselle et Euzet, 1994 from *Tylochromis intermedius*, ***T. jentinki*** (Ivory Coast)

Cichlidogyrus pseudoaspiralis Rahmouni, Vanhove et Šimková, 2017 from ***Aulonocranus dewindti*** (Burundi)

Cichlidogyrus quaestio Douëllou, 1993 from ***Coptodon rendalli*** (Zimbabwe), *Sargochromis codringtonii*, *Serranochromis macrocephalus*, *Tilapia sparrmanii*

Cichlidogyrus raeymaekersi Pariselle et Vanhove, 2015 from ***Simochromis diagramma*** (Democratic Republic of the Congo)

Cichlidogyrus rectangulus Rahmouni, Vanhove et Šimková, 2017 from ***Ophthalmotilapia nasuta*** (Burundi)

Cichlidogyrus reversati Pariselle et Euzet, 2003 from ***Pelmatolapia cabrae*** (Republic of the Congo)

Cichlidogyrus rognoni Pariselle, Bilong Bilong et Euzet, 2003 from ***Oreochromis niloticus*** (Senegal)

Cichlidogyrus sanjeani Pariselle et Euzet, 1997 from ***Sarotherodon occidentalis*** (Guinea)

Cichlidogyrus sanseoi Pariselle et Euzet, 2004 from ***Hemichromis fasciatus*** (Ivory Coast)

Cichlidogyrus schreyenbrichardorum Pariselle et Vanhove, 2015 from ***Interochromis loocki*** (Zambia)

Cichlidogyrus sclerosus Paperna et Thurston, 1969 [syn. *Cichlidogyrus bangladeshi* Ferdousi et Chandra, 2002] from *Coptodon zillii*, *Haplochromis* sp., *Oreochromis leucostictus*, *O. mortimeri*, ***O. mossambicus*** (Uganda), *O. mweruensis*, *O. niloticus*, *O. spilurus*, *Sarotherodon galilaeus*, *Serranochromis macrocephalus*

Cichlidogyrus sigmocirrus Pariselle, Bitja Nyom et Bilong Bilong, 2014 from ***Tylochromis sudanensis*** (Cameroon)

Cichlidogyrus slembroucki Pariselle et Euzet, 1998 from ***Heterotilapia buttikoferi*** (Guinea), *Pelmatolapia mariae*

Cichlidogyrus steenbergei Gillardin, Vanhove, Pariselle, Huyse et Volckaert, 2012 from ***Limnotilapia dardennii*** (Zambia)

Cichlidogyrus sturmbaueri Vanhove, Volckaert et Pariselle, 2011 from *Ophthalmotilapia nasuta*, ***O. ventralis*** (Zambia)

Cichlidogyrus testificatus Dossou, 1982 from ***Pelmatolapia mariae*** (Benin)

Cichlidogyrus teugelsi Pariselle et Euzet, 2004 from ***Hemichromis fasciatus*** (Ivory Coast)

Cichlidogyrus thurstonae Ergens, 1981 [syns *Cichlidogyrus tiberianus* Paperna, 1960 (*partim*); *Cichlidogyrus gilli* Ferdousi et Chandra, 2002] from *Haplochromis longirostris*, *Oreochromis esculentus*, ***O. niloticus*** (Egypt), *O. variabilis*, *Sarotherodon galilaeus*

Cichlidogyrus tiberianus Paperna, 1960 from *Coptodon bakossiorum*, *C. coffea*, *C. dageti*, *C. guineensis*, *C. gutturosa*, *C. kottae*, *C. rendalli*, *C. walteri*, ***C. zillii***, *Oreochromis mweruensis*, *Pelmatolapia mariae*, *Tilapia sparrmanii*

Cichlidogyrus tilapiae Paperna, 1960 [syns *Cleidodiscus tilapiae* Price et Géry, 1967; *Cichlidogyrus tubicirrus minutus* Paperna et Thurston, 1969 (*partim*); *Cichlidogyrus tubicirrus* Paperna et Thurston, 1969 (*partim*); *Cichlidogyrus chandrai* Ferdousi et Chandra, 2002] from *Chromidotilapia guntheri*, *Coptodon camerunensis*, *C. guineensis*, *C. gutturosa*, *C. kottae*, *C. zillii*, *Haplochromis macrognathus*, *Hemichromis fasciatus, Oreochromis aureus*, *O. leucostictus*, *O. mortimeri*, *O. mossambicus*, *O. mweruensis*, *O. niloticus*, *O. spilurus*, *O. urolepis*, *O. variabilis*, *Pelmatolapia mariae*, ***Sarotherodon galilaeus***, *Tilapia busumana*

Cichlidogyrus vandekerkhovei Vanhove, Volckaert et Pariselle, 2011 from *Ophthalmotilapia boops*, *O. nasuta*, ***O. ventralis*** (Zambia)

Cichlidogyrus vealli Pariselle et Vanhove, 2015 from ***Interochromis loocki*** (Zambia)

Cichlidogyrus vexus Pariselle et Euzet, 1995 from ***Coptodon guineensis*** (Ivory Coast), *C. zillii*

Cichlidogyrus yanni Pariselle et Euzet, 1996 from *Coptodon camerunensis*, *C. dageti*, *C. guineensis*, *C. louka*, *C. walteri*, ***C. zillii*** (Guinea)

Cichlidogyrus zambezensis Douëllou, 1993 from *Oreochromis mortimeri*, *Sargochromis mellandi*, *Serranochromis angusticeps*, ***S. macrocephalus*** (Zimbabwe), *S. robustus*, *S. stappersi*, *S. thumbergi*

Dactylogyrus Diesing, 1850 [syn. *Neodactylogyrus* Price, 1938]

Dactylogyrus afer Price et Géry, 1968 from ***Labeobarbus batesii*** (Gabon)

Dactylogyrus aferoides Guégan et Lambert, 1990 from ***Labeobarbus bynni*** [syns *Barbus occidentalis*, *B. waldroni*] (Mali), *L. parawaldroni*, *L. petitjeani*

Dactylogyrus afrobarbae Paperna, 1968 from *Enteromius ablabes*, *E. sublineatus*, *E. trispilopleura* (Ghana; type host not indicated)

Dactylogyrus afrochelatus Paperna, 1973 from ***Enteromius paludinosus*** [syn. *Barbus amphigramma*] (Kenya)

Dactylogyrus afrofluviatilis Paperna, 1973 from ***Barbus*** sp. (Kenya), *Enteromius neglectus*, *E. perince*

Dactylogyrus afrolongicornis Paperna, 1973 from ***Enteromius kerstenii*** (Uganda), *E. perince*, *E. trimaculatus*

Dactylogyrus afropsilovaginus Paperna, 1973 from *Enteromius kerstenii*, ***E. paludinosus*** [syn. *Barbus amphigramma*] (Uganda), *E. perince*

Dactylogyrus afroruahae Paperna, 1973 from ***Barbus*** sp. (Tanzania)

Dactylogyrus afrosclerovaginus Paperna, 1973 from *Enteromius magdalenae*, ***E. neglectus*** (Uganda), *E. paludinosus*, *E. perince*

Dactylogyrus afrotoxopous Paperna, 1973 from ***Enteromius kerstenii*** (Uganda)

Dactylogyrus allolongionchus Paperna, 1973 from ***Enteromius perince*** (Uganda), *E. trimaculatus*

Dactylogyrus amieti Birgi et Lambert, 1987 from ***Enteromius camptacanthus*** (Cameroon)

Dactylogyrus archaeopenis Guégan et Lambert, 1990 from ***Labeobarbus parawaldroni*** (Guinea), *L. petitjeani*, *L. sacratus*

Dactylogyrus aspili Birgi et Lambert, 1987 from ***Enteromius aspilus*** (Cameroon)

Dactylogyrus atlasensis El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus pallaryi*** (Morocco)

Dactylogyrus barbus Price et Géry, 1968 from ***Barbus*** sp. (*brichardi* aff.) (Gabon)

Dactylogyrus batesii Birgi et Euzet, 1983 from ***Aphyosemion batesii*** (Cameroon)

Dactylogyrus benhoussai Rahmouni, Řehulková et Šimková, 2017 from ***Luciobarbus moulouyensis*** (Morocco)

Dactylogyrus biradius Birgi et Lambert, 1987 from ***Enteromius jae*** (Cameroon)

Dactylogyrus birgii Timofeeva, Gerasev et Gibson, 1996 [syn. *Dactylogyrus simplex* Birgi et Lambert, 1987] from ***Enteromius martorelli*** (Cameroon)

Dactylogyrus bopeleti Birgi et Lambert, 1987 from ***Enteromius martorelli*** (Cameroon)

Dactylogyrus borjensis El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus nasus*** (Morocco)

Dactylogyrus brachydiscus Paperna, 1973 from ***Labeo victorianus*** (Kenya)

Dactylogyrus brevicirrus Paperna, 1973 from *Enteromius kerstenii*, *E. neglectus*, *E. perince*, *Labeo cylindricus*, *L. forskalii*, *L. parvus*, ***L. victorianus*** (Uganda), *Labeobarbus altianalis*, *Leptocypris niloticus*

Dactylogyrus brevicornis Paperna, 1973 from ***Enteromius kerstenii*** (Uganda)

Dactylogyrus clani Guégan et Lambert, 1990 from ***Labeobarbus petitjeani*** (Guinea)

Dactylogyrus clavatovaginus Paperna, 1973 from *Enteromius nyanzae*, ***E. paludinosus*** [syn. *Barbus amphigramma*] (Uganda)

Dactylogyrus cyclocirrus Paperna, 1973 from *Labeo coubie*, ***L. cylindricus*** (Tanzania), *L. senegalensis*, *L. victorianus*

Dactylogyrus decaspirus Guégan, Lambert et Euzet, 1988 [syns *Dactylogyrus senegalensis* Paperna, 1969 (*partim*); *Dactylogyrus* cf. *senegalensis* Paperna, 1979] from ***Labeo coubie*** (Ghana)

Dactylogyrus dembae Musilová, Řehulková et Gelnar, 2009 [syn. *Dactylogyrus* cf. *labeous* Paperna, 1979 (*partim*)] from ***Labeo coubie*** (Senegal)

Dactylogyrus digitalis Paperna, 1969 from ***Labeo coubie*** (Ghana)

Dactylogyrus dominici Mashego, 1983 from ***Enteromius paludinosus*** (South Africa)

Dactylogyrus draaensis El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus pallaryi*** (Morocco)

Dactylogyrus enidae Mashego, 1983 from ***Enteromius neefi*** (South Africa)

Dactylogyrus falcilocus Guégan, Lambert et Euzet, 1988 from ***Labeo coubie*** (Mali), *L. parvus*, *Labeobarbus wurtzi*

Dactylogyrus falsiphallus Rahmouni, Řehulková et Šimková, 2017 from ***Luciobarbus maghrebensis*** (Morocco)

Dactylogyrus fimbriphallus El Gharbi, Birgi et Lambert, 1994 from *Carasobarbus moulouyensis*, ***Luciobarbus callensis*** [syns *Barbus figuiensis*, *B. issenensis*, *B. massaensis*] (Morocco), *L. pallaryi* [syn. *Barbus lepineyi*]

Dactylogyrus gabonensis Price et Géry, 1968 from ***Barbus*** sp. (*occidentalis* aff.) (Gabon)

Dactylogyrus guirensis El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus pallaryi*** (Morocco)

Dactylogyrus helicophallus Paperna, 1973 from ***Labeo forskalii*** (Uganda), *L. victorianus*

Dactylogyrus heteromorphus El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus callensis*** (Tunisia)

Dactylogyrus insolitus Birgi et Lambert, 1987 from ***Enteromius martorelli*** (Cameroon)

Dactylogyrus iwani Crafford, Luus-Powell et Avenant-Oldewage, 2012 from ***Labeo capensis*** (South Africa), *L. umbratus*

Dactylogyrus jaculus Guégan, Lambert et Euzet, 1988 [syn. *Dactylogyrus* cf. *labeous* Paperna, 1979 (*partim*)] from ***Labeo coubie*** (Mali)

Dactylogyrus jaei Birgi et Lambert, 1987 from ***Enteromius jae*** (Cameroon)

Dactylogyrus jubbstrema Price, Korach et McPott, 1969 from ***Glossogobius giuris*** (South Africa)

Dactylogyrus jucundus Guégan et Lambert, 1991 from *Labeo parvus*, ***L. rouaneti*** (Guinea)

Dactylogyrus kii Birgi et Lambert, 1987 from ***Enteromius jae*** (Cameroon)

Dactylogyrus ksibii El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus callensis*** [syn. *Barbus ksibi*] (Morocco), *L. magniatlantis*, *L. setivimensis*

Dactylogyrus ksibioides El Gharbi, Birgi et Lambert, 1994 from *Carasobarbus moulouyensis*, ***Luciobarbus setivimensis*** (Morocco)

Dactylogyrus kulindrii El Gharbi, Birgi et Lambert, 1994 from *Carasobarbus fritschii*, *Labeobarbus reinii* (Morocco; type host not indicated)

Dactylogyrus labeous Paperna, 1969 from *Labeo coubie*, *L. senegalensis* (Ghana; type host not indicated)

Dactylogyrus larindae Crafford, Luus-Powell et Avenant-Oldewage, 2012 from *Labeo capensis*, ***L. umbratus*** (South Africa)

Dactylogyrus leonis Musilová, Řehulková et Gelnar, 2009 from ***Labeo coubie*** (Senegal)

Dactylogyrus longionchus Paperna, 1973 from ***Enteromius kerstenii*** (Uganda)

Dactylogyrus longiphalloides Guégan et Lambert, 1991 from ***Labeo alluaudi*** (Sierra Leone)

Dactylogyrus longiphallus Paperna, 1973 from *Enteromius kerstenii*, *Labeo forskalii*, *L. parvus*, ***L. victorianus*** (Kenya), *Labeobarbus altianalis*

Dactylogyrus macrocleithrum Price et Géry, 1968 from ***Barbus*** sp. (*holotaenia* aff.) (Gabon)

Dactylogyrus maillardi Birgi et Lambert, 1987 from ***Enteromius martorelli*** (Cameroon)

Dactylogyrus marocanus El Gharbi, Birgi et Lambert, 1994 from ***Carasobarbus fritschii*** [syn. *Barbus paytonii*] (Morocco), *C. harterti*, *Labeobarbus reinii*, *Luciobarbus callensis* [syn. *B. ksibi*], *L. nasus*, *L. setivimensis*

Dactylogyrus mawli Paperna, 1969 from ***Enteromius macrops*** (Ghana)

Dactylogyrus mendehei Birgi et Lambert, 1987 from ***Enteromius aspilus*** (Cameroon), *E. guirali*

Dactylogyrus myersi Price, McClellan, Druckenmiller et Jacobs, 1969 from *Enteromius perince*, ***E. trimaculatus*** (South Africa)

Dactylogyrus nanocirrus Paperna, 1973 from *Enteromius apleurogramma*, *E. neglectus*, *E. perince*, ***E. trispilos*** (Ghana), *E. sublineatus*

Dactylogyrus nathaliae Guégan, Lambert et Euzet, 1988 from ***Labeo*** sp. (Mali)

Dactylogyrus nicolettae Crafford, Luus-Powell et Avenant-Oldewage, 2012 from ***Labeo capensis*** (South Africa)

Dactylogyrus njinei Birgi et Lambert, 1987 from ***Enteromius camptacanthus*** (Cameroon)

Dactylogyrus nyongensis Birgi et Lambert, 1987 from *Enteromius aspilus*, *E. guirali* (Cameroon; type host not indicated)

Dactylogyrus oligospirophallus Paperna, 1973 [syn. *Dactylogyrus afrobarbae* Paperna, 1968 (*partim*)] from ***Labeo coubie*** (Ghana)

Dactylogyrus omega Guegan et Lambert, 1991 from *Labeo parvus*, ***L. rouaneti*** (Guinea)

Dactylogyrus oumiensis El Gharbi, Birgi et Lambert, 1994 from *Carasobarbus fritschii* [syn. *Barbus paytonii*], *C. harterti*, *Labeobarbus reinii* (Morocco; type host not indicated)

Dactylogyrus papernai Timofeeva, Gerasev et Gibson, 1996 [*D. magnum* Paperna, 1973 renamed] from ***Labeobarbus macrolepis*** (Tanzania)

Dactylogyrus parawaldronii Guégan et Lambert, 1990 from ***Labeobarbus parawaldroni*** (Guinea)

Dactylogyrus parviphallus Paperna, 1973 from *Enteromius apleurogramma*, *E. kerstenii* (Uganda; type host not indicated)

Dactylogyrus petitjeani Guégan et Lambert, 1990 from ***Labeobarbus petitjeani*** (Guinea)

Dactylogyrus pienaari Price, Korach et McPott, 1969 from ***Labeo rosae*** (South Africa)

Dactylogyrus pokoase Paperna, 1973 from ***Enteromius ablabes*** (Ghana)

Dactylogyrus pseudanchoratus Price et Géry, 1968 from ***Labeobarbus bynni*** [syns *Barbus occidentalis*, *B. waldroni*] (Gabon), *L. macrolepis*, *L. parawaldroni*, *L. petitjeani*, *L. sacratus*, *L. wurtzi*

Dactylogyrus rastellus Guégan, Lambert et Euzet, 1988 from ***Labeo senegalensis*** (Mali)

Dactylogyrus reinii El Gharbi, Birgi et Lambert, 1994 from ***Labeobarbus reinii*** (Morocco)

Dactylogyrus retroversus Guégan, Lambert et Euzet, 1988 from ***Labeo coubie*** (Mali)

Dactylogyrus ruahae Paperna, 1973 from ***Labeobarbus macrolepis*** (Tanzania), *L. parawaldroni*, *L. sacratus*, *L. wurtzi*

Dactylogyrus rufijii Paperna, 1973 from ***Labeobarbus macrolepis*** (Tanzania)

Dactylogyrus sacrati Guégan et Lambert, 1990 from ***Labeobarbus sacratus*** (Guinea)

Dactylogyrus sahelensis Guégan et Lambert, 1990 from ***Labeobarbus bynni*** [syns *Barbus occidentalis*, *B. waldroni*] (Mali), *L. petitjeani*

Dactylogyrus scorpius Rahmouni, Řehulková et Šimková, 2017 from ***Luciobarbus rifensis*** (Morocco)

Dactylogyrus sematus Guégan et Lambert, 1991 from ***Labeo rouaneti*** (Guinea)

Dactylogyrus senegalensis Paperna, 1969 from ***Labeo senegalensis*** (Ghana)

Dactylogyrus spinicirrus (Paperna et Thurston, 1968) [syn. *Neodactylogyrus spinicirrus* Paperna et Thurston, 1968] from *Enteromius nyanzae*, *E. radiatus*, *E. trimaculatus*, ***Labeobarbus altianalis*** (Uganda), *L. marequensis*, *L. somereni*

Dactylogyrus teresae Mashego, 1983 from ***Enteromius paludinosus*** (South Africa)

Dactylogyrus titus Guégan, Lambert et Euzet, 1988 from ***Labeo coubie*** (Mali)

Dactylogyrus tubarius Guégan, Lambert et Euzet, 1988 from ***Labeo senegalensis*** (Mali)

Dactylogyrus tunisiensis El Gharbi, Birgi et Lambert, 1994 from ***Luciobarbus callensis*** (Tunisia)

Dactylogyrus valeti Birgi et Lambert, 1987 from ***Enteromius camptacanthus*** (Cameroon)

Dactylogyrus varicorhini Bychowsky, 1958 from ***Labeobarbus kimberleyensis*** (South Africa)

Dactylogyrus varius Rahmouni, Řehulková et Šimková, 2017 from ***Luciobarbus maghrebensis*** (Morocco)

Dactylogyrus volutus El Gharbi, Birgi et Lambert, 1994 from ***Carasobarbus fritschii*** (Morocco)

Dactylogyrus wurtzii Guégan et Lambert, 1990 from ***Labeobarbus wurtzi*** (Guinea)

Dactylogyrus yassensis Musilová, Řehulková et Gelnar, 2009 from ***Labeo coubie*** (Senegal) [Fig. 4.4.9A]

Dactylogyrus zatensis El Gharbi, Birgi et Lambert, 1994 from ***Carasobarbus fritschii*** (Morocco)

Dogielius Bychowsky, 1936

Dogielius anthocolpos Guégan, Lambert et Euzet, 1989 from ***Labeo coubie*** (Mali) [Fig. 4.4.8B]

Dogielius clavipenis Guégan, Lambert et Euzet, 1989 from ***Labeo coubie*** (Mali)

Dogielius complicitus Guégan, Lambert et Euzet, 1989 from ***Labeo coubie*** (Mali)

Dogielius djolibaensis Guégan et Lambert, 1990 from ***Labeobarbus bynni*** [syns *Barbus occidentalis*, *B. waldroni*] (Mali), *L. petitjeani*

Dogielius dublicornis Paperna, 1973 from ***Labeo cylindricus*** (Tanzania)

Dogielius flagellatus Guégan, Lambert et Euzet, 1989 from ***Labeo coubie*** (Mali)

Dogielius flosculus Guégan, Lambert et Euzet, 1989 from ***Labeo senegalensis*** (Mali)

Dogielius grandijugus Guégan, Lambert et Euzet, 1989 from ***Labeo*** sp. (Mali)

Dogielius grandiphallus Paperna, 1973 from ***Labeobarbus macrolepis*** (Tanzania)

Dogielius harpagatus Guégan, Lambert et Euzet, 1989 from ***Labeo coubie*** (Mali)

Dogielius intorquens Crafford, Luus-Powell et Avenant-Oldewage, 2012 from *Enteromius paludinosus*, *Labeo capensis*, ***L. umbratus*** (South Africa)

Dogielius junorstrema Price et Yurkiewicz, 1968 from ***Labeo ruddi*** (Zimbabwe)

Dogielius kabaensis Guégan et Lambert, 1991 from *Labeo alluaudi*, ***L. parvus*** (Guinea)

Dogielius martorellii Birgi et Lambert, 1987 from ***Enteromius martorelli*** (Cameroon)

Dogielius njinei Birgi et Lambert, 1987 from ***Enteromius camptacanthus*** (Cameroon)

Dogielius parvus Guégan, Lambert et Euzet, 1989 from ***Labeo parvus*** (Mali)

Dogielius pedaloe Guégan et Lambert, 1990 from ***Labeobarbus parawaldroni*** (Guinea), *L. wurtzi*

Dogielius phrygieus Guégan et Lambert, 1990 from ***Labeobarbus sacratus*** (Guinea)

Dogielius rosumplicatus Guégan et Lambert, 1991 from *Labeo parvus*, ***L. rouaneti*** (Guinea)

Dogielius tropicus Paperna, 1969 from *Labeo coubie*, *L. senegalensis* (Ghana; type host not indicated)

Dogielius vexillus Guégan et Lambert, 1990 from ***Labeobarbus wurtzi*** (Ivory Coast)

Enterogyrus Paperna, 1963

Enterogyrus amieti Bilong Bilong, Euzet et Birgi, 1996 from ***Sarotherodon galilaeus*** (Cameroon) [Fig. 4.4.7A]

Enterogyrus barombiensis Bilong Bilong, Birgi et Euzet, 1991 from *Konia eisentrauti*, ***Pungu maclareni*** (Cameroon), *Stomatepia pindu*

Enterogyrus cichlidarum Paperna, 1963 [syn. *Enterogyrus niloticus* Eid et Negm, 1987] from *Coptodon nyongana*, ***C. zillii***, *Oreochromis mossambicus*, *O. niloticus*, *Pseudocrenilabrus philander*, *Sarotherodon galilaeus*

Enterogyrus coronatus Pariselle, Lambert et Euzet, 1991 from *Coptodon dageti*, ***C. guineensis*** (Ivory Coast), *Pseudocrenilabrus philander*

Enterogyrus crassus Bilong Bilong, Euzet et Birgi, 1996 from ***Coptodon nyongana*** (Cameroon)

Enterogyrus foratus Pariselle, Lambert et Euzet, 1991 from ***Sarotherodon melanotheron*** (Senegal or Ivory Coast; type locality not indicated)

Enterogyrus malmbergi Bilong Bilong, 1988 from ***Oreochromis niloticus*** (Cameroon)

Enterogyrus melenensis Bilong Bilong, Birgi et Lambert, 1989 from ***Hemichromis fasciatus*** (Cameroon)

Eutrianchoratus Paperna, 1969

Eutrianchoratus chibami Bilong Bilong, Euzet et Birgi, 1994 from ***Parachanna obscura*** (Cameroon)

Eutrianchoratus imbachi Dossou et Euzet, 1984 from ***Parachanna obscura*** (Benin)

Eutrianchoratus magnus Paperna, 1969 from ***Parachanna obscura*** (Ghana)

Eutrianchoratus malleus Bilong Bilong, Euzet et Birgi, 1994 from ***Parachanna obscura*** (Cameroon) [Fig. 4.4.10B]

Eutrianchoratus minutus Paperna, 1969 from ***Parachanna obscura*** (Ghana)

Heteronchocleidus Bychowsky, 1957

Heteronchocleidus adjanohouni Euzet et Dossou, 1975 from *Ctenopoma kingsleyae*, *C. petherici* (Benin; type host not indicated) [Fig. 4.4.10A]

Heteronchocleidus ctenopomae Paperna, 1969 from ***Ctenopoma kingsleyae*** (Ghana), *C. petherici*

Heteronchocleidus niloticus Paperna, 1973 from ***Ctenopoma muriei*** (Uganda)

Heteronchocleidus ouemensis Euzet et Dossou, 1975 from *Ctenopoma kingsleyae*, *C. petherici* (Benin; type host not indicated)

Heteronchocleidus tuzetae Euzet et Dossou, 1975 from *Ctenopoma kingsleyae*, *C. petherici* (Benin; type host not indicated)

Heterotesia Paperna, 1969

Heterotesia voltae Paperna, 1969 from ***Heterotis niloticus*** (Ghana) [Fig. 4.4.12B]

Insulacleidus Rakotofiringa et Euzet, 1983

Insulacleidus paratilapiae Rakotofiringa et Euzet, 1983 from ***Paratilapia polleni*** (Madagascar) [Fig. 4.4.19A]

Insulacleidus paretropli Rakotofiringa et Euzet, 1983 from ***Paretroplus polyactis*** (Madagascar)

Insulacleidus ptychochromidis Rakotofiringa et Euzet, 1983 from ***Ptychochromis oligacanthus*** (Madagascar)

Nanotrema Paperna, 1969

Nanotrema citharini Paperna, 1969 from ***Citharinus citharus*** (Ghana) [Fig. 4.4.8A]

Nanotrema niokoloensis Musilová, Řehulková et Gelnar, 2011 from ***Citharinus citharus*** (Senegal)

Onchobdella Paperna, 1968

Onchobdella aframae Paperna, 1968 from ***Hemichromis fasciatus*** (Ghana)

Onchobdella bopeleti Bilong Bilong et Euzet, 1995 from ***Hemichromis fasciatus*** (Cameroon or Mali; type locality not indicated)

Onchobdella krachii Paperna, 1968 from ***Chromidotilapia guntheri*** (Ghana)

Onchobdella melissa Pariselle et Euzet, 1995 from ***Pelmatochromis buettikoferi*** (Guinea)

Onchobdella pterigyalis Paperna, 1968 from ***Hemichromis bimaculatus*** (Ghana)

Onchobdella spirocirra Paperna, 1968 from ***Hemichromis bimaculatus*** (Ghana)

Onchobdella sylverai Pariselle et Euzet, 1995 from ***Pelmatochromis buettikoferi*** (Guinea)

Onchobdella voltensis Paperna, 1968 from *Hemichromis bimaculatus*, ***H. fasciatus*** (Ghana) [Fig. 4.4.11B]

Paraquadriacanthus Ergens, 1988 [syn. *Quadriacanthoides* Kritsky et Kulo, 1988]

Paraquadriacanthus nasalis Ergens, 1988 [syn. *Quadriacanthoides andersoni* Kritsky et Kulo, 1988] from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Egypt) [Fig. 4.4.14B]

Protoancylodiscoides Paperna, 1969

Protoancylodiscoides aurratum Bassock Bayiha, Nack et Pariselle, 2016 from ***Chrysichthys auratus*** (Cameroon)

Protoancylodiscoides chrysichthes Paperna, 1969 from *Chrysichthys auratus*, ***C. nigrodigitatus*** (Togo)

Protoancylodiscoides combesi Bassock Bayiha, Nack et Pariselle, 2016 from ***Chrysichthys auratus*** (Cameroon), *C. longidorsalis*, *C. nigrodigitatus*

Protoancylodiscoides katii N'Douba et Lambert, 1999 from ***Malapterurus electricus*** (Ivory Coast)

Protoancylodiscoides malapteruri Bilong Bilong, Birgi et Le Brun, 1997 from ***Malapterurus electricus*** (Cameroon)

Protoancylodiscoides mansourensis El-Naggar, 1987 from ***Chrysichthys auratus*** (Egypt) [Fig. 4.4.13]

Protoancylodiscoides sanagaensis Bassock Bayiha, Nack et Pariselle, 2017 from *Chrysichthys longidorsalis*, ***C. nigrodigitatus*** (Cameroon)

Protoancylodiscoides spirovagina Bassock Bayiha, Nack et Pariselle, 2017 from ***Chrysichthys nigrodigitatus*** (Cameroon)

Protoancylodiscoides valentini Bassock Bayiha, Nack et Pariselle, 2017 from *Chrysichthys longidorsalis*, ***C. nigrodigitatus*** (Cameroon)

Quadriacanthus Paperna, 1961 [syn. *Anacornuatus* Dubey, Gupta et Agarwal, 1992]

Quadriacanthus aegypticus El-Naggar et Serag, 1986 [syns *Anacornuatus aegypticus* (El-Naggar et Serag, 1986) Dubey, Gupta et Agarwal, 1992; *Quadriacanthus clariadis* Paperna, 1961 (*partim*)] from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Egypt)

Quadriacanthus agnebiensis N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus isopterus*** (Ivory Coast)

Quadriacanthus allobychowskiella Paperna, 1979 [syns *Quadriacanthus clariadis allobychowskiella* Paperna, 1979; *Quadriacanthus kearni* El-Naggar et Serag, 1985] from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Uganda)

Quadriacanthus anaspidoglanii Akoumba, Pariselle et Tombi, 2017 from ***Notoglanidium macrostoma*** (Cameroon)

Quadriacanthus ashuri Kritsky et Kulo, 1988 from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Egypt)

Quadriacanthus ayameensis N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus isopterus*** (Ivory Coast)

Quadriacanthus bagrae Paperna, 1979 [syn. *Quadriacanthus clariadis bagrae* Paperna, 1979] from *Bagrus bajad*, ***B. docmak*** (Uganda), *B. orientalis*, *Clarias gariepinus* [syn. *Clarias lazera*]

Quadriacanthus clariadis Paperna, 1961 [syns *Quadriacanthus clariadis clariadis* Paperna, 1979; *Quadriacanthus bagrae* Paperna, 1979 (*partim*)] from ***Clarias gariepinus*** [syn. *Clarias lazera*] [Fig. 4.4.12A]

Quadriacanthus dageti Birgi, 1988 from ***Clarias jaensis*** (Cameroon)

Quadriacanthus eboreus N'Douba et Lambert, 2001 from ***Clarias ebriensis*** (Ivory Coast)

Quadriacanthus euzeti Nack, Pariselle et Bilong Bilong, 2016 from ***Papyrocranus afer*** (Cameroon)

Quadriacanthus fornicatus Francová et Řehulková, 2017 from ***Clarias gariepinus*** (Sudan)

Quadriacanthus gourenei N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus isopterus*** (Ivory Coast)

Quadriacanthus ivoiriensis N'Douba et Lambert, 2001 from ***Clarias ebriensis*** (Ivory Coast)

Quadriacanthus levequei Birgi, 1988 from ***Clarias pachynema*** (Cameroon)

Quadriacanthus longifilisi N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus longifilis*** (Ivory Coast)

Quadriacanthus macrocirrus N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus isopterus*** (Ivory Coast)

Quadriacanthus macruncus Bahanak, Nack et Pariselle, 2016 from ***Clarias submarginatus*** (Cameroon)

Quadriacanthus mandibulatus Francová et Řehulková, 2017 from ***Heterobranchus bidorsalis*** (Sudan)

Quadriacanthus ndoubai Bahanak, Pariselle et Bilong Bilong, 2017 from ***Heterobranchus longifilis*** (Cameroon)

Quadriacanthus numidus Kritsky et Kulo, 1988 from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Egypt)

Quadriacanthus nyongensis Birgi, 1988 from *Clarias jaensis*, *C. pachynema* (Cameroon; type host not indicated)

Quadriacanthus ossaensis Bahanak, Nack et Pariselle, 2016 from ***Clarias submarginatus*** (Cameroon)

Quadriacanthus papernai Kritsky et Kulo, 1988 from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Egypt)

Quadriacanthus pravus Francová et Řehulková, 2017 from ***Clarias gariepinus*** (Sudan)

Quadriacanthus simplex N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus isopterus*** (Ivory Coast)

Quadriacanthus submarginati Bahanak, Nack et Pariselle, 2016 from ***Clarias submarginatus*** (Cameroon)

Quadriacanthus teugelsi Birgi, 1988 from *Clarias jaensis*, *C. pachynema* (Cameroon; type host not indicated)

Quadriacanthus thysi N'Douba, Lambert et Euzet, 1999 from ***Heterobranchus longifilis*** (Ivory Coast)

Quadriacanthus tilapiae Paperna, 1973 from ***Oreochromis esculentus*** (Uganda)

Quadriacanthus tricorniculai Bahanak, Pariselle et Bilong Bilong, 2017 from ***Heterobranchus longifilis*** (Cameroon)

Quadriacanthus triunguisi Bahanak, Pariselle et Bilong Bilong, 2017 from ***Heterobranchus longifilis*** (Cameroon)

Quadriacanthus voltaensis Paperna, 1965 from ***Clarias gariepinus*** [syn. *Clarias lazera*] (Ghana), *C. camerunensis* [syn. *C. walkeri*]

Quadriacanthus zuheiri Francová et Řehulková, 2017 from ***Clarias gariepinus*** (Sudan)

Schilbetrema Paperna et Thurston, 1968

Schilbetrema acornis Paperna et Thurston, 1968 from ***Schilbe intermedius*** [syn. *Schilbe mystus*] (Uganda)

Schilbetrema aegyptica El-Naggar, 1985 from ***Schilbe intermedius*** [syn. *Schilbe mystus*] (Egypt)

Schilbetrema biclavula N'Douba, Pariselle, Thys van den Audenaerde et Euzet, 1997 from ***Schilbe mandibularis*** (Ivory Coast)

Schilbetrema bicornis Paperna, 1969 – *species inquirenda* (Kritsky and Kulo 1992) from ***Parailia pellucida*** (Ghana)

Schilbetrema calamocleithrum Kritsky et Kulo, 1992 from ***Schilbe intermedius*** (Togo)

Schilbetrema dissimilis N'Douba, Pariselle, Thys van den Audenaerde et Euzet, 1997 from ***Schilbe mandibularis*** (Ivory Coast)

Schilbetrema eutropii Paperna, 1969 [syn. *Schilbetrema quadricornis eutropii* Paperna, 1969] from ***Schilbe mystus*** [syn. *Eutropius niloticus*] (Ghana)

Schilbetrema hexacornis Paperna, 1969 from ***Schilbe mystus*** [syn. *Eutropius niloticus*] (Ghana) [Fig. 4.4.17B]

Schilbetrema quadricornis Paperna et Thurston, 1968 [syn. *Schilbetrema quadricornis schilbae* Paperna, 1969] from ***Schilbe intermedius*** [syn. *Schilbe mystus*] (Uganda)

Schilbetrema spirocirra Paperna, 1969 from ***Schilbe mystus*** [syn. *Eutropius niloticus*] (Ghana)

Schilbetrema torula Kritsky et Kulo, 1992 from ***Schilbe mystus*** (Togo)

Schilbetrema tricera Paperna, 1973 from ***Schilbe*** sp. (Tanzania)

Schilbetrema undinula Kritsky et Kulo, 1992 from ***Schilbe intermedius*** (Togo)

Schilbetrema vacillans Kritsky et Kulo, 1992 from ***Schilbe intermedius*** (Togo)

Schilbetrematoides Kritsky et Kulo, 1992

Schilbetrematoides manizani N'Douba, Lambert, Pariselle et Euzet, 2000 from *Schilbe intermedius* (new host record), ***S. mandibularis*** (Ivory Coast) [Fig. 4.4.9B]

Schilbetrematoides pseudodactylogyrus Kritsky et Kulo, 1992 from ***Schilbe intermedius*** (Togo)

Scutogyrus Pariselle et Euzet, 1995

Scutogyrus bailloni Pariselle et Euzet, 1995 from ***Sarotherodon galilaeus*** (Niger)

Scutogyrus chikhii Pariselle et Euzet, 1995 from ***Oreochromis mossambicus*** (Republic of the Congo)

Scutogyrus ecoutini Pariselle et Euzet, 1995 from ***Sarotherodon occidentalis*** (Guinea)

Scutogyrus gravivaginus (Paperna et Thurston, 1969) [syns *Cichlidogyrus longicornis gravivaginus* Paperna et Thurston, 1969; *Cichlidogyrus gravivaginus* Paperna et

Thurston, 1969 of Douëllou (1993)] from ***Oreochromis leucostictus*** (Uganda), *O. mortimeri*, *O. mweruensis*, *O. tanganicae*, *O. variabilis*

Scutogyrus longicornis (Paperna et Thurston, 1969) [syns *Cichlidogyrus longicornis longicornis* Paperna et Thurston, 1969; *Cichlidogyrus longicornis* of Douëllou (1993); *Actinocleidus muelleri* Ferdousi et Chandra, 2002] from *Coptodon zillii*, *Oreochromis aureus*, *O. mortimeri*, *O. mossambicus*, *O. niloticus*, ***Sarotherodon galilaeus*** (Ghana)

Scutogyrus minus (Dossou, 1982) [syns *Cichlidogyrus longicornis minus* Dossou, 1982; *Cichlidogyrus minus* of Pariselle & Euzet, 1995] from *Oreochromis niloticus,* ***Sarotherodon melanotheron*** (Benin) [Fig. 4.4.15A]

Scutogyrus vanhovei Pariselle, Bitja Nyom et Bilong Bilong, 2013 from ***Pelmatolapia mariae*** (Cameroon)

Synodontella Dossou et Euzet, 1993

Synodontella apertipenis Mbondo, Nack et Pariselle, 2017 from ***Synodontis rebeli*** (Cameroon)

Synodontella arcopenis Dossou et Euzet, 1993 from ***Synodontis sorex*** (Benin or Mali; type locality not indicated)

Synodontella davidi Dossou et Euzet, 1993 from ***Synodontis membranaceus*** (Mali)

Synodontella melanoptera Dossou et Euzet, 1993 from ***Synodontis melanopterus*** (Benin), *S. obesus*, *S. rebeli* [Fig. 4.4.18A]

Synodontella sanagaensis Mbondo, Nack et Pariselle, 2017 from ***Synodontis rebeli*** (Cameroon)

Synodontella synodontii (Paperna et Thurston, 1968) [syns *Ancyrocephalus synodontii* Paperna et Thurston, 1968; *Schilbetrema synodontii* (Paperna et Thurston, 1968)] from *Synodontis membranaceus*, ***S. victoriae*** (Uganda), *S. zambezensis*

Synodontella zambezensis Douëllou et Chishawa, 1995 from ***Synodontis zambezensis*** (Zimbabwe)

Urogyrus Bilong Bilong, Birgi et Euzet, 1994

Urogyrus cichlidarum Bilong Bilong, Birgi et Euzet, 1994 from ***Benitochromis batesii*** (Cameroon), *Parananochromis caudifasciatus*, *Pungu maclareni*, *Stomatepia pindu*, *Tilapia* sp. [Fig. 4.4.7B]

DIPLECTANIDAE Monticelli, 1903

Diplectanum Diesing, 1858

Diplectanum lacustre Thurston et Paperna, 1969 – *incertae sedis* (Domingues and Boeger 2008) from ***Lates niloticus*** [syn. *Lates albertianus*] (Ghana) [Fig. 4.4.6B]

GYRODACTYLIDAE van Beneden et Hesse, 1863

Afrogyrodactylus Paperna, 1968

Afrogyrodactylus ardae Přikrylová, Smit et Gelnar 2017 from ***Rhabdalestes septentrionalis*** (Senegal)

Afrogyrodactylus characinis Paperna, 1968 from ***Micralestes*** sp. (Ghana)

Afrogyrodactylus girgifae Přikrylová et Luus-Powell, 2016 from ***Brycinus nurse*** (Sudan)

Afrogyrodactylus kingi Přikrylová et Luus-Powell, 2014 from ***Micralestes acutidens*** (South Africa) [Fig. 4.4.21B]

Citharodactylus Přikrylová, Shinn et Paladini, 2017

Citharodactylus gagei Přikrylová, Shinn et Paladini, 2017 from ***Citharinus citharus*** (Kenya) [Fig. 4.4.22B]

Diplogyrodactylus Přikrylová, Matějusová, Musilová, Gelnar et Harris, 2009

Diplogyrodactylus martini Přikrylová, Matějusová, Musilová, Gelnar et Harris, 2009 from ***Polypterus senegalus*** (Senegal) [Fig. 4.4.20A]

Gyrodactylus von Nordmann, 1832 [Fig. 4.4.22A]

Gyrodactylus alberti Paperna, 1973 from ***Clarias gariepinus*** (Uganda)

Gyrodactylus alekosi Přikrylová, Blažek et Vanhove, 2012 from ***Clarias gariepinus*** (Mozambique)

Gyrodactylus amphiliusi Paperna, 1973 from ***Amphilius atesuensis*** (Ghana)

Gyrodactylus anabanti Paperna, 1973 from ***Ctenopoma muriei*** (Uganda)

Gyrodactylus camerunensis Nack, Bilong Bilong et Euzet, 2005 from ***Clarias camerunensis*** (Cameroon)

Gyrodactylus chitandiri Zahradníčková, Barson, Luus-Powell et Přikrylová, 2016 from ***Coptodon rendalli*** (Zimbabwe), *Pseudocrenilabrus philander*

Gyrodactylus cichlidarum Paperna, 1968 from *Coptodon guineensis*, *C. zilii*, *Hemichromis bimaculatus*, *H. fasciatus*, ***Sarotherodon galilaeus*** (Ghana), *S. melanotheron*

Gyrodactylus clarii Paperna, 1973 from ***Clarias gariepinus*** (Uganda)

Gyrodactylus ctenopomi Paperna, 1973 from ***Ctenopoma muriei*** (Uganda)

Gyrodactylus cyprinodonti Paperna, 1968 from ***Epiplatys*** sp. (Ghana)

Gyrodactylus cytophagus Paperna, 1968 from ***Poropanchax normani*** (Ghana)

Gyrodactylus ergensi Přikrylová, Matějusová, Musilová et Gelnar, 2012 from ***Sarotherodon galilaeus*** (Senegal), *Oreochromis niloticus*

Gyrodactylus gelnari Přikrylová, Blažek et Vanhove, 2012 from ***Clarias anguillaris*** (Senegal), *C. gariepinus*

Gyrodactylus groschafti Ergens, 1973 from ***Clarias gariepinus*** (Egypt)

Gyrodactylus haplochromi Paperna, 1973 from ***Haplochromis elegans*** (Uganda)

Gyrodactylus hildae García-Vásquez, Hansen, Christison, Bronn et Shinn, 2011 from ***Oreochromis niloticus*** (Ethiopia)

Gyrodactylus ivindoensis Price et Géry, 1968 from ***Enteromius holotaenia*** (Gabon)

Gyrodactylus kyogae Paperna, 1973 from ***Enteromius perince*** (Uganda)

Gyrodactylus malalai Přikrylová, Blažek et Gelnar, 2012 from *Coptodon zillii*, ***Oreochromis niloticus*** (Kenya)

Gyrodactylus micralestes Paperna, 1968 from ***Micralestes*** sp. (Ghana)

Gyrodactylus nigritae Přikrylová, Blažek et Vanhove, 2012 from ***Synodontis nigrita*** (Senegal)

Gyrodactylus nyanzae Paperna, 1973 from *Coptodon rendalli*, *Oreochromis mweruensis*, *O. niloticus*, *O. niloticus* x *mweruensis*, ***O. variabilis*** (Uganda)

Gyrodactylus nyongensis Nack, Bilong Bilong et Euzet, 2005 from ***Clarias camerunensis*** (Cameroon)

Gyrodactylus occupatus Zahradníčková, Barson, Luus-Powell et Přikrylová, 2016 from ***Oreochromis niloticus*** (Zimbabwe), *Pharyngochromis acuticeps*, *Pseudocrenilabrus philander*, *Tilapia* sp.

Gyrodactylus parisellei Zahradníčková, Barson, Luus-Powell et Přikrylová, 2016 from *Oreochromis niloticus*, ***Pseudocrenilabrus philander*** (Zimbabwe), *Tilapia* sp.

Gyrodactylus rysavyi Ergens, 1973 from *Clarias anguillaris*, ***C. gariepinus*** (Egypt)

Gyrodactylus sturmbaueri Vanhove, Snoeks, Volckaert et Huyse, 2011 from *Pseudocrenilabrus philander*, ***Simochromis diagramma*** (Zambia)

Gyrodactylus synodonti Přikrylová, Blažek et Vanhove, 2012 from ***Synodontis nigrita*** (Senegal)

Gyrodactylus thlapi Christison, Shinn et Van As, 2005 from ***Pseudocrenilabrus philander*** (Botswana)

Gyrodactylus thysi Vanhove, Snoeks, Volckaert et Huyse, 2011 from ***Simochromis diagramma*** (Zambia)

Gyrodactylus tranvaalensis Prudhoe et Hussey, 1977 from *Clarias anguillaris*, ***C. gariepinus*** (South Africa)

Gyrodactylus turkanaensis Přikrylová, Blažek et Vanhove, 2012 from ***Clarias gariepinus*** (Kenya)

Gyrodactylus ulinganisus García-Vásquez, Hansen, Christison, Bronn et Shinn, 2011 from ***Oreochromis mossambicus*** (South Africa)

Gyrodactylus yacatli García-Vásquez, Hansen, Christison, Bronn et Shinn, 2011 from ***Oreochromis niloticus***, *Pseudocrenilabrus philander*

Gyrodactylus zimbae Vanhove, Snoeks, Volckaert et Huyse, 2011 from *Ctenochromis horei*, ***Simochromis diagramma*** (Zambia)

Macrogyrodactylus Malmberg, 1957

Macrogyrodactylus anabanti Paperna, 1973 from ***Ctenopoma muriei*** (Uganda)

Macrogyrodactylus clarii Gussev, 1961 from *Clarias gariepinus*, ***Clarias*** sp. (Ethiopia)

Macrogyrodactylus congolensis (Prudhoe, 1957) [syn. *Neogyrodactylus congolensis* Prudhoe, 1957] from *Clarias anguillaris*, ***C. gariepinus*** (Democratic Republic of the Congo) [Fig. 4.4.20B]

Macrogyrodactylus ctenopomi Paperna, 1973 from ***Ctenopoma muriei*** (Uganda)

Macrogyrodactylus heterobranchii N'Douba et Lambert, 1999 from *Clarias anguillaris*, ***Heterobranchus longifilis*** (Ivory Cost)

Macrogyrodactylus karibae Douëllou et Chishawa, 1995 from ***Clarias gariepinus*** (Zimbabwe)

Macrogyrodactylus latesi Paperna, 1969 from ***Lates niloticus*** (Ghana)

Macrogyrodactylus polypteri Malmberg, 1957 from ***Polypterus senegalus*** (Gambia)

Macrogyrodactylus simetiensis Přikrylová et Gelnar, 2008 from ***Polypterus senegalus*** (Senegal)

Mormyrogyrodactylus Luus-Powell, Mashego et Khalil, 2003

Mormyrogyrodactylus gemini Luus-Powell, Mashego et Khalil, 2003 from ***Marcusenius macrolepidotus*** (South Africa) [Fig. 4.4.21A]

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Chapter 4.5.

TREMATODA

Olena KUDLAI, Tomáš SCHOLZ & Nico SMIT

Flukes (Trematoda) – basic characteristics, life cycles, classification and principal diagnostic features

- parasitic flatworms (Platyhelminthes: Neodermata)
- almost 20,000 species classified in 2 subclasses
- subclass Digenea with 2 orders, 25 superfamilies and 148 families
- obligate parasites, almost exclusively endoparasites
- in the digestive system and other organs
- in all groups of vertebrates, with the highest number of species in bony fishes; exceptionally, adults in invertebrates
- body dorsoventrally flattened in most species
- usually with two muscular suckers: the oral sucker at the anterior end of the body and the ventral one (acetabulum) in the mid-region (close to the posterior extremity of the body in Paramphistomatoidea)
- digestive tract well-developed
- body surface covered with tegument (neodermis), sometimes with spines
- all African species in fishes hermaphroditic
- except for some aspidogastreans, life cycles indirect (1-3 intermediate hosts)
- first intermediate hosts molluscs, especially gastropods
- wide spectrum of second intermediate hosts, *e.g.*, molluscs, insects, oligochaetes, fishes
- some species, especially larvae (*e.g.*, metacercariae of the Diplostomidae and Clinostomidae), can be pathogenic for fish hosts
- causative agents of human fish-borne diseases (small liver and intestinal flukes) – not common in Africa

The life cycle of trematodes is complex with both free-living and parasitic stages and involves several hosts (Fig. 4.5.1 A, B). Adults produce eggs that pass in the faeces of the host to the environment where they hatch to release a free-living larva, the miracidium. The miracidium penetrates a molluscan first intermediate host (some marine species infect annelids). After penetration the miracidium develops into a mother sporocyst, the first intramolluscan generation. It multiplies parthenogenetically, producing the second intramolluscan generation – multiple daughter sporocysts or multiple rediae. Daughter sporocysts and rediae reproduce parthenogenetically generating the larvae of the sexual adult – cercariae. There are several ways for cercariae to infect the invertebrate or vertebrate second intermediate host, which host metacercariae. Cercariae may actively penetrate

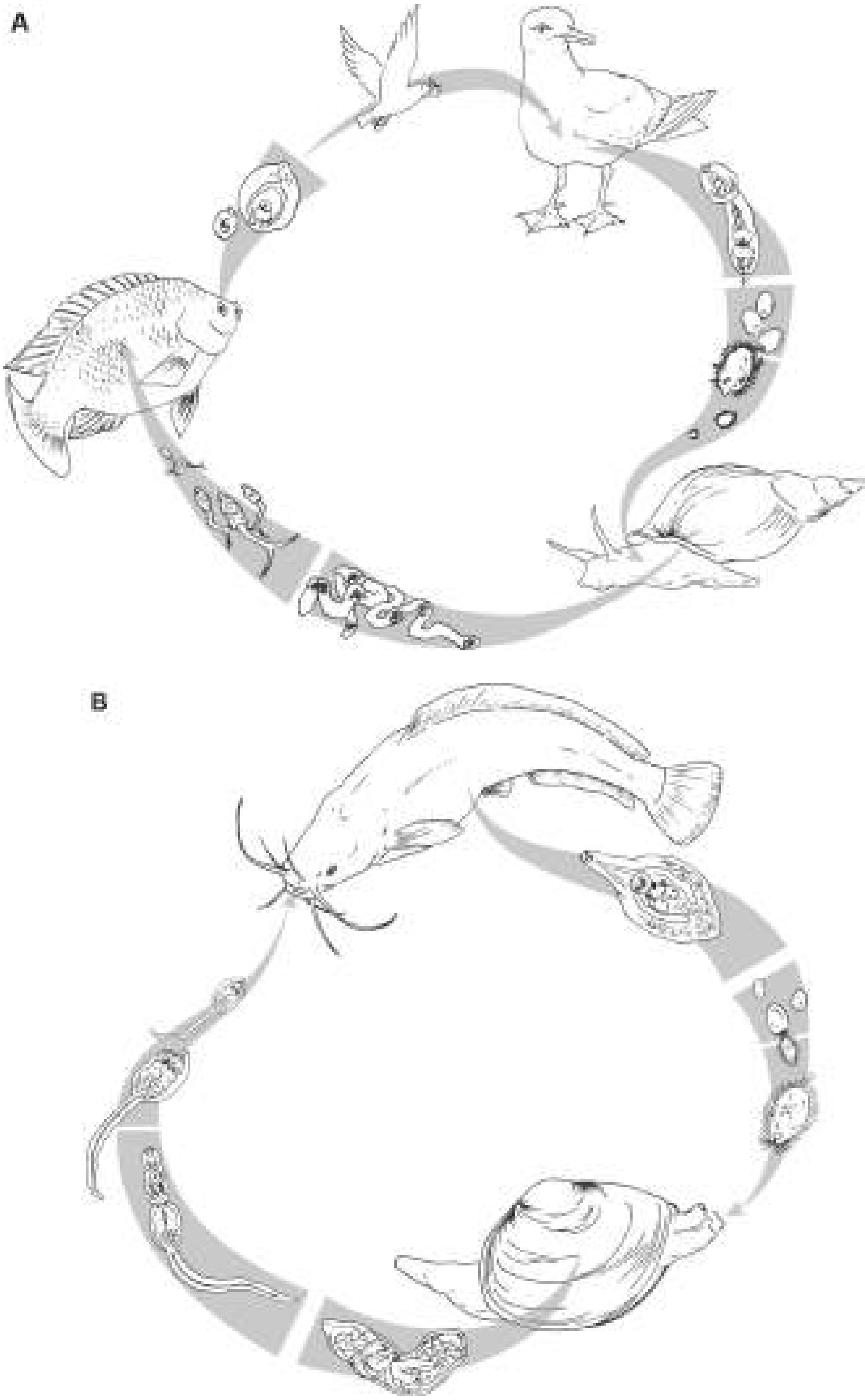

Fig. 4.5.1. Life cycles of trematodes. **A.** *Diplostomum* sp. (fish serves as the second intermediate host); **B.** *Phyllodistomum* sp. (fish serves as the definitive host). (Illustrations by M. Luo.)

through the skin or be eaten by the host. The vertebrate definitive hosts are infected when they consume the second intermediate host.

Trematoda includes two subclasses, Aspidogastrea and Digenea. The former subclass includes only 61 species (Alves *et al.* 2015) compared with almost 20,000 nominal species of digeneans. The fundamental unit of the **classification** of the Digenea is the superfamily. A total of 25 superfamilies with 148 families have been recognised (see Bray *et al.* 2008). As many as 60 families of digeneans include parasites of fishes, and 38 families are exclusively fish parasites. Relatively few of these families possess unique morphological traits that would differentiate them easily from each other. Therefore, individual families are usually characterised by a unique combination of non-unique morphological characteristics. Life cycles and cercarial morphology also play an important role in family definition. As a result, it is very difficult or almost impossible to present simple keys to the families of fish trematodes in Africa.

Seven basic morphotypes of digeneans are recognised based on their general body plan, especially the number and position of suckers, but most trematodes parasitic in African freshwater fishes belong to the distomatous morphological type, which has two suckers, the oral sucker at the anterior end of the body and the ventral one in the mid-region of the body.

Identification to the genus and species level is based on a high number of morphological characteristics, many of them related to the genital organs, such as relative position of the gonads, their shape and extent, the position of the genital pores, structure of the cirrus-sac, the size of the eggs, etc. (see Gibson *et al.* 2002; Jones *et al.* 2005; Bray *et al.* 2008 for keys to trematode genera).

Key to the genera of the Trematoda (adults) of freshwater fishes in Africa

1 (2) Ventral surface of the body with a large muscular, pad-like Baer's disc with numerous alveoli………………***Aspidogaster*** (**subclass Aspidogastrea**) [Fig. 4.5.2A]

2 (1) Ventral surface of the body without the Baer's disc….(**subclass Digenea**)

3 (4) Parasitic in the alimentary canal and associated cavities and organs ……7

4 (3) Not parasitic in the alimentary canal and associated cavities and organs………………………………………………………………………… 5

5 (6) Adults in the blood system……***Sanguinicola*** (Aporocotylidae) [Fig. 4.5.2D]

6 (5) Adults in tissues of the body……………………………………………………………………………………..***Nematobothrium*** (Didymozoidae) [Fig. 4.5.4H]

7 (8) Parasitic in gall or urinary bladders……………………………………………9

8 (7) Parasitic in other organs……………………………………………………13

9 (10) Parasitic in gall-bladder……………………………………………………11

10 (9) Parasitic in urinary bladder……………***Phyllodistomum*** (Gorgoderidae) [Fig. 4.5.4E]

11 (12) Body pyriform; testes symmetrical, extracaecal, *i.e.*, external to the caeca, at level of the ventral sucker; ovary between testes…………………………………***Callodistomum*** (Callodistomidae) [Fig. 4.5.2E]

12 (11) Body cylindrical; testes oblique, overlying caeca, posterior to the ventral sucker; ovary pretesticular, lateral, on the same side as the posterior testis……………………………***Cholepotes*** (Callodistomidae) [Fig. 4.5.2F]

13 (14) Circumoral crown of spines present………………………………………15

14 (13) Circumoral crown of spines absent………………………………………17

15 (16) Intestinal bifurcation just anterior to the ventral sucker; caeca open externally by separate ani; cirrus-sac absent…………………***Acanthostomum*** (Cryptogonimidae) [Fig. 4.5.3F]

16 (15) Intestinal bifurcation close to pharynx; caeca blind; cirrus-sac present …………………………………..***Masenia*** (Cephalogonimidae) [Fig. 4.5.3A]

17 (18) Forebody expanded laterally…………………***Deropristis*** (Deropristidae) [Fig. 4.5.4D]

18 (17) Forebody not expanded laterally…………………………………………19

19 (20) Ventral sucker at the posterior extremity……………………………………21

20 (19) Ventral sucker ventral……………………………………………………27

21 (22) Testes caecal or extracaecal………………………………………………23

22 (21) Testes intercaecal, in middle third of body close to caecal arch, may overlie caeca slightly……………***Panamphistomum*** (Cladorchiidae) [Fig. 4.5.3D]

23 (24) Testes diagonal or tandem; anterior testis caecal or extracaecal, posterior testis intercaecal……………………………………………………………25

24 (23) Testes symmetrical, overlap caeca at or just behind level of intestinal bifurcation……………………………***Brevicaecum*** (Cladorchiidae) [Fig. 4.5.3C]

25 (26) Body conical; acetabulum massive, wider than the body, with very large, prominent papillae, but without powerful horseshoe-shaped sphincter …………………………………***Basidiodiscus*** (Cladorchiidae) [Fig. 4.5.3B]

26 (25) Body elongate oval; acetabulum not massive, without very large papillae, but with a powerful horseshoe-shaped sphincter……………………………***Sandonia*** (Cladorchiidae) [Fig. 4.5.3E]

27 (28) One testis only……………………………………………………………29

28 (27) Two or more testes………………………………………………………33

29 (30) Cirrus-sac absent……………***Haplorchoides*** (Heterophyidae) [Fig. 4.5.5B]

30 (29) Cirrus-sac present……………………………………………………….31

31 (32) Body elongate; prepharynx indistinct; caeca cylindrical, long, often terminating near the posterior end of the body; testis entirely intracaecal; vitellarium at mid-body; uterus occupying much of hindbody, often extending into forebody…………………..***Paralecithobotrys*** (Haploporidae) [Fig. 4.5.4F]

32 (31) Body fusiform; prepharynx somewhat longer or shorter than pharynx; caeca sac-like, relatively short; testis entirely postcaecal; vitellarium in hind-body; uterus restricted to the hind-body………………***Saccocoelium*** (Haploporidae) [Fig. 4.5.4G]

33 (34) Testes two……………………………………………………………….35

34 (33) Testes nine…………………….***Siphodera*** (Cryptogonimidae) [Fig. 4.5.3H]

35 (36) Caeca terminate blindly…………………………………………………39

36 (35) Caeca unite to form cyclocoel………………………………………….37

37 (38) Cirrus-sac present; testes symmetrical …………………***Trematobrien*** (Apocreadiidae) [Fig. 4.5.2C]

38 (37) Cirrus-sac absent; testes tandem to oblique……………………***Nicolla*** (Opecoelidae) [Fig. 4.5.5E]

39 (40) Cirrus-sac present……………………………………………………….53

40 (39) Cirrus-sac absent………………………………………………………..41

41 (42) Sinus-sac present………………………………………………………..43

42 (41) Sinus-sac absent………………………………………………………...45

43 (44) Pars prostatica long, may be sparsely surrounded by gland cells; seminal vesicle trilocular…………………………***Dinurus*** (Hemiuridae) [Fig. 4.5.6A]

44 (43) Pars prostatica short, connected to the seminal vesicle by the long glandular duct; seminal vesicle variable, tubular, saccular or divided into two or three sections………………………..***Ectenurus*** (Hemiuridae) [Fig. 4.5.5A]

45 (46) Vitellarium two masses at the posterior extremity of the body ……………47

46 (45) Vitellarium follicular, form two lateral bands in hind-body…………………49

47 (48) Testes anterior to the ovary and vitellarium ……………………***Halipegus*** (Derogenidae) [Fig. 4.5.4B]

48 (47) Testes posterior to the ovary and vitellarium …………………***Gonocerca*** (Gonocercidae) [Fig. 4.5.4C]

49 (50) The vitelline follicles extend posteriorly to level of the ovary or anterior testis……………………………***Thaparotrema*** (Opisthorchiidae) [Fig. 4.5.5G]

50 (49) The vitelline follicles extend posteriorly to level of posterior testis……….51

51 (52) Body oval; oral sucker without enlarged oral spines, opens subterminally…………………………***Neocladocystis*** (Cryptogonimidae) [Fig. 4.5.4A]

52 (51) Body fusiform; oral sucker with enlarged oral spines, opens terminally ………………………………***Brientrema*** (Cryptogonimidae) [Fig. 4.5.3G]

53 (54) Vitellarium reaching posterior extremity…………………………………….55

54 (53) Vitellarium not reaching posteriorly extremity……………………………59

55 (56) Uterus extending to, or near to, posterior extremity……***Orientocreadium*** (Orientocreadiidae) [Fig. 4.5.5H]

56 (55) Uterus not extending to, or near to, posterior extremity…………………. 57

57 (58) Intestinal bifurcation at the posterior margin of the ventral sucker ……………………………….***Allocreadium*** (Allocreadiidae) [Fig. 4.5.2B]

58 (57) Intestinal bifurcation anterior to the ventral sucker…………***Plagioporus*** (Opecoelidae) [Fig. 4.5.5F]

59 (60) Testes symmetrical…………***Malawitrema*** (Macroderoididae) [Fig. 4.5.5D]

60 (59) Testes oblique……………………………………………………………61

61 (62) Genital pore extracaecal……………………………………………….63

62 (61) Genital pore intercaecal………………………………………………65

63 (64) Genital pore submedian at level of oral sucker……………***Emoleptalea*** (Cephalogonimidae) [Fig. 4.5.2G]

64 (63) Genital pore submarginal, sinistral, at the level of pharynx…***Heterorchis*** [*incertae sedis* in the superfamily Plagiorchioidea (*sensu lato*)] [Fig. 4.5.5C]

65 (66) Seminal vesicle bipartite………………………………………***Glossidium*** [*incertae sedis* in the superfamily Plagiorchioidea (*sensu lato*)] [Fig. 4.5.5J]

66 (65) Siminal vesicle unipartite…………………………………….***Astiotrema*** [*incertae sedis* in the superfamily Plagiorchioidea (*sensu lato*)] [Fig. 4.5.5I]

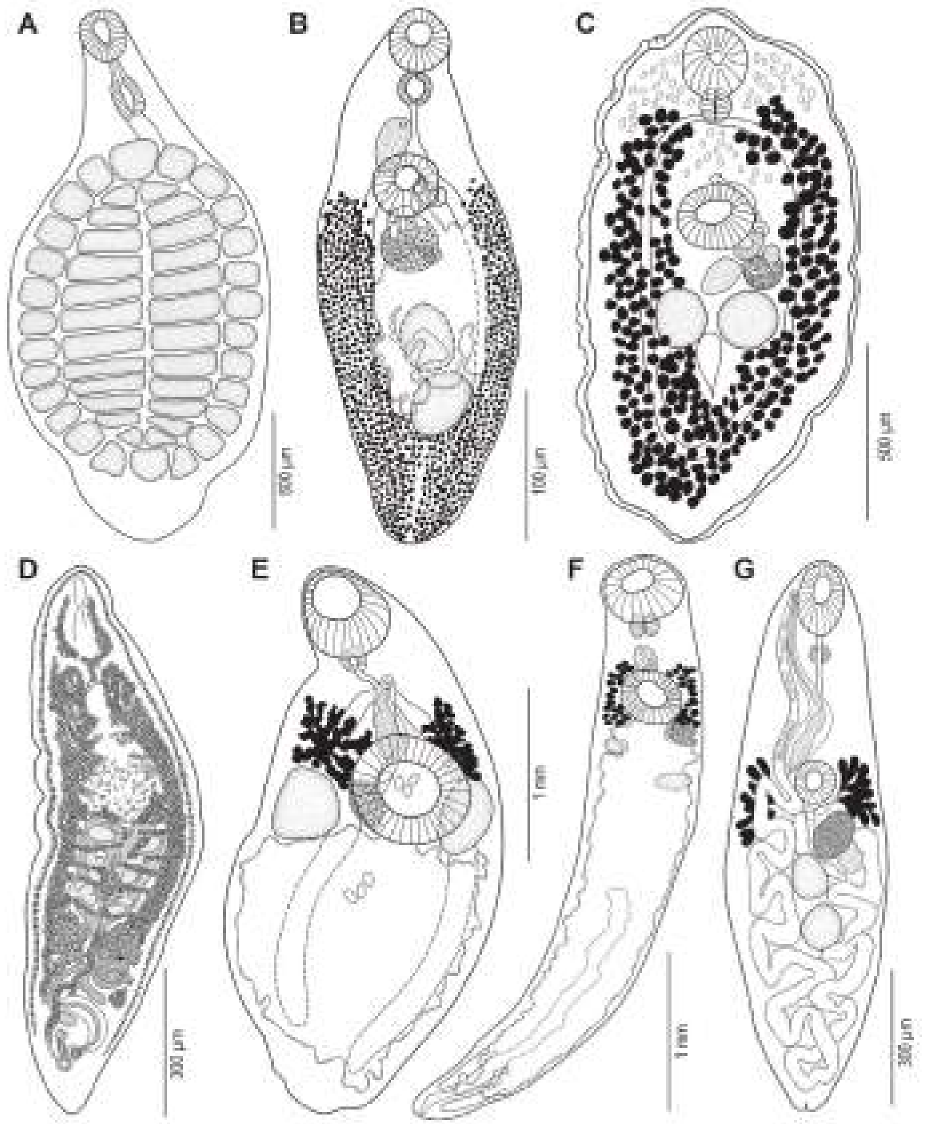

Fig. 4.5.2. Trematoda. **A.** *Aspidogaster africanus* Saoud, Mohamed et Abdel-Hamid, 1974 from *Labeobarbus bynni*; **B.** *Allocreadium mazoensis* Beverly-Burton, 1962 from *Clarias gariepinus*; **C.** *Trematobrien haplochromios* Dollfus, 1950 from *Pseudocrenilabrus philander*; **D.** *Sanguinicola chalmersi* Odhner, 1924 from *Auchenoglanis occidentalis*; **E.** *Callodistomum diaphanum* Odhner, 1902 from *Ctenopoma kingsleyae*; **F.** *Cholepotes ovofarctus* (Odhner, 1902) from *Synodontis schall*; **G.** *Emoleptalea exilis* (Looss, 1899) from *Bagrus bajad*. (Modified from Looss 1899; Odhner 1924; Beverly-Burton 1962; Manter 1962; Saoud *et al.* 1974; Jones 1982; Bray 2002.)

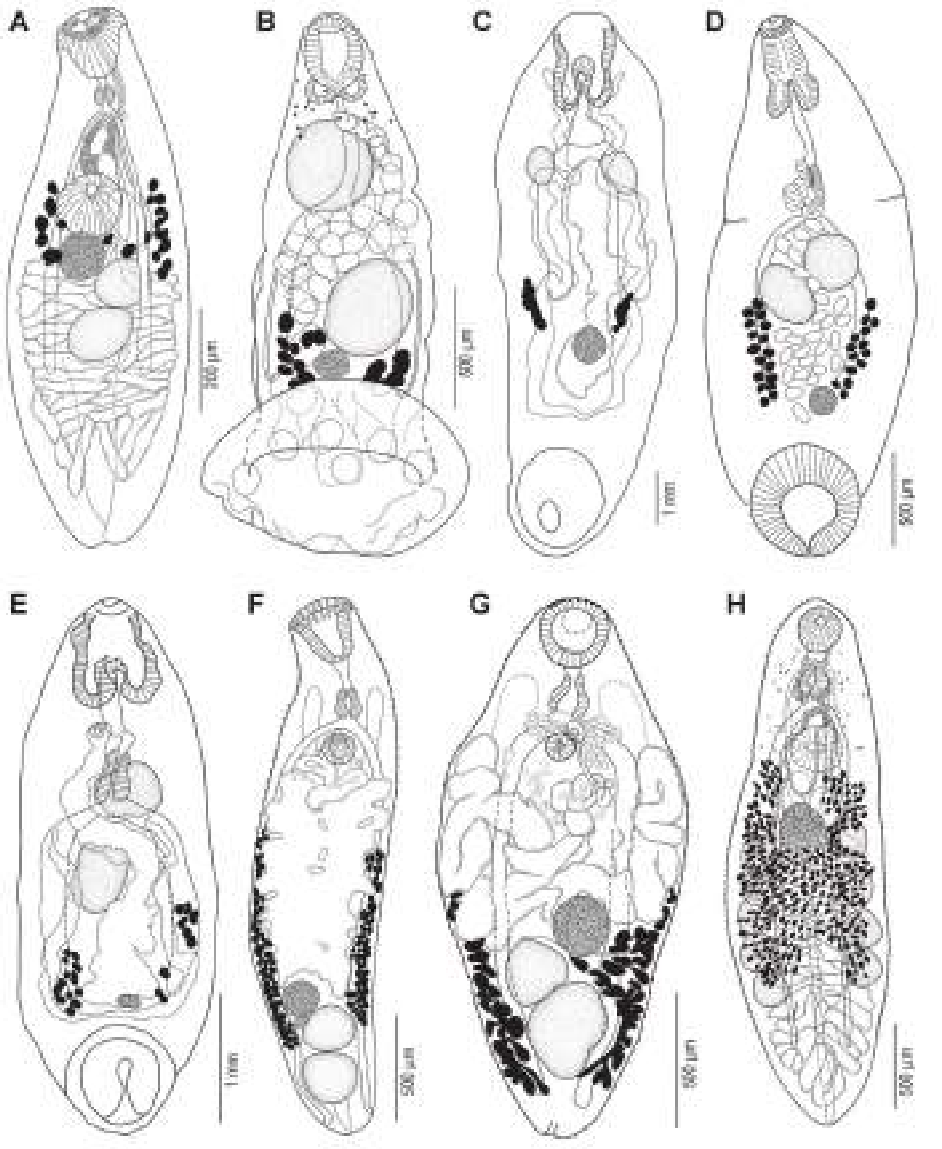

Fig. 4.5.3. Trematoda. **A.** *Masenia ghanensis* (Fischthal et Thomas, 1968) from *Heterobranchus longifilis*; **B.** *Basidiodiscus ectorchis* Fischthal et Kuntz, 1959 from *Synodontis schall*; **C.** *Brevicaecum niloticum* McClelland, 1957 from *Citharinus citharus*; **D.** *Panamphistomum benoiti* Manter et Pritchard, 1964 from *Pseudocrenilabrus philander*; **E.** *Sandonia sudanensis* McClelland, 1957 from *Distichodus nefasch* or *S. schall* (host not specified); **F.** *Acanthostomum absconditum* (Looss, 1901) from *Bagrus bajad*; **G.** *Brientrema malapteruri* Dollfus, 1950 from *Malapterurus electricus*; **H.** *Siphodera ghanensis* Fischthal et Thomas, 1968 from *Chrysichthys nigrodigitatus*. (Modified from McClelland 1957; Manter & Pritchard 1964; Fischthal & Thomas 1968a, b.; Moravec 1976; B and G – holotypes illustrated by T. Scholz).

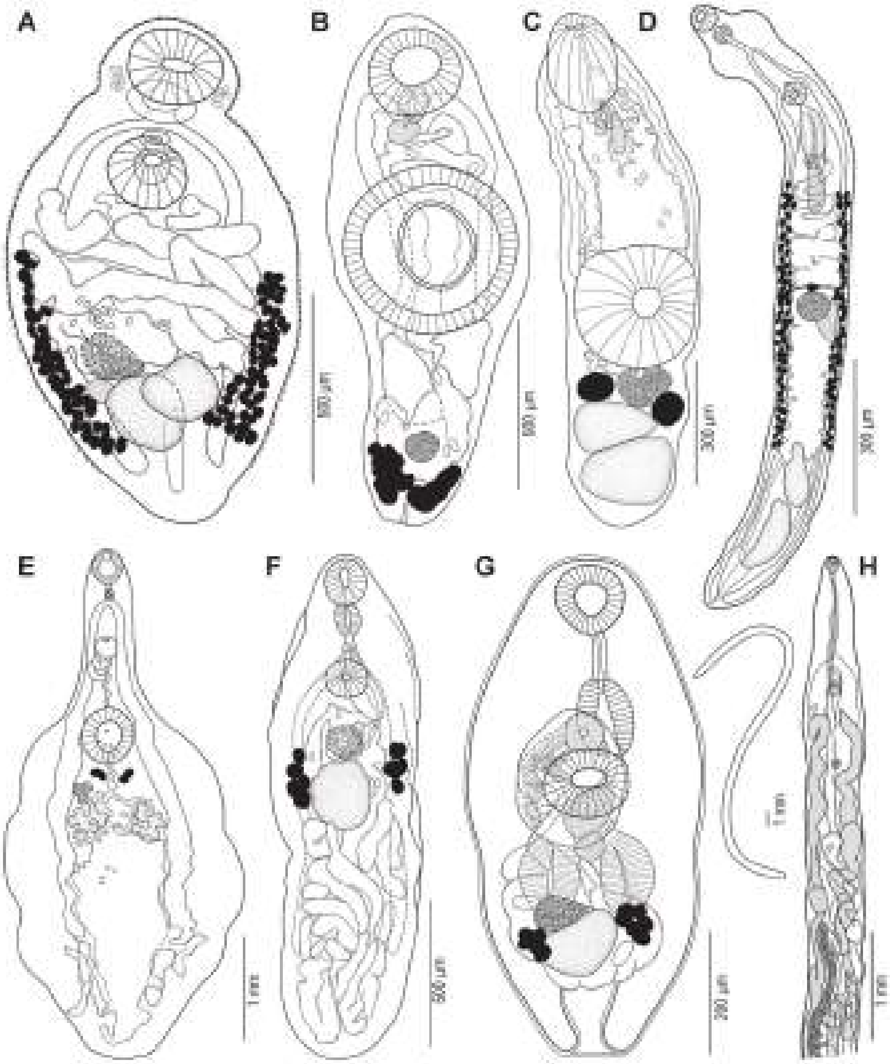

Fig. 4.5.4. Trematoda. **A.** *Neocladocystis congoensis* Manter et Pritchard, 1969 from *Parauchenoglanis monkei*; **B.** *Halipegus ctenopomi* Jones, 1982 from *Ctenopoma kingsleyae*; **C.** *Gonocerca phycidis* Manter, 1925 from *Urophycis chuss*; **D.** *Deropristis inflata* (Molin, 1859) from *Anguilla anguilla*; **E.** *Phyllodistomum bavuri* Boomker, 1984 from *Clarias gariepinus*; **F.** *Paralecithobotrys africanus* Manter et Pritchard, 1964 from *Pseudocrenilabrus philander*; **G.** *Saccocoelium obesum* Looss, 1902 from *Liza aurata*; **H.** *Nematobothrium labeonis* McClelland, 1955 from *Labeo coubie* or *L. horie* (host not specified). (Modified from Odhner 1902; Manter 1925; McClelland 1955; Boomker 1984; Blasco-Costa *et al.* 2009; A, B and F – holotypes illustrated by T. Scholz.)

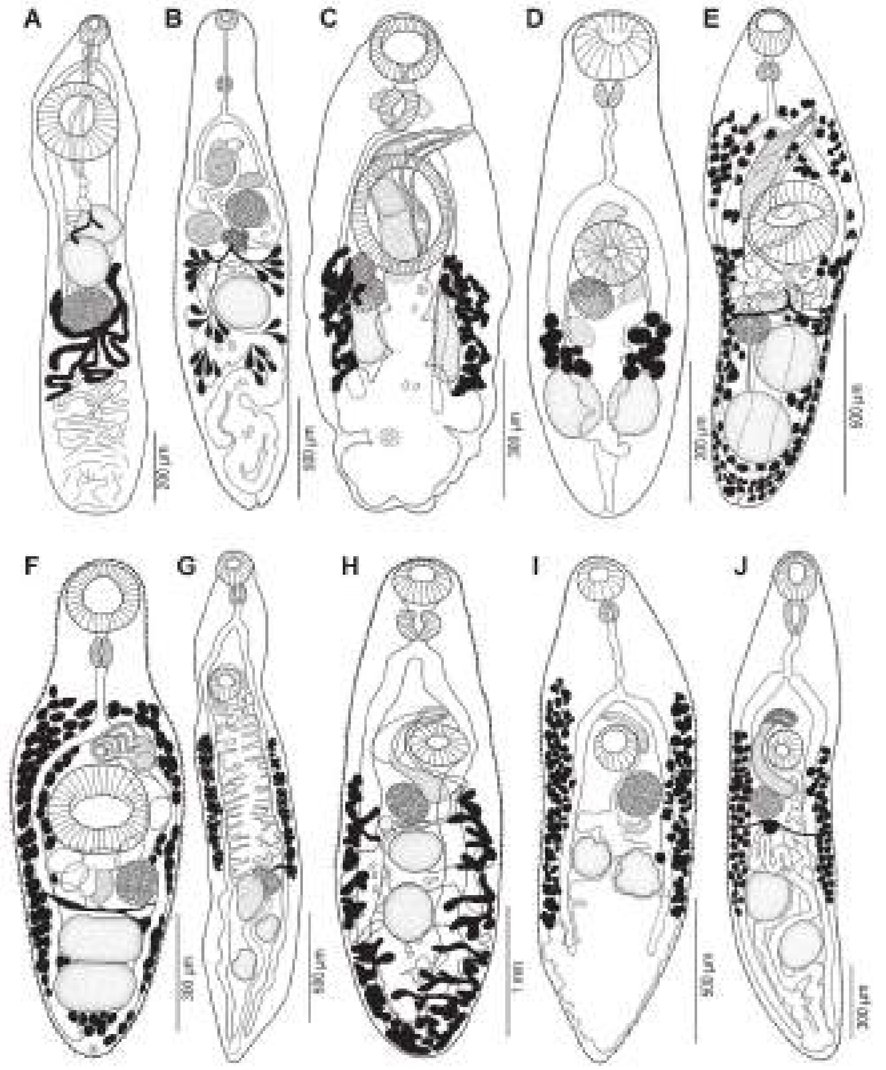

Fig. 4.5.5. Trematoda. **A.** *Ectenurus labeonis* (Fischthal et Kuntz, 1963) from *Labeo forskalii*; **B.** *Haplorchoides cahirinus* (Looss, 1896) from *Bagrus bajad*; **C.** *Heterorchis senegalensis* Vassiliadès et Richard, 1970 from *Protopterus annectens*; **D.** *Malawitrema staufferi* Bray et Hendrix, 2007 from *Clarias gariepinus*; **E.** *Nicolla gallica* (Dollfus, 1941) from *Cottus gobio*; **F.** *Plagioporus niloticus* Vercammen-Grandjean, 1960 from *Oreochromis niloticus*; **G.** *Thaparotrema botswanensis* Van Rensburg, Van As et King, 2013 from *C. gariepinus*; **H.** *Orientocreadium batrachoides* Tubangui, 1931 from *Clarias batrachus* or *Glossogobius giurus* (host not specified); **I.** *Astiotrema turneri* Bray, Van Oosterhout, Blais et Cable, 2006 from *Maylandia zebra*; **J.** *Glossidium lazerae* (Khalil, 1972) from *C. gariepinus*. (Modified from Looss 1899; Dollfus 1959; Vercammen-Grandjean 1960; Fischthal & Kuntz 1963; Vassiliadès & Richard 1970; Khalil 1972; Bray *et al.* 2006; Bray & Hendrix 2007; Jones & Bray 2008; Van Rensburg *et al.* 2013.)

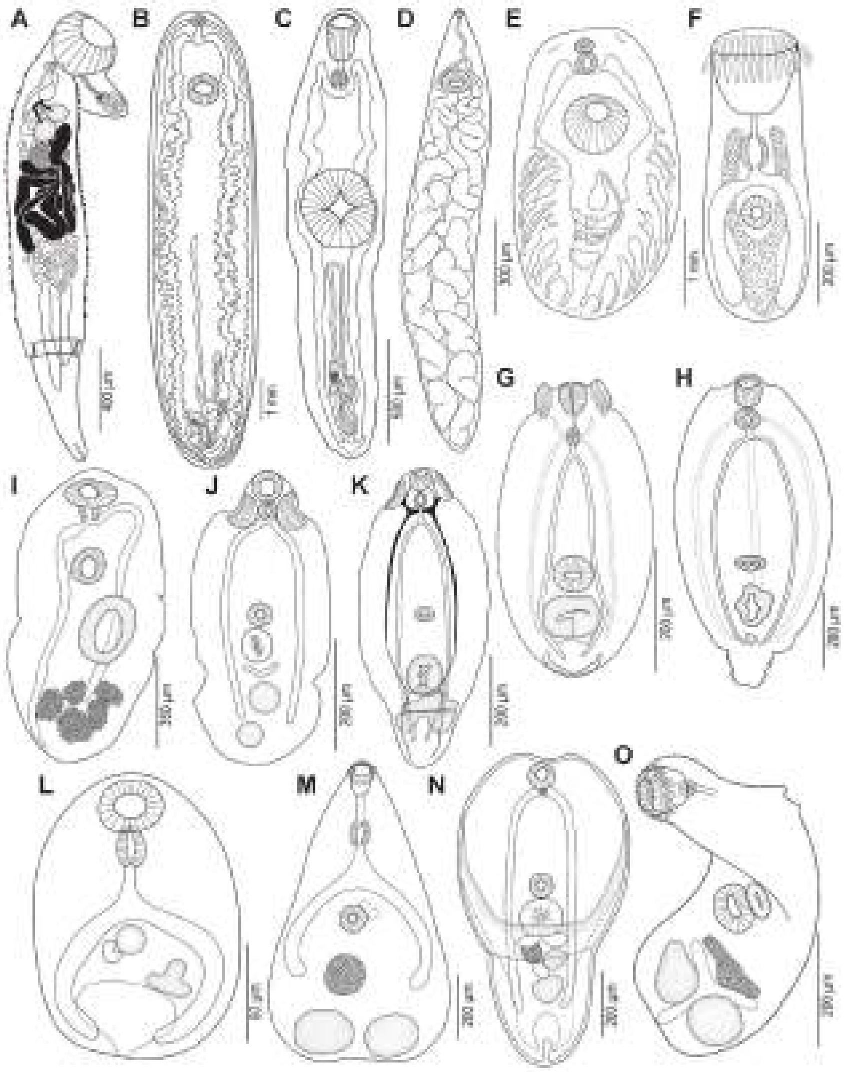

Fig. 4.5.6. Trematoda. **A.** *Dinurus gizae* Fischthal et Kuntz, 1963 from *Hydrocynus forskahlii*; **B.** *Clinostomoides brieni* Dollfus, 1950 from *Clarias gariepinus*; **C.** *Nephrocephalus bagriincapsulatus* (Wedl, 1861) from *Heterotis niloticus*; **D.** *Didymozoidae* gen. sp. from *H. forskahlii*; **E.** *Euclinostomum heterostomum* (Rudolphi, 1809) from *Coptodon zillii*; **F.** Cryptogonimidae gen. sp. (as Metacercariae alestesi Fain, 1953) from *Alestes baremoze* or *C. gariepinus* (host not specified); **G.** *Diplostomum montanum* Zhokhov, 2014 from a cyprinid fish (host not specified); **H.** *Neodiplostomum* sp. from *C. gariepinus*; **I.** *Ornithodiplostomum* sp. from *C. gariepinus*; **J.** *Posthodiplostomoides leonensis* (Williams, 1967) from *Epiplatys* spp.; **K.** *Tylodelphys mashonensis* Beverley-Burton, 1963 from *C. gariepinus*; **L.** *Heterophyes* sp. from *Bagrus bajad*; **M.** *Pygidiopsis genata* Looss, 1907 from non-specified host; **N.** *Posthodiplostomum nanum* Dubois, 1937 from *Epiplatys spilargyreius*; **O.** *Centrocestus cuspidatus* (Looss, 1896) from *Gambusia affinis*. (Modified from Fain 1953; Beverly-Burton 1963; Fischthal & Kuntz 1963; Williams 1967a, b; Fischthal & Thomas 1972; Moravec 1977; Barson & Avenant-Oldewage 2006; Van Rensburg *et al.* 2013; Zhokhov 2014.)

Systematic survey of flukes (Trematoda) in African freshwater fishes

Trematodes identified at least to the genus level are ordered according to their families, which are listed alphabetically, as are their fish hosts. Identification keys for trematode families and genera are provided in the Keys to the Trematoda (Gibson *et al.* 2002; Jones *et al.* 2005; Bray *et al.* 2008). Type species and type hosts are highlighted in bold. The African country where the type locality lies is given if known. Fish names follow FishBase (Froese & Pauly 2017).

List of adult flukes (Trematoda) from African freshwater fishes

Subclass **Aspidogastrea** Faust et Tang, 1936

ASPIDOGASTRIDAE Poche, 1907

Aspidogaster Baer, 1827

Aspidogaster africanus Saoud, Mohamed et Abdel-Hamid, 1974 from *Chrysichthys nigrodigitatus, Labeobarbus bynni* [Fig. 4.5.2A]

Aspidogaster limacoides Diesing, 1834 from *Barbus* sp.

Subclass **Digenea** Carus, 1863

ALLOCREADIIDAE Stossich, 1903

Allocreadium Looss, 1900

Allocreadium aswanensis El-Naffar, Saoud et Hassan, 1984 from ***Labeobarbus bynni*** (Egypt)

Allocreadium engraulicypridis Khalil et Thurston, 1973 from ***Rastrineobola argentea*** (Uganda)

Allocreadium ghanensis Fischthal et Thomas, 1972 from *Synodontis batensoda*, ***Synodontis* sp.** (Ghana)

Allocreadium indistinctum Baer, 1959 from ***Barbus* sp.** (Democratic Republic of the Congo)

Allocreadium mazoensis Beverly-Burton, 1962 from ***Clarias gariepinus*** (Zimbabwe), *Enteromius camptacanthus*, *E. paludinosus*, *E. trimaculatus*, *Haplochromis teegelaari*, *Labeobarbus marequensis* [Fig. 4.5.2B]

Allocreadium sudanensis Saoud, Abdel-Hamid et Ibrahim, 1974 from ***Labeobarbus bynni*** (Sudan)

Allocreadium voltanum Thomas, 1957 from ***Brycinus macrolepidotus*** (Black Volta River)

APOCREADIIDAE Skrjabin, 1942

Trematobrien Dollfus, 1950

Trematobrien haplochromios Dollfus, 1950 from ***Pseudocrenilabrus philander*** (Democratic Republic of the Congo) [Fig. 4.5.2C]

APOROCOTYLIDAE Odhner, 1912

Sanguinicola Plehn, 1905

Sanguinicola chalmersi Odhner, 1924 from ***Auchenoglanis occidentalis*** (Sudan), *Synodontis schall* [Fig. 4.5.2D]

Sanguinicola clarias Imam, Marzouk, Hassan et Itman, 1984 from ***Clarias gariepinus*** (Egypt)

CALLODISTOMIDAE Poche, 1926

Callodistomum Odhner, 1902

Callodistomum diaphanum Odhner, 1902 from *Ctenopoma kingsleyae*, ***Polypterus bichir*** (Sudan), *P. endlicheri* [Fig. 4.5.2E]

Cholepotes Odhner, 1910

Cholepotes ovofarctus (Odhner, 1902) from *Synodontis schall*, ***Synodontis* sp.** (Sudan) [Fig. 4.5.2F]

CEPHALOGONIMIDAE Looss, 1899

Emoleptalea Looss, 1900

Emoleptalea exilis (Looss, 1899) from ***Bagrus bajad*** (Egypt) [Fig. 4.5.2G]

Emoleptalea nwanedi King, Smit, Baker et Luus-Powell, 2018 from *Schilbe intermedius* (South Africa)

Emoleptalea rifaati (Ramadam, Saoud et Taha, 1987) from ***Synodontis schall***, ***S. serratus*** (type host not explicitly mentioned) (Egypt)

Emoleptalea synodontidos Dollfus, 1950 from ***Synodontis notatus*** (Democratic Republic of the Congo)

Emoleptalea sp. from *Nothobranchius furzeri*, *N. kadleci*

Masenia Chatterji, 1933

Masenia bangweulensis (Beverly-Burton, 1962) from *Clarias gariepinus*, ***C. ngamensis*** (Zambia), *Heterobranchus isopterus*

Masenia ghanensis (Fischthal et Thomas, 1968) from *Clarias gariepinus*, *Heterobranchus isopterus*, ***H. longifilis*** (Ghana) [Fig. 4.5.3A]

Masenia proteropora (Thomas, 1958) from ***Clarias anguillaris*** (Ghana)

Masenia synodontis (Khalil et Thurston, 1973) from ***Synodontis victoriae*** (Uganda)

CLADORCHIIDAE Fischoeder, 1901

Basidiodiscus Fischthal et Kuntz, 1959

Basidiodiscus ectorchis Fischthal et Kuntz, 1959 from *Mormyrus kannume*, *Synodontis batensoda*, *S. clarias*, ***S. schall*** (Egypt) [Fig. 4.5.3B]

Brevicaecum McClelland, 1957

Brevicaecum niloticum McClelland, 1957 from ***Citharinus citharus*** (Sudan) [Fig. 4.5.3C]

Panamphistomum Manter et Pritchard, 1964

Panamphistomum benoiti Manter et Pritchard, 1964 from *Clarias gariepinus*, ***Pseudocrenilabrus philander*** (Democratic Republic of the Congo) [Fig. 4.5.3D]

Sandonia McClelland, 1957

Sandonia sudanensis McClelland, 1957 from *Bagrus docmak*, ***Distichodus nefasch***, *D. rostratus*, *Synodontis batensoda*, *S. budgetti*, *S. clarias*, *S. membranaceus*, *S. nigrita*, *S. ocellifer*, ***S. schall*** (type host not explicitly mentioned; Sudan), *S. sorex*, *S. vermiculatus*, *Synodontis* sp. [Fig. 4.5.3E]

CRYPTOGONIMIDAE Ward, 1917

Acanthostomum Looss, 1899

Acanthostomum absconditum (Looss, 1901) from ***Bagrus bajad***, *B. docmak* (Egypt) [Fig. 4.5.3F]

Acanthostomum gymnarchi (Dollfus, 1950) from ***Gymnarchus niloticus*** (Sudan)

Acanthostomum spiniceps (Looss, 1896) from ***Bagrus bajad*** (Egypt), *B. docmak*, *B. filamentosus*, *Chrysichthys nigrodigitatus*

Brientrema Dollfus, 1950

Brientrema malapteruri Dollfus, 1950 from *Distichodus lusosso*, ***Malapterurus electricus*** (Democratic Republic of the Congo) [Fig. 4.5.3G]

Neocladocystis Manter et Pritchard, 1969

Neocladocystis congoensis Manter et Pritchard, 1969 from ***Parauchenoglanis monkei*** (Cameroon) [Fig. 4.5.4A]

Neocladocystis tanganyikae (Prudhoe, 1951) from **unidentified cichlid** (Democratic Republic of the Congo)

Siphodera Linton, 1910

Siphodera ghanensis Fischthal et Thomas, 1968 from ***Chrysichthys nigrodigitatus*** (Ghana), *Hydrocynus brevis*, *Lutjanus goreensis* [Fig. 4.5.3H]

DEROGENIDAE Nicoll, 1910

Halipegus Looss, 1899

Halipegus ctenopomi Jones, 1982 from ***Ctenopoma kingsleyae*** (Senegal) [Fig. 4.5.4B]

DEROPRISTIDAE Cable et Hunninen, 1942

Deropristis Odhner, 1902

Deropristis inflata (Molin, 1859) from *Anguilla anguilla* [Fig. 4.5.4D]

DIDYMOZOIDAE Poche, 1907

Nematobothrium van Beneden, 1858

Nematobothrium labeonis McClelland, 1955 from ***Labeo coubie***, *L. forskalii*, ***L. horie*** [type host not explicitly mentioned], *L. niloticus* (Sudan) [Fig. 4.5.4H]

Nematobothrium sp. from *Labeo coubie*, *L. senegalensis*

GONOCERCIDAE Skrjabin et Guschanskaja, 1955

Gonocerca Manter, 1925

Gonocerca phycidis Manter, 1925 from *Clarias gariepinus* [Fig. 4.5.4C]

GORGODERIDAE Looss, 1899

Phyllodistomum Braun, 1899

Phyllodistomum bavuri Boomker, 1984 from ***Clarias gariepinus*** (South Africa) [Fig. 4.5.4E]

Phyllodistomum ghanense Thomas, 1958 from *Ctenopoma kingsleyae*, ***Mastacembelus nigromarginatus*** (Ghana)

Phyllodistomum linguale Odhner, 1902 from ***Gymnarchus niloticus*** (Egypt)

Phyllodistomum spatula (Odhner, 1902) from ***Bagrus bajad*** (Egypt), *B. docmak*

Phyllodistomum spatulaeforme (Odhner, 1902) from ***Malapterurus electricus*** (Egypt)

Phyllodistomum symmetrorchis Thomas, 1958 from ***Auchenoglanis occidentalis*** (Ghana), *Bagrus bajad*

Phyllodistomum cf. *symmetrorchis sensu* Cutmore, Miller, Curran, Bennett et Cribb, 2013 from *Clarias gariepinus*

Phyllodistomum tana Zhokhov, 2010 from ***Clarias gariepinus*** (Ethiopia)

Phyllodistomum vanderwaali Prudhoe et Hussey, 1977 from ***Clarias gariepinus*** (South Africa)

HAPLOPORIDAE Nicoll, 1914

Paralecithobotrys Freitas, 1948

Paralecithobotrys africanus Manter et Pritchard, 1964 from ***Pseudocrenilabrus philander*** (Democratic Republic of the Congo) [Fig. 4.5.4F]

Saccocoelium Looss, 1902

Saccocoelium obesum Looss, 1902 from *Chelon ramada*, *Mugil cephalus* [Fig. 4.5.4G]

HEMIURIDAE Looss, 1899*

Dinurus Looss, 1907

Dinurus gizae Fischthal et Kuntz, 1963 from ***Hydrocynus forskahlii*** (Egypt) [Fig. 4.5.6A]

Ectenurus Looss, 1907

Ectenurus labeonis (Fischthal et Kuntz, 1963) from ***Labeo forskalii*** (Egypt) [Fig. 4.5.5A]

*The marine genus *Lecithochirium* Lühe, 1901 is not included because reports of *L. magnicaudatum* Fischthal et Kuntz, 1963 from *Labeo forskalii* and *L. musculus* Looss, 1907 from *Heterotis niloticus* are doubtful and apparently represent accidental infections.

HETEROPHYIDAE Leiper, 1909

Haplorchoides Chen, 1949

Haplorchoides cahirinus (Looss, 1896) from ***Bagrus bajad*** (Egypt), *B. docmak*, *B*. *meridionalis*, *Clarias gariepinus* [Fig. 4.5.5B]

MACRODEROIDIDAE McMullen, 1937

Malawitrema Bray et Hendrix, 2007

Malawitrema staufferi Bray et Hendrix, 2007 from *Bagrus meridionalis*, ***Clarias gariepinus*** (Malawi) [Fig. 4.5.5D]

OPECOELIDAE Ozaki, 1925

Nicolla Wiśniewski, 1933

Nicolla gallica (Dollfus, 1941) from *Anguilla anguilla* [Fig. 4.5.5E]

Plagioporus Stafford, 1904

Plagioporus niloticus Vercammen-Grandjean, 1960 from *Anguilla anguilla*, ***Oreochromis niloticus*** (Democratic Republic of the Congo) [Fig. 4.5.5F]

OPISTHORCHIIDAE Looss, 1899

Thaparotrema Gupta, 1955

Thaparotrema botswanensis Van Rensburg, Van As et King, 2013 from ***Clarias gariepinus*** (Botswana) [Fig. 4.5.5G]

Thaparotrema piscicola (Odhner, 1902) from ***Gymnarchus niloticus*** (Sudan)

ORIENTOCREADIIDAE Yamaguti, 1958

Orientocreadium Tubangui, 1931

Orientocreadium batrachoides Tubangui, 1931 from *Clarias anguillaris*, *C. gariepinus*, *C*. *ngamensis* [Fig. 4.5.5H]

Orientocreadium indicum Pande, 1934 from *Clarias gariepinus*, *Heterobranchus longifilis*

Genera *incertae sedis* in the superfamily Plagiorchioidea (*sensu lato*)

Astiotrema Looss, 1899

Astiotrema impletum (Looss, 1899) from ***Tetraodon lineatus*** (Egypt)

Astiotrema lazeri El-Naffar, Saoud et Hassan, 1984 from ***Clarias gariepinus*** (Egypt)

Astiotrema reniferum (Looss, 1898) from *Bagrus docmak*, *Clarias gariepinus*

Astiotrema turneri Bray, Van Oosterhout, Blais et Cable, 2006 from *Labeotropheus trewavasae*, *Maylandia emmiltos*, ***M. zebra*** (Malawi), *Melanochromis vermivorus* [Fig. 4.5.5I]

Glossidium Looss, 1899

Glossidium lazerae (Khalil, 1972) from ***Clarias gariepinus*** (Sudan) [Fig. 4.5.5J]

Glossidium pedatum Looss, 1899 from ***Bagrus bajad***, ***B. docmak*** (type host not explicitly mentioned) (Egypt), *Clarias gariepinus*

Heterorchis Baylis, 1915

Heterorchis crumenifer Baylis, 1915 from ***Protopterus aethiopicus*** (Uganda), *P. annectens*, *Protopterus* sp.

Heterorchis protopteri Thomas, 1958 from ***Protopterus annectens*** (Ghana)

Heterorchis senegalensis Vassiliadès et Richard, 1970 from ***Protopterus annectens*** (Senegal) [Fig. 4.5.5C]

List of metacercariae of flukes (Trematoda) from African freshwater fishes

Note: Due to the simple morphology of larval stages, the existence of morphologically similar species and the lack of knowledge on trematode life cycles, reliable identification of larval stages to the species level is usually impossible based only on morphological characteristics. As a result, some morphology-based identification of metacercariae reported from freshwater fishes in Africa may be misidentifications. Molecular techniques have proven to be efficient for identification and elucidation of the life cycles of parasites and should be applied in future studies.

CLINOSTOMIDAE Lühe, 1901

Clinostomoides Dollfus, 1950

Clinostomoides brieni Dollfus, 1950 from *Clarias gariepinus* [Fig. 4.5.6B]

Clinostomum Leidy, 1856

Clinostomum chrysichthys Dubois, 1930 from *Chrysichthys auratus*

Clinostomum complanatum (Rudolphi, 1819) from *Chrysichthys nigrodigitatus*, *Coptodon zillii*, *Enteromius multilineatus*, *Oreochromis niloticus*, *Sarotherodon galilaeus*

Clinostomum macrosomum Jaiswal, 1957 from *Clarias gariepinus*, *Oreochromis niloticus*

Clinostomum tilapiae Ukoli, 1966 from *Chromidotilapia guntheri*, *Coptodon zillii*, *Cyprinus carpio*, *Hemichromis fasciatus*, *Oreochromis mossambicus*, *O. niloticus*, *Pelmatolapia mariae*, *Sarotherodon galilaeus*, *S. melanotheron*

Clinostomum vandehorsti Ortlepp, 1935 from *Marcusenius macrolepidotus*, *Schilbe mystus*

Clinostomum sp. from *Amphilius uranoscopus*, *Chiloglanis pretoriae*, *Citharinus citharus*, *Clarias gariepinus*, *Coptodon zillii*, *Ctenopoma kingsleyae*, *Epiplatys* sp., *Haplochromis obliquidens*, *Hemichromis fasciatus*, *Labeo coubie*, *Labeobarbus beso*, *Oreochromis leucostictus*, *O*. *mossambicus*, *O*. *niloticus*, *Oreochromis* sp., *Sarotherodon galilaeus*, *Schilbe intermedius*, *Synodontis eupterus*, *S*. *nigrita*, *S*. *schall*, *Tilapia* sp.

Euclinostomum Travassos, 1928

Euclinostomum ardeolae El-Naffar et Khalifa, 1981 from *Oreochromis niloticus*

Euclinostomum clarias (Dubois, 1930) from *Clarias angolensis*

Euclinostomum dollfusi Fischthal et Kuntz, 1963 from *Clarias gariepinus*

Euclinostomum heterostomum (Rudolphi, 1809) from *Chromidotilapia kingsleyae*, *Clarias gariepinus*, *Clarias* sp., *Coptodon zillii*, *Oreochromis mossambicus*, *Sarotherodon melanotheron*, *Tilapia* sp. [Fig. 4.5.6E]

Nephrocephalus Odhner, 1902

Nephrocephalus bagriincapsulatus (Wedl, 1861) from *Auchenoglanis occidentalis*, *Bagrus* sp., *Clarias anguillaris*, *Heterotis niloticus* [Fig. 4.5.6C]

CRYPTOGONIMIDAE Ward, 1917

Cryptogonimidae gen. sp. (as Metacercariae alestesi Fain, 1953) from *Alestes baremoze*, *Clarias gariepinus* [Fig. 4.5.6F]

CYATHOCOTYLIDAE Mühling, 1898

Prohemistomum Odhner, 1913

Prohemistomum vivax (Sonsino, 1892) from *Oreochromis niloticus*, *Schilbe mystus*

DIDYMOZOIDAE Poche, 1907

Didymozoidae gen. sp. from *Hydrocynus forskahlii* [Fig. 4.5.6D]

DIPLOSTOMIDAE Poirier, 1886

Diplostomum von Nordmann, 1832

Diplostomum garrae Zhokhov, 2014 from ***Garra dembecha*** (Ethiopia)

Diplostomum heterobranchi (Wedl, 1861) from *Clarias gariepinus*

Diplostomum longicollis Zhokhov, 2014 from ***Enteromius humilis***, ***Garra dembecha*** (type host not specified) (Ethiopia)

Diplostomum magnicaudum El-Naffar, 1979 from *Oreochromis niloticus*

Diplostomum montanum Zhokhov, 2014 from ***Enteromius humilis***, ***Garra dembecha***, ***Labeobarbus beso***, ***L. gorgorensis*** (type host not specified) (Ethiopia) [Fig. 4.5.6G]

Diplostomum tilapiae Zhokhov, 2014 from ***Oreochromis niloticus*** (Ethiopia)

Diplostomum sp. from *Clarias gariepinus*, *Enteromius humilis*, *Garra dembecha, Oreochromis mossambicus*, *Synodontis nigrita*

Dolichorchis Dubois, 1961

Dolichorchis tregenna (Nazmi Gohar, 1932) from *Clarias gariepinus*

Neodiplostomum Railliet, 1919

Neodiplostomum sp. from *Clarias gariepinus* [Fig. 4.5.6H]

Ornithodiplostomum Dubois, 1936

Ornithodiplostomum sp. from *Clarias gariepinus* [Fig. 4.5.6I]

Posthodiplostomoides Williams, 1969

Posthodiplostomoides leonensis (Williams, 1967) from *Epiplatys sexfasciatus*, *E. spilargyreius* [Fig. 4.5.6J]

Posthodiplostomum Dubois, 1936

Posthodiplostomum nanum Dubois, 1937 from *Coptodon zillii*, *Enteromius humilis*, *Epiplatys sexfasciatus*, *E. spilargyreius*, *Garra dembecha*, *Hemichromis fasciatus*, *Heterobranchus longifilis* [Fig. 4.5.6N]

Tylodelphys Diesing, 1850

Tylodelphys grandis Zhokhov, Morozova et Tessema, 2010 from ***Clarias gariepinus*** (Ethiopia)

Tylodelphys mashonensis Beverley-Burton (1963) from *Clarias gariepinus*, *C. ngamensis* [Fig. 4.5.6K]

Tylodelphys sp. from *Clarias gariepinus*, *Coptodon zillii*, *Cyprinus carpio*, *Micropterus salmoides*, *Oreochromis leucostictus*

Diplostomidae gen. sp. from *Clarias gariepinus*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii*

ECHINOCHASMIDAE Odhner, 1910

Echinochasmus Dietz, 1909

Echinochasmus liliputans (Looss, 1896) from *Oreochromis niloticus*

HETEROPHYIDAE Leiper, 1909

Centrocestus Looss, 1899

Centrocestus cuspidatus (Looss, 1896) from *Gambusia affinis* [Fig. 4.5.6O]

Haplorchis Looss, 1899

Haplorchis sp. from *Oreochromis niloticus*

Heterophyes Cobbold, 1866

Heterophyes aequalis Looss, 1902 from *Oreochromis niloticus*

Heterophyes heterophyes (von Siebold, 1852) from *Oreochromis niloticus*

Heterophyes sp. from *Bagrus bajad* [Fig. 4.5.6L]

Pygidiopsis Looss, 1907

Pygidiopsis genata Looss, 1907 from *Oreochromis niloticus*, *Synodontis batensoda* [Fig. 4.5.6M]

Stellantchasmus Onji et Nishio, 1915

Stellantchasmus pseudocirratus (Witenberg, 1929) from *Oreochromis niloticus*

Stictodora Looss, 1899

Stictodora sawakinensis Looss, 1899 from *Oreochromis niloticus*

PROTERODIPLOSTOMIDAE Dubois, 1936

Pseudoneodiplostomum Dubois, 1936

Pseudoneodiplostomum thomasi (Dollfus, 1935) from *Clarias anguillaris*

STRIGEIDAE Railliet, 1919

Apatemon Szidat, 1928

Apatemon barbusi Zhokhov, Miretskaya, Pugacheva et Tessema, 2008 from *Enteromius humilis*, *E. pleurogramma*, ***E. tanapelagius*** (Ethiopia), *Labeobarbus acutirostris*, *L. dainellii*, *L. gorguari*, *L. intermedius*, *L. nedgia*, *L. beso*

Apatemon tilapiae Zhokhov, Miretskaya, Pugacheva et Tessema, 2008 from ***Oreochromis niloticus*** (Ethiopia)

Apatemon sp. from *Nothobranchius furzeri*

Ichthyocotylurus Odening, 1969

Ichthyocotylurus sp. from *Garra dembecha*

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Chapter 4.6.

CESTODA

Tomáš SCHOLZ & Roman KUCHTA

Tapeworms (Cestoda) – basic characteristics, life cycles, classification and principal diagnostic features

- parasitic flatworms (Platyhelminthes: Neodermata)
- about 5,000 species classified in 19 orders
- obligate endoparasites usually of the digestive system of all groups of vertebrates
- body (strobila) dorsoventrally flattened, usually composed of proglottids (“segments”)
- attachment organs (bothria, bothridia, suckers) on the anterior end called scolex (may be absent)
- digestive tract (intestine) absent
- body surface with hair-like structures called microtriches (absorption of nutrients and attachment)
- all African species hermaphroditic, with well-developed vitellarium (vitelline follicles)
- indirect life cycles (1-2 intermediate hosts: copepods, amphipods, oligochaetes, fishes) [Fig. 4.6.1]
- causative agents of human fish-borne diseases (*e.g.*, broad fish tapeworm) – not in Africa

The classification of cestodes is based on well-defined orders (19 in total at present – Caira & Jensen 2017). They are characterised mainly by the morphology of the scolex (the number and type of attachment organs such as paired bothria and bothridia or four muscular suckers – see Figs 4.6.2B, 4.6.5A) and the morphology of the reproductive organs such as the structure of vitelline follicles (diffuse in the cortex, in lateral bands or compact), position of gonads (in the cortex or medulla), egg morphology, structure of the uterus, etc. (*e.g.*, Fig. 4.6.5B; see Khalil *et al.* 1994 for keys to the orders, families and genera of cestodes, and Caira & Jensen 2017 for updated information on all cestodes).

Generic classification and species identification is based on the size and shape of the scolex and its attachment organs such as suckers and apical organs (it may be muscular, *i.e.*, resembling suckers, or glandular, *i.e.*, comprising gland cells), on proglottid morphology, especially the size, position and shape of gonads (*e.g.*, shape, relative position and size of the terminal genitalia, number of testes, extent of vitelline follicles, shape of the uterus and the number of its lateral diverticula, etc.), size and shape of the eggs, morphology of excretory canals, and many other characteristics.

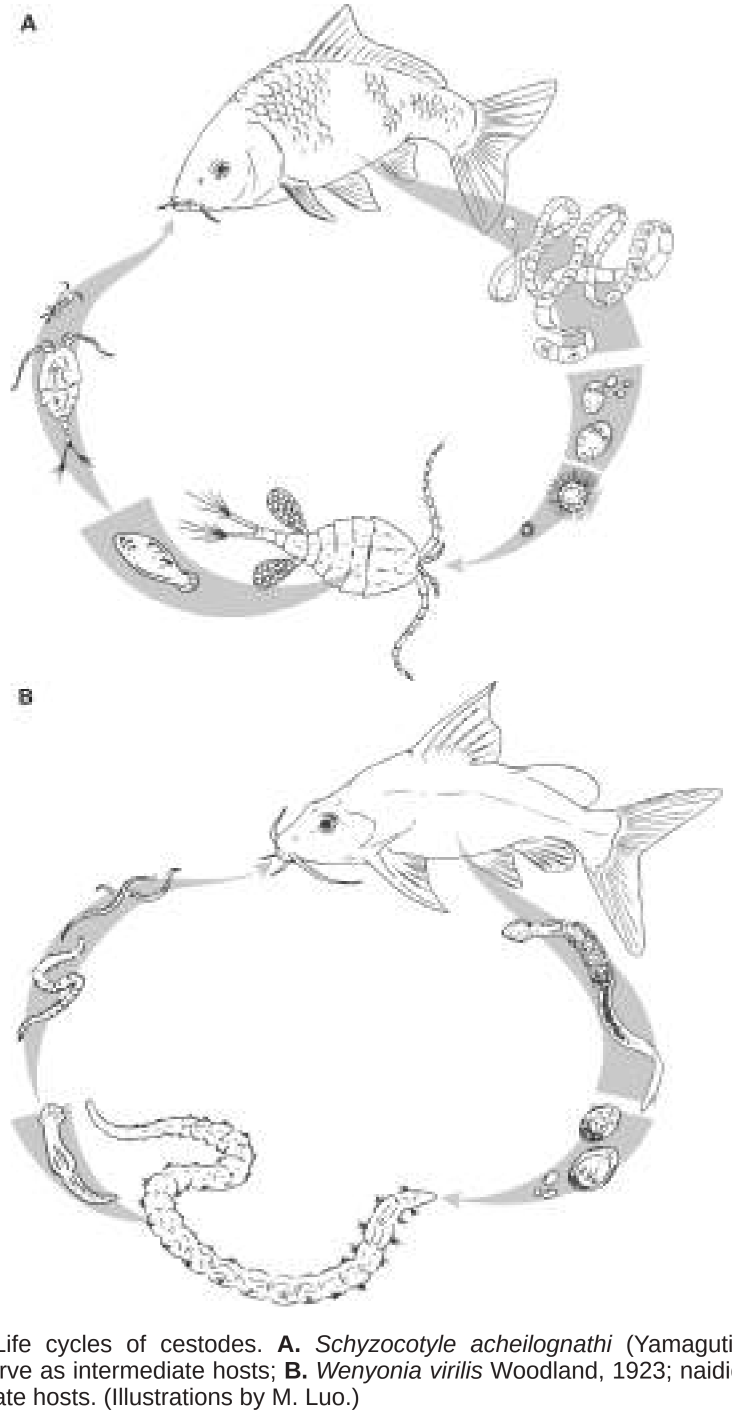

Fig. 4.6.1. Life cycles of cestodes. **A.** *Schyzocotyle acheilognathi* (Yamaguti, 1934); copepods serve as intermediate hosts; **B.** *Wenyonia virilis* Woodland, 1923; naidids serve as intermediate hosts. (Illustrations by M. Luo.)

Key to the orders of tapeworms (Cestoda)

Adults which contain fully developed genital organs and eggs if gravid occur almost exclusively in the intestinal lumen, only *Tetracampos ciliotheca* is also reported to occur in the gall bladder. Larvae called metacestodes, which include plerocercoids and plerocerci, are parenteral, *i.e.*, outside the intestinal lumen except for some tiny larvae of the Gryporhynchidae. Metacestodes do not have fully developed genital organs including eggs and they occur in the mesenteries, intestinal wall, gall bladder and liver.

1 (2) Strobila monozoic (with one genital complex per strobila) [Fig. 4.6.2A]……3

2 (1) Strobila polyzoic (proglottised with several genital complexes per strobila) [Fig. 4.6.2B] ……………………………………………………………………7

3 (4) Strobila without scolex or attachment organ; adults in body cavity [Fig. 4.6.2A]……………………………………………………..**Amphilinidea**

4 (3) Strobila with scolex [Fig. 4.6.3A]………………………………………….. 5

5 (6) Scolex unarmed, without suckers [Fig. 4.6.3E]; adults in intestine …………………………………………………………………**Caryophyllidea**

6 (5) Scolex armed with hooks [Fig. 4.6.6A]; larvae in internal organs ………………………………………..**Cyclophyllidea** (Gryporhynchidae)

7 (8) Scolex with two bothria or attachment grooves [Fig. 4.6.2E]………………9

8 (7) Scolex with four suckers [Fig. 4.6.4A]; adults in intestine…………………………………**Onchoproteocephalidea** (formerly Proteocephalidea)

9 (10) Large worms with weakly developed scolex [Fig. 4.6.5D]; larvae free in body cavity……………………………………………..**Diphyllobothriidea**

10 (9) Scolex with two bothria, armed with hooks or not [Fig. 4.6.2D]; adults in intestine…………………………………………………**Bothriocephalidea**

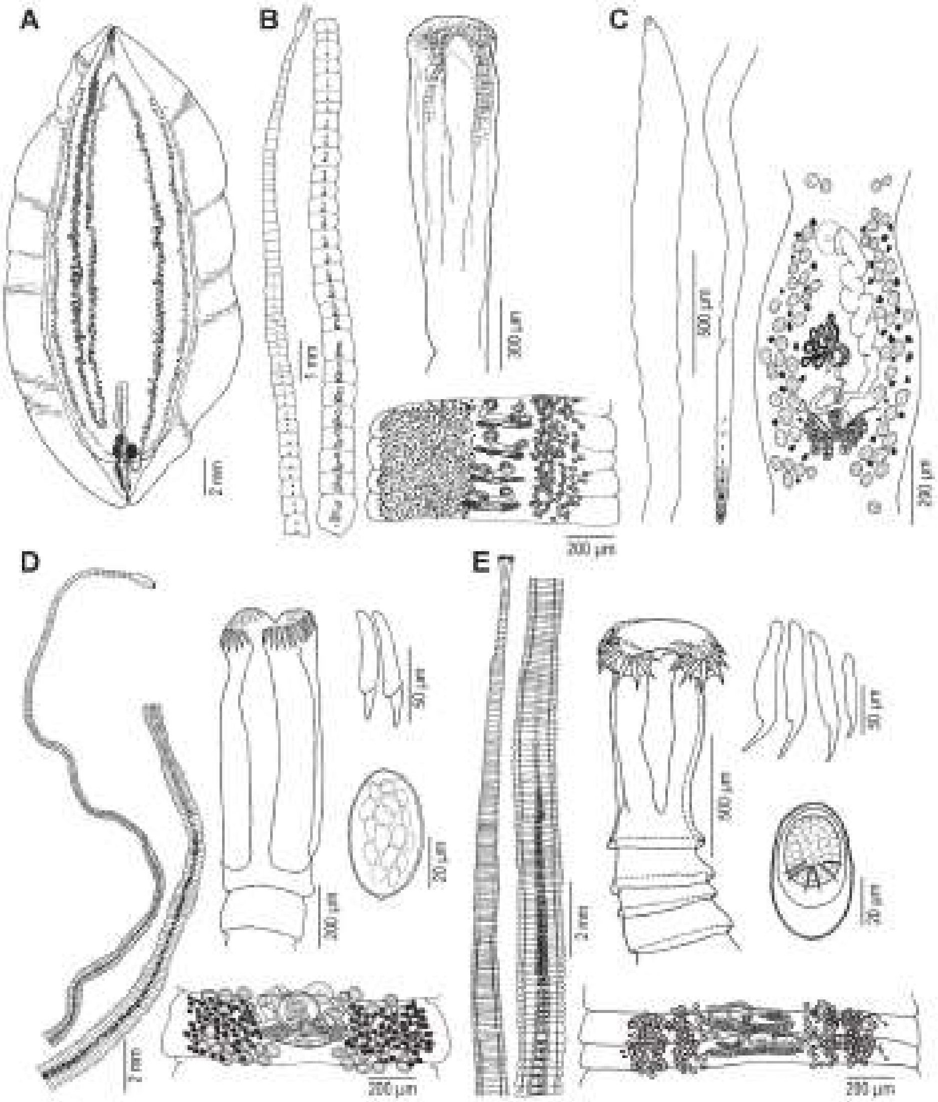

Fig. 4.6.2. Cestoda (Amphilinidea and Bothriocephalidea). **A.** *Nesolecithus africanus* Dönges et Harder, 1966 from *Gymnarchus niloticus*; **B.** *Bothriocephalus claviceps* (Goeze, 1782) from *Anguilla anguilla*; **C.** *Ichthybothrium ichthybori* Khalil, 1971 from *Ichthyborus besse*; **D.** *Kirstenella gordoni* (Woodland, 1937) from *Heterobranchus bidorsalis*; **E.** *Polyonchobothrium polypteri* (Leydig, 1853) from *Polypterus bichir*. (Modified from Dubinina 1982 and Kuchta *et al.* 2012.)

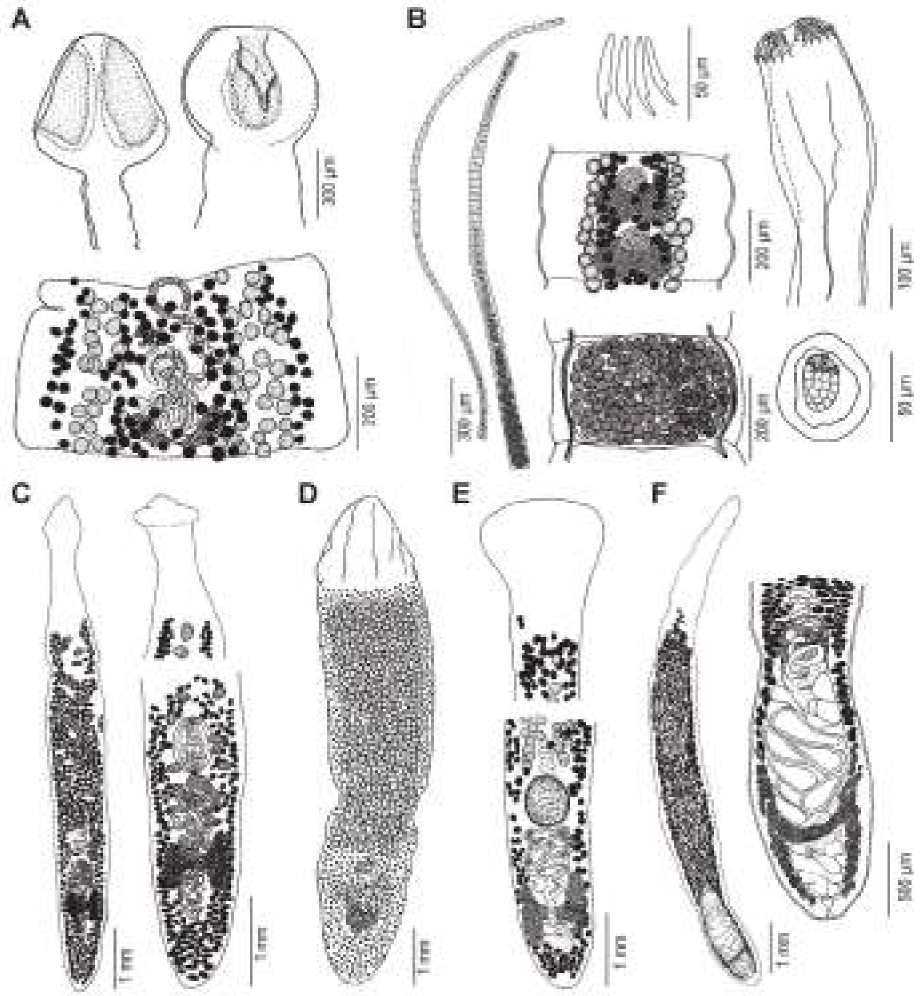

Fig. 4.6.3. Cestoda (Bothriocephalidea and Caryophyllidea). **A.** *Schyzocotyle acheilognathi* (Yamaguti, 1934) from *Cyprinus carpio*; **B.** *Tetracampos ciliotheca* Wedl, 1861 from *Clarias anguillaris*; **C.** *Atractolytocestus huronensis* Anthony, 1958 from *C. carpio*; **D.** *Lytocestoides tanganyikae* Baylis, 1928 from a cichlid; **E.** *Khawia armeniaca* (Cholodkovsky, 1915) from *Arabibarbus grypus*; **F.** *Lytocestus marcuseni* Troncy, 1978 from *Hippopotamyrus harringtoni*. (Modified from Woodland 1937; Troncy 1978; Scholz *et al.* 2011a; Kuchta *et al.* 2012.)

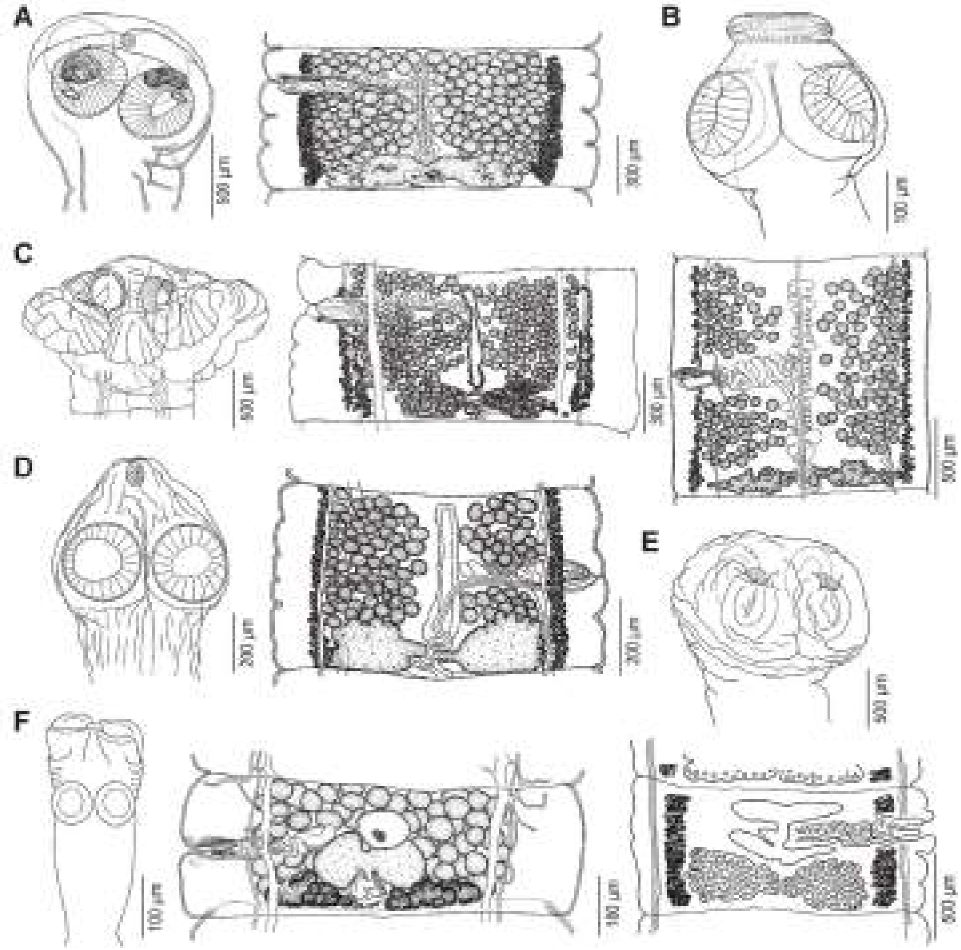

Fig. 4.6.4. Cestoda (Onchoproteocephalidea). **A.** *Barsonella lafoni* de Chambrier, Scholz, Beletew et Mariaux, 2009 from *Clarias gariepinus*; **B.** *Electrotaenia malopteruri* (Fritsch, 1886) from *Malapterurus electricus*; **C.** *Corallobothrium solidum* Fritsch, 1886 from *M. electricus*; **D.** *Proteocephalus synodontis* Woodland, 1925 from *Synodontis schall*; **E.** *Marsypocephalus tanganyikae* (Fuhrmann et Baer, 1925) from *C. gariepinus*; **F.** *Sandonella sandoni* (Lynsdale, 1960) from *Heterotus niloticus*. (Modified from Fuhrmann and Baer 1925; de Chambrier *et al.* 2004, 2008, 2009, 2011; Scholz *et al.* 2011b.)

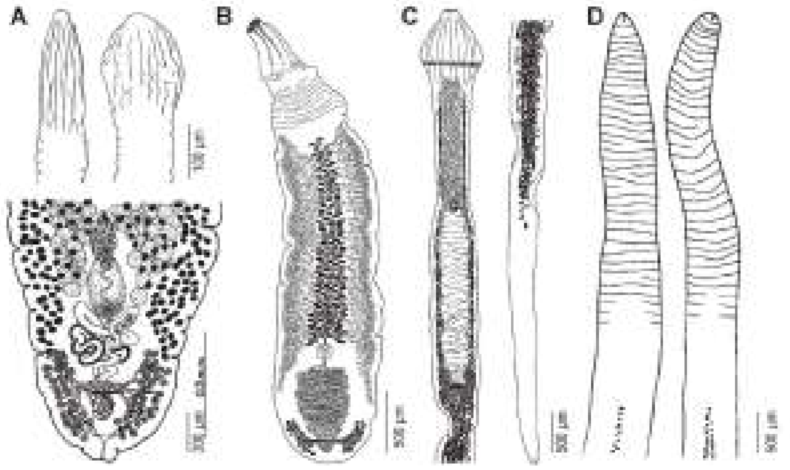

Fig. 4.6.5. Cestoda (Caryophyllidea and Diphyllobothriidea). **A.** *Monobothrioides cunningtoni* Fuhrmann et Baer, 1925 from *Auchenoglanis occidentalis* (scoleces) and *M. woodlandi* Mackiewicz et Beverley-Burton, 1967 from *Clarias ngamensis*; **B.** *Stocksia pujehuni* Woodland, 1937 from *Clarias gariepinus*; **C.** *Wenyonia virilis* Woodland, 1923 from *Synodontis schall*; **D.** *Ligula intestinalis* (Linnaeus, 1758) (plerocercoids) from *Cyprinus carpio*. (Modified from Woodland 1937; Mackiewicz & Beverley-Burton 1967; Dubinina 1980; Schaeffner *et al.* 2011.)

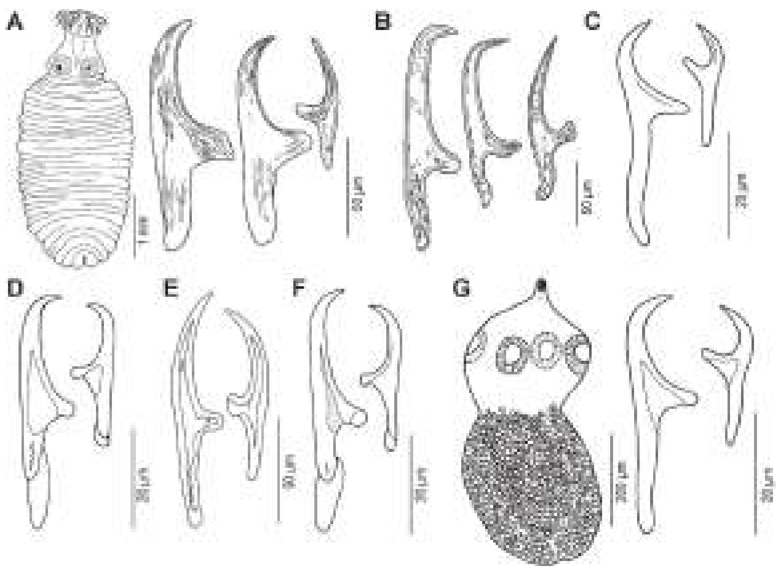

Fig. 4.6.6. Cestoda (Gryporhynchidae – larvae). **A.** *Amirthalingamia macracantha* (Joyeux et Baer, 1935) from *Coptodon zillii*; **B.** *Cyclustera magna* (Baer, 1959) from *C. zillii*; **C.** *Dendrouterina herodiae* Fuhrmann, 1912 from *Schilbe intermedius*; **D.** *Neogryporhynchus lasiopeius* Baer et Bona, 1960 from *Pseudocrenilabrus philander*; **E.** *Paradilepis scolecina* Hsü, 1935 from *P. philander*; **F.** *Parvitaenia samfyia* Mettrick, 1967 from *Tilapia sp.* **G.** *Valipora campylancristrota* (Wedl, 1855) from *Tinca tinca* (total view) and *V. minuta* (Coil, 1950) from *P. philander.* (Modified from Bray 1974; Scholz 1989; Scholz *et al.* 2004, 2018.)

A systematic survey of tapeworms (Cestoda) in African freshwater fish

Species are listed alphabetically according to individual cestode orders. Keys to individual cestode orders, their families and genera were provided by Khalil *et al.* (1994). Kuchta *et al.* (2008) split the order Pseudophyllidea Van Beneden in Carus, 1863 into the Bothriocephalidea and Diphyllobothriidea; members of both orders parasitise teleost fishes, but those of the latter order only as larvae (plerocercoids). The type species of each genus and the type host of each species are highlighted in bold. The country where the type locality occurs is given if known. Host names are according to Froese & Pauly (2017).

AMPHILINIDEA Poche, 1922

List of the Amphilinidea (adults) from African freshwater fishes

Nesolecithus Poche, 1922

Nesolecithus africanus Dönges et Harder, 1966 from ***Gymnarchus niloticus*** (Nigeria) [Fig. 4.6.2A]

BOTHRIOCEPHALIDEA Kuchta, Scholz, Brabec et Bray, 2008

Key to the genera of the Bothriocephalidea (adults) from African freshwater fishes (for keys to the species – see Kuchta *et al.* 2012)

1 (2) Apical part of scolex unarmed (without hooks) [Figs 4.6.2B, C; 4.6.3A]……3

2 (1) Apical part of scolex armed (with hooks) [Figs 4.6.2D, E; 4.6.3B]…………7

3 (4) Proglottids well demarcated from each other; scolex with well-developed bothria……………………………………………………………………………4

4 (3) Proglottids without obvious demarcation; scolex with weakly developed bothria [Fig. 4.6.2C]…………………………………………..***Ichthybothrium***

5 (6) Scolex heart-shaped, with deep, sucker-like bothria [Fig. 4.6.3A]………… ………………………………………………………………….***Schyzocotyle***

6 (5) Scolex elongate to lanceolate, with shallow bothria [Fig. 4.6.2B]………… …………………………………………………………..***Bothriocephalus***

7 (8) Scolex small (< 650 μm); vitelline follicles medullary; testes few (5-20); eggs with transparent, hyaline outer envelope [Fig. 4.6.3B]……………… …………………………………………………………………***Tetracampos***

8 (7) Scolex large (> 700 μm); vitelline follicles cortical, numerous; testes numerous (> 30, usually more than 100); eggs with hard shell capsule…………9

9 (10) Apical disc prominent, wider than scolex, armed with < 36 large, massive hooks, up to 190 μm long; cirrus-sac small, its width representing 5-10% of proglottid width [Fig. 4.6.2E]…………………………..***Polyonchobothrium***

10 (9) Apical disc weakly developed, as wide as scolex or narrower, armed with > 40 hooks shorter than 90 µm; cirrus-sac large, its width representing 16-39% of proglottid width [Fig. 4.6.2D]……………………………..***Kirstenella***

List of the Bothriocephalidea (adults) from African freshwater fishes

Bothriocephalus Rudolphi, 1808

Bothriocephalus claviceps (Goeze, 1782) from ***Anguilla anguilla*** [Fig. 4.6.2B]

Bothriocephalus sp. from *Tilapia* sp.

Ichthybothrium Khalil, 1971

Ichthybothrium ichthybori Khalil, 1971 from ***Ichthyborus besse*** (Sudan), *I. quadrilineatus* [Fig. 4.6.2C]

Ichthybothrium sp. from *Mesoborus crocodilus* [Fig. 4.1F]

Kirstenella Kuchta in Kuchta *et al.* 2012

Kirstenella gordoni (Woodland, 1937) [syn. *Bothriocephalus prudhoei* Tadros, 1966] from ***Heterobranchus bidorsalis*** (Sierra Leone); accidental hosts: *Clarias anguillaris*, *Schilbe mystus* [Fig. 4.6.2D]

Polyonchobothrium Diesing, 1854

Polyonchobothrium polypteri (Leydig, 1853) [syns *Tetrabothrium polypteri* Leydig, 1853; *Onchobothrium septicolle* Diesing, 1854; *Polyonchobothrium septicolle* Diesing 1863; *Anchistrocephalus polypteri* Monticelli, 1900; *Rhynchobothrium polypteri* Klaptocz, 1906; *Polyonchobothrium pseudopolypteri* Meggitt, 1930] from ***Polypterus bichir*** (Egypt), *P. endlicheri*, *P. senegalus* [Fig. 4.6.2E]

Schyzocotyle Akhmerov, 1960

Schyzocotyle acheilognathi (Yamaguti, 1934) [syns *Bothriocephalus acheilognathi* Yamaguti, 1934; *Bothriocephalus* (*Clestobothrium*) *kivuensis* Baer et Fain, 1958; *Bothriocephalus aegyptiacus* Ryšavý et Moravec, 1975; *Bothriocephalus barbus* Fahmy, Mandour et El-Naffar, 1978] from *Carasobarbus fritchii*, *Cyprinus carpio*, *Enteromius annectens*, *E. argenteus*, *E. bifrenatus*, *E. brevipinnis*, *E. mattozi*, *E. paludinosus*, *E. trimaculatus*, *Hydrocynus vittatus*, *Labeobarbus aeneus*, *L. altianalis*, *Labeobarbus bynni*, *L. kimberleyensis*, *L. marequensis*, *L. nedgia*, *Luciobarbus callensis*, *Ptychochromis* cf. *inornatus* [Fig. 4.6.3A]. Records from *Clarias gariepinus*, *Clarotes laticeps*, *Oreochromis mossambicus* and *O. niloticus* need verification.

Tetracampos Wedl, 1861

Tetracampos ciliotheca Wedl, 1861 [syns *Clestobothrium clarias* Woodland, 1925; *Polyonchobothrium cylindraceum* forma *major* Janicki, 1926; *P. cylindraceum* forma *minor* Janicki, 1926; *Polyonchobothrium fulgidum* Meggitt, 1930; *Polyonchobothrium clarias* (Woodland, 1925) Meggitt, 1930; *Polyonchobothrium ciliotheca* (Wedl, 1861) Dollfus, 1934; *Polyoncobothrium ciliotheca* (Wedl, 1861) Yamaguti, 1959; *Polyoncobothrium clarias* (Woodland, 1925) Yamaguti, 1959] from ***Clarias anguillaris*** (Egypt), *C. gariepinus*, *C. liocephalus*, *C. werneri* [Fig. 4.6.3B]

Tetracampos martinae Kuchta in Kuchta *et al.* 2012 from ***Bagrus meridionalis*** (Malawi)

CARYOPHYLLIDEA van Beneden in Carus, 1863

Key to the genera of the Caryophyllidea (adults) from African freshwater fishes

1 (2) Postovarian vitelline follicles present [Figs 4.6.3C-F, 4.6.5C]………………3

2 (1) Postovarian vitelline follicles absent [Fig. 4.6.5 A, B]…………………………7

3 (4) Body with tail-like posterior part; genital pores in anterior half of the body; in mochokid catfishes (*Synodontis*) [Fig. 4.6.5C]………………..***Wenyonia***

4 (3) Body without tail-like posterior part; genital pores near the posterior end of the body………………………………………………………………………5

5 (6) Vitelline follicles present alongside lateral ovarian lobes; in carp (introduced) [Fig. 4.6.3C]……………………………………..***Atractolytocestus***

6 (5) Vitelline follicles absent alongside lateral ovarian lobes; in barbels [Fig. 4.6.3E]………………………………………………………..***Khawia***

7 (8) Vitelline follicles absent medially; in *Clarias* [Fig. 4.6.5B]…………***Stocksia***

8 (7) Vitelline follicles present also medially (on ventral and dorsal side of cortex) ……………………………………………………………………………9

9 (10) Body small (maximum length 11 mm), tapering slightly from conical scolex with longitudinal wrinkles; vitelline follicles extensive, filling most of cortex throughout body; in cichlids [Fig. 4.6.3D]……………………***Lytocestoides***

10 (9) Body larger, of different shapes; vitelline follicles less extensive…………11

11 (12) Scolex with longitudinal wrinkles; in *Auchenoglanis* and *Clarias* [Fig. 4.6.5A]……………………………………………………***Monobothrioides***

12 (12) Scolex elongate, simple, without longitudinal wrinkles; in Alestidae, Mormyridae and *Clarias* [Fig. 4.6.3F]……………………………….***Lytocestus***

List of the Caryophyllidea (adults) from African freshwater fishes

Atractolytocestus Anthony, 1958

Atractolytocestus huronensis Anthony, 1958 from ***Cyprinus carpio*** [introduced with common carp] [Fig. 4.6.3C]

Khawia Hsü, 1935

Khawia armeniaca (Cholodkovsky, 1915) from *Labeobarbus bynni*, *L. callensis*, *L. tropidolepis*, *L. setivimensis* [reported as *Caryophyllaeus laticeps* (Pallas, 1781)] [Fig. 4.6.3E]

Khawia sp. from *Labeobarbus bynni*

Lytocestus Cohn, 1908

Lytocestus filiformis (Woodland, 1923) from ***Mormyrus caschive*** (Sudan), *Mormyrus* sp.

Lytocestus marcuseni Troncy, 1978 [syn. *L. alestesi* Lynsdale, 1956] from *Brycinus nurse*, ***Hippopotamyrus harringtoni*** (Chad) [Fig. 4.6.3F]

Lytocestus puylaerti Khalil, 1973 from ***Clarias buettikoferi*** (Sierra Leone)

Lytocestus sp. from *Auchenoglanis occidentalis*

Lytocestoides Baylis, 1928

Lytocestoides tanganyikae Baylis, 1928 from a **cichlid** (Tanzania) [Fig. 4.6.3D]

Lytocestoides sp. from *Parectodus* sp.

Monobothrioides Fuhrmann et Baer, 1925

Monobothrioides chalmersius (Woodland, 1924) from ***Clarias anguillaris*** (Sudan)

Monobothrioides cunningtoni Fuhrmann et Baer, 1925 from ***Auchenoglanis occidentalis*** (Zambia) [Fig. 4.6.5A]

Monobothrioides tchadensis Troncy, 1978 from ***Auchenoglanis biscutatus*** (Chad)

Monobothrioides woodlandi Mackiewicz et Beverley-Burton, 1967 from ***Clarias ngamensis*** (Zambia) [Fig. 4.6.5A]

Monobothrioides sp. from *Parauchenoglanis ballayi*, *Synodontis schall*

Stocksia Woodland, 1937

Stocksia pujehuni Woodland, 1937 from ***Clarias gariepinus*** (Sierra Leone) [Fig. 4.6.5B]

Wenyonia Woodland, 1923 (see Schaeffner *et al.* 2011 for a key to species)

Wenyonia acuminata Woodland, 1923 from *Synodontis acanthomias*, ***S. membranaceus*** (Sudan)

Wenyonia longicauda Woodland, 1937 from **Synodontis gambiensis** (now considered to be a synonym of *S. schall*) (Sierra Leone)

Wenyonia minuta Woodland, 1923 [syn. *Wenyonia mcconnelli* Ukoli, 1972] from *Synodontis caudovittatus*, *S. frontosus*, *S. nigrita*, *S. schall*, *S. serratus*; ***Chrysichthys auratus*** (Sudan) is incidental host

Wenyonia synodontis Ukoli, 1972 from *Synodontis schall*, ***S. sorex*** (Nigeria), *S. vermiculatus*

Wenyonia virilis Woodland, 1923 [syns *Caryophyllaeus niloticus* Kulmatycki, 1928; *Wenyonia kainjii* Ukoli, 1972] from *Synodontis batensoda*, *S. budgetti*, *S. caudovittatus*, *S. clarias*, *S. eupterus*, *S. frontosus*, *S.* cf. *geledensis*, *S. nigrita*, *S. ocellifer*, ***S. schall*** (Sudan), *S. serratus*, *S. sorex* [Fig. 4.6.5C]

Wenyonia youdeoweii Ukoli, 1972 from *Synodontis caudovittatus*, ***S. gobroni*** (Nigeria), *S. schall*, *S. serratus*

Wenyonia sp. from *Synodontis batensoda*

Key to the genera of the Onchoproteocephalidea (only family Proteocephalidae; adults) from African freshwater fishes

1 (2) Vitellarium formed by numerous follicles arranged in paired lateral bands; scolex without a highly modified apical structure with retractile lappets……………………………………………………………………………3

2 (1) Vitellarium formed by two compact, but deeply lobulated, postovarian masses near the posterior margin of proglottids; scolex with a highly modified apical structure formed by four muscular retractile lappets [Fig. 4.6.4F] ……………………………………………………………………***Sandonella***

3 (4) Scolex umbrella-shaped, with widely pyramidal apex and well-developed metascolex, which forms folded collar surrounding suckers; external margins of suckers with semispherical sphincter; body surface with deep longitudinal and transverse grooves (wrinkles) forming rectangular network; in electric catfish (*Malapterurus*) [Fig. 4.6.4C]……………***Corallobothrium***

4 (3) Scolex of different shapes, metascolex absent; sphincter on suckers and longitudinal and transverse grooves on the strobila usually absent……………………………………………………………………………5

5 (6) Scolex with apical rostellum armed with tiny hooks; in electric catfish (*Malapterurus*) [Fig. 4.6.4B]………………………………………..***Electrotaenia***

6 (5) Scolex without rostellum, without any hooks; in other fishes………………7

7 (8) Tapeworms very large, robust (total length up to 173 mm; maximum width 1.8 mm); scolex round, suckers with muscular sphincter; in clariid catfishes ……………………………………………………………………….9

8 (7) Tapeworms smaller; scolex often conical; suckers without sphincters [Fig. 4.6.4D]………………………………………………..***Proteocephalus***

9 (10) Scolex with additional posterior orifices and muscular sphincters; testes medullary [Fig. 4.6.4A]…………………………………………….***Barsonella***

10 (9) Suckers without additional posterior orifices and muscular sphincters; testes in dorsal cortex [Fig. 4.6.4E]……………………..***Marsypocephalus***

List of the Onchoproteocephalidea (Proteocephalidae; adults) from African freshwater fishes

Barsonella de Chambrier, Scholz, Beletew et Mariaux, 2009

Barsonella lafoni de Chambrier, Scholz, Beletew et Mariaux, 2009 from ***Clarias gariepinus*** (Ethiopia) [Fig. 4.6.4A]

Corallobothrium Fritsch, 1886

Corallobothrium solidum Fritsch, 1886 from ***Malapterurus electricus*** (Egypt) [Fig. 4.6.4C]

Electrotaenia Nybelin, 1942

Electrotaenia malopteruri (Fritsch, 1886) from ***Malapterurus electricus*** (Egypt) [Fig. 4.6.4B]

Marsypocephalus Wedl, 1861

Marsypocephalus aegyptiacus El-Naffar, Saoud et Hassan, 1984 from ***Clarias gariepinus*** (Egypt)

Marsypocephalus daveyi Woodland, 1937 from ***Heterobranchus bidorsalis*** (Sierra Leone)

Marsypocephalus heterobranchus Woodland, 1925 from ***Heterobranchus bidorsalis*** (Sudan)

Marsypocephalus rectangulus Wedl, 1861 from *Clarias anguillaris*, ***C. gariepinus*** (Egypt), *Heterobranchus bidorsalis*

Marsypocephalus tanganyikae (Fuhrmann et Baer, 1925) from ***Clarias gariepinus*** (Zambia) [Fig. 4.6.4E]

Marsypocephalus sp. from *Heterobranchus bidorsalis*

Proteocephalus Weinland, 1858

Proteocephalus beauchampi Fuhrmann et Baer, 1925 from *Chrysichthys brachynema*, ***Chrysichthys* sp.** (Democratic Republic of the Congo); a record from *Synodontis schall* is doubtful

Proteocephalus bivitellatus Woodland, 1923 from a **carnivorous cichlid** (Sierra Leone)

Proteocephalus cunningtoni Fuhrmann et Baer, 1925 from ***Dinotopterus cunningtoni*** (Zambia)

Proteocephalus dinotopteri Fuhrmann et Baer, 1925 from ***Dinotopterus cunningtoni*** (Zambia)

Proteocephalus glanduligerus Janicki, 1928 from ***Clarias anguillaris*** (Egypt), *C. gariepinus*

Proteocephalus membranacei Troncy, 1978 [syn. *Proteocephalus largoproglottis* Troncy, 1978] from ***Synodontis membranaceus*** (Chad)

Proteocephalus pentastomus (Klaptocz, 1906) from ***Polypterus bichir*** (Sudan), *P. endlicheri*, *P. senegalus*

Proteocephalus sulcatus (Klaptocz, 1906) from ***Clarotes laticeps*** (Sudan); accidental hosts: *Chrysichthys* sp., *Clarias anguillaris*, *C. gariepinus*, *Clarotes laticeps*, *Polypterus endlicheri*

Proteocephalus synodontis Woodland, 1925 from *Auchenoglanis* cf. *occidentalis*, *Synodontis batensoda*, *S. caudovittatus*, *S. eupterus*, *S. frontosus*, *S. nigrita*, ***S. schall*** (Sudan), *S. serratus* [Fig. 4.6.4D]

Proteocephalus sp. from *Ichthyborus besse*

Sandonella Khalil, 1960

Sandonella sandoni (Lynsdale, 1960) from ***Heterotis niloticus*** (Sudan) [Fig. 5.6.4F]

CYCLOPHYLLIDEA van Beneden in Braun, 1900 – family Gryporhynchidae Spassky et Spasskaya, 1973

Key to the larvae (metacestodes) of the family Gryporhynchidae (Cyclophyllidea) from African freshwater fishes

1 (2) Hooks of three shapes (4 + 6 + 10 in number), massive, very large (> 200 μm long)……………………………………………………………3

2 (1) Hooks of two different shapes (10 + 10 in number), more delicate, smaller …………………………………………………………………………...5

3 (4) Hooks > 390 μm (larger) and > 240 μm (smaller) long [Fig. 4.6.6A]……… ………………………………………………………….***Amirthalingamia***

4 (3) Hooks < 200 μm (larger) and < 150 μm (smaller) long [Fig. 4.6.6B] ……………………………………………………………………***Cyclustera***

5 (6) Large hooks > 90 μm long, massive, with slightly curved blade [Fig. 4.6.6E] ……………………………………………………………………***Paradilepis***

6 (5) Large hooks < 50 μm long, delicate, with abruptly curved blade……………7

7 (8) Hooks tiny, large hooks < 31 μm long; in gall bladder [Fig. 4.6.6G]………… ……………………………………………………………………….***Valipora***

8 (7) Hooks larger, large hooks > 45 μm long; larvae in other sites of infection…………………………………………………………………………9

9 (10) Blade of larger hooks slightly longer, straighter [Fig. 4.6.6F]……………… ……………………………………………………………………***Parvitaenia***

10 (9) Blade of larger hooks slightly shorter, more curved; hooks 48-50 μm long ……………………………………………………………………………11

11 (12) Hooks more robust, with blade tip of larger hooks directed more anteriorly [Fig. 4.6.6D]…………………………………………..***Neogryporhynchus***

12 (11) Hooks more slender, with blade tip of larger hooks more curved [Fig. 4.6.5C] ……………………………………………………………….***Dendrouterina***

List of the larvae (metacestodes) of the family Gryporhynchidae (Cyclophyllidea) from African freshwater fishes (see Scholz et al. 2018 for a review of African gryporhynchid larvae)

Amirthalingamia Bray, 1974

Amirthalingamia macracantha (Joyeux et Baer, 1935) from *Coptodon zillii*, *Oreochromis niloticus*, *Pharyngochromis acuticeps*, *Pseudocrenilabrus philander*, *Tilapia sparrmanii* [Fig. 4.6.6A]

Anomotaenia Cohn, 1900

Anomotaenia sp. from *Hemichromis fasciatus*, *Oreochromis niloticus*; identification needs verification because the genus belongs to the family Dilepididae and no vouchers were deposited by Aderounmu & Adeniyi (1972).

Cyclustera Fuhrmann, 1901

Cyclustera magna (Baer, 1959) from *Coptodon zillii*, *Labeo horie*, *Oreochromis niloticus*, *Sarotherodon galilaeus* [Fig. 4.6.6B]

Cyclustera sp. from *Cyprinus carpio* – see Scholz *et al.* (2008)

Dendrouterina Fuhrmann, 1912

Dendrouterina herodiae Fuhrmann, 1912 from *Schilbe intermedius* [Fig. 4.6.6C]

Neogryporhynchus Baer et Bona, 1960

Neogryporhynchus lasiopeius Baer et Bona, 1960 from *Bathybathes graueri*, *Chetia flaviventris*, *Coptodon rendalli*, *Cyprinus carpio*, *Heterotis niloticus*, *Oreochromis mossambicus*, *O. niloticus*, *Pseudocrenilabrus philander*, *Tilapia sparrmannii* [Fig. 4.6.6D]

Paradilepis Hsü, 1935

Paradilepis delachauxi (Fuhrmann, 1909) from *Chetia flaviventris*, *Coptodon rendalli*, *Labeobarbus marequensis*, *Oreochromis macrochir*, *Pharyngochromis acuticeps*, *Pseudocrenilabrus philander*

Paradilepis maleki Khalil, 1961 from *Benthochromis horii*, *Pseudocrenilabrus philander*

Paradilepis scolecina Hsü, 1935 from *Coptodon rendalli*, *Enteromius paludinosus*, *E. trimaculatus*, *E. unitaeniatus*, *Labeobarbus kimberleyensis*, *Oreochromis mossambicus*, *Pseudocrenilabrus philander* [Fig. 4.6.6E]

Paradilepis sp. from *Chetia flaviventris*, *Coptodon rendalli*, *Oreochromis mossambicus*, *Pharyngochromis acuticeps*, *Pseudocrenilabrus philander*

Parvitaenia Burt, 1940

Parvitaenia macropeos (Wedl, 1855) from *Coptodon rendalli*, *Hemichromis letourneuxi*, *Oreochromis mossambicus*, *O. niloticus*

Parvitaenia samfyia Metrick, 1967 from *Pseudocrenilabrus philander*, *Tilapia* sp. [Fig. 4.6.6F]

Parvitaenia sp. 1 from *Enteromius treurensis*, *E. trimaculatus*

Parvitaenia sp. 2 from '*Barbus*' sp., *Enteromius macrops*, *E. trimaculatus*

Parvitaenia sp. 3 from *Pseudocrenilabrus philander*

Valipora Linton, 1927

Valipora campylancristrota (Wedl, 1855) from *Enteromius paludinosus*, *Pseudocrenilabrus philander* [Fig. 4.6.5G]

Valipora minuta (Coil, 1950) from *Chetia flaviventris*, *Ophthalmotilapia nasuta*, *Pseudocrenilabrus philander*, *Ptychochromis grandidieri* [Fig. 4.6.5G]

DIPHYLLOBOTHRIIDEA Kuchta, Scholz, Brabec et Bray, 2008

List of the Diphyllobothriidea (larvae) from African freshwater fishes

Ligula Bloch, 1782

Ligula intestinalis (Linnaeus, 1758) (plerocercoids) from *Chagunius nicholsi*, *Enteromius kamolondoensis*, *E. lineomaculatus*, *E. lukusiensis*, *E. paludinosus*, *E. radiatus*, *E. unitaeniatus*, *Haplochromis* sp., *Labeobarbus marequensis*, *L. microbarbis*, *Labeo lukulae*, *Rastrineobola argentea* [Fig. 4.6.5D]

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Chapter 4.7.

ACANTHOCEPHALA

Bernd SURES, Yuriy KVACH & Roman KUCHTA

Thorny-headed worms (Acanthocephala) – basic characteristics, life cycles, classification and principal diagnostic features

- parasitic 'worms' (Syndermata: Rotifera)
- about 1,300 species classified in 4 classes (Archiacanthocephala, Eoacanthocephala, Palaeacanthocephala and Polyacanthocephala) and 10 orders (Amin 2013; Warner 2014), from which 19 species occur in Africa
- heteroxenous parasites, with adults in the intestine of vertebrates (definitive hosts) and larvae (cystacanths) in haemocoel of arthropods (intermediate hosts); paratenic hosts (vertebrates) occur occasionally
- behavioural changes of intermediate hosts induced by acanthocephalan larvae (cystacanths) increase their vulnerability to predation and thus foster transmission rates to the definitive host (Sures 2014)
- body divided into a trunk (metasoma) and anterior tip with proboscis armed with hooks (prosoma)

Most of the inner organs of acanthocephalans are located within the trunk. Acanthocephalans are dioecious. The ovaries (ovarian balls) of female worms float in the body cavity. Following fertilisation of mature eggs the ovary degenerates and the body cavity is filled with developing eggs. As soon as eggs contain fully developed first stage larvae (acanthors) they are released by the female through an apparatus called the uterine bell.

In addition to the testes, male worms have one to eight cement glands whose secretions enable a male to plug the vagina of a female after fertilisation. Acanthocephalans lack an intestinal tract and take up all nutrients through their body wall. In addition to nutrients, acanthocephalans take up and accumulate toxic substances such as metals, which makes them excellent indicators of environmental pollution (Sures *et al.* 2017).

Higher-level classification (families, orders and classes – see Amin 2013) is based on the amount of cement glands, the shape of the eggs, presence/absence of subtegumental giant nuclei and spines on the trunk, size and number of proboscis hooks, structure of the excretory system, etc.

At the genus and species-level, the key morphological structure for identification is the proboscis (shape and size of the proboscis, the number of files of proboscis hooks, the number of hooks in individual files, size, shape and type of proboscis hooks, etc.). Other morphological characteristics used for identification include the size and structure of the egg containing the fully developed acanthor, the position of the cephalic ganglion, the morphology of the reproductive system, *e.g.*, the shape and supination of the penis (male copulatory organ) and the vulva (in females), the number of giant hypodermal nuclei, the length of lemnisci, the size of males and females, etc.

Fig. 4.7.1. Life cycle of acanthocephalans. *Acanthocephalus lucii* (Müller, 1776); isopods serve as intermediate hosts. (Illustration by M. Luo.)

Key to the classes and orders of acanthocephalans in African fishes

1 (2) Lemnisci, cement gland and hypodermal nuclei fragmented; ligament sacs in females single, not persistent; proboscis receptacle double walled. Parasites of fishes, amphibians, reptiles, birds, and mammals (class **Palaeacanthocephala**)…………………………………………..5

2 (1) Lemnisci, cement gland and/or hypodermal nuclei not fragmented, usually giant; ligament sacs in females double, persistent; proboscis receptacle single-walled, complex or absent……………………………………………3

3 (4) Trunk spined; proboscis claviform with numerous longitudinal rows of hooks; cement glands separate, elongate pyriform to tubular; eggs with acanthor oval with radial sculpturings at right angles to surface. Parasites of fishes and Crocodilia (class **Polyacanthocephala**) [Fig. 4.7.2D] ………………………………………………order **Polyacanthorhynchida**

4 (3) Trunk may be spined; proboscis usually small with few radially arranged hooks; cement gland single, syncytial, additional distinct cement reservoir; eggs with acanthor variably shaped but not like above. Parasites of fishes and occasionally amphibians and reptiles (class **Eoacanthocephala**)…..7

5 (6) Parasites of fishes and amphibians [Figs 4.7.2A–C]…………………………………………………………………………………...order **Echinorhynchida**

6 (5) Trunk spinose. Parasites of reptiles (rare), birds and mammals; larvae in fish………………………………………order **Polymorphida** [Fig. 4.7.2E]

7 (8) Trunk entirely or only anteriorly spined. Parasites of freshwater and marine fishes [Fig. 4.7.3A,C]……………………………order **Gyracanthocephala**

8 (9) Trunk unarmed [Fig. 4.7.3B,D,E]……………..order **Neoechinorhynchida**

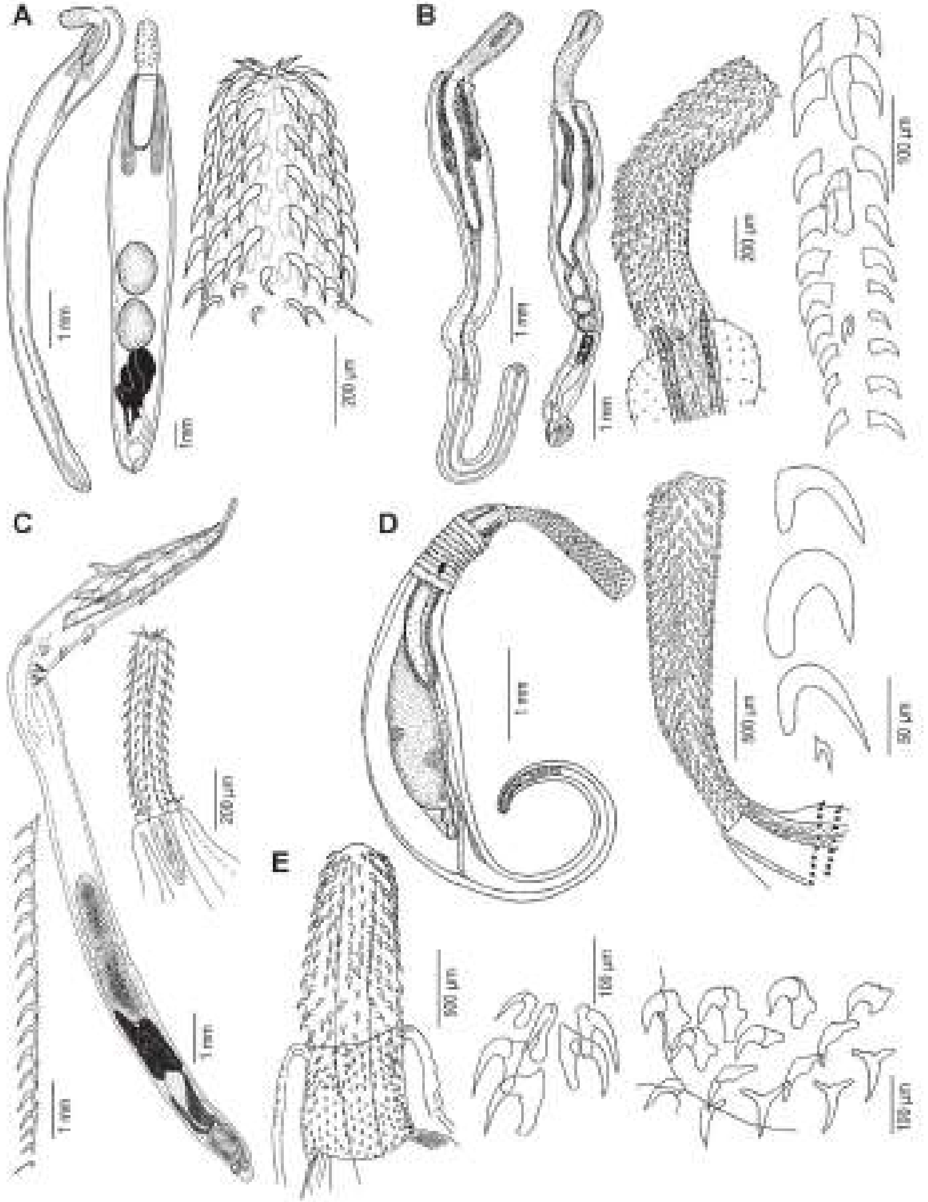

Fig. 4.7.2. Acanthocephala. **A.** *Acanthocephalus lucii* (Müller, 1776) from *Barbus* sp; **B.** *Paragorgorhynchus albertianus* Golvan, 1957 from *Alestes dentex*; **C.** *Megistacantha horridum* (Lühe, 1912) from *Hyperopisus bebe*; **D.** *Polyacanthorhynchus kenyensis* Schmidt et Canaris, 1967 from *Tilapia* sp.; **E.** *Arhythmorhynchus siluricola* Dollfus, 1929 from *Gephyroglanis* sp. (Modified from Dollfus 1929; Petrochenko 1956; Yamaguti 1963; Schmidt & Canaris 1967; Kvach *et al.* 2016.)

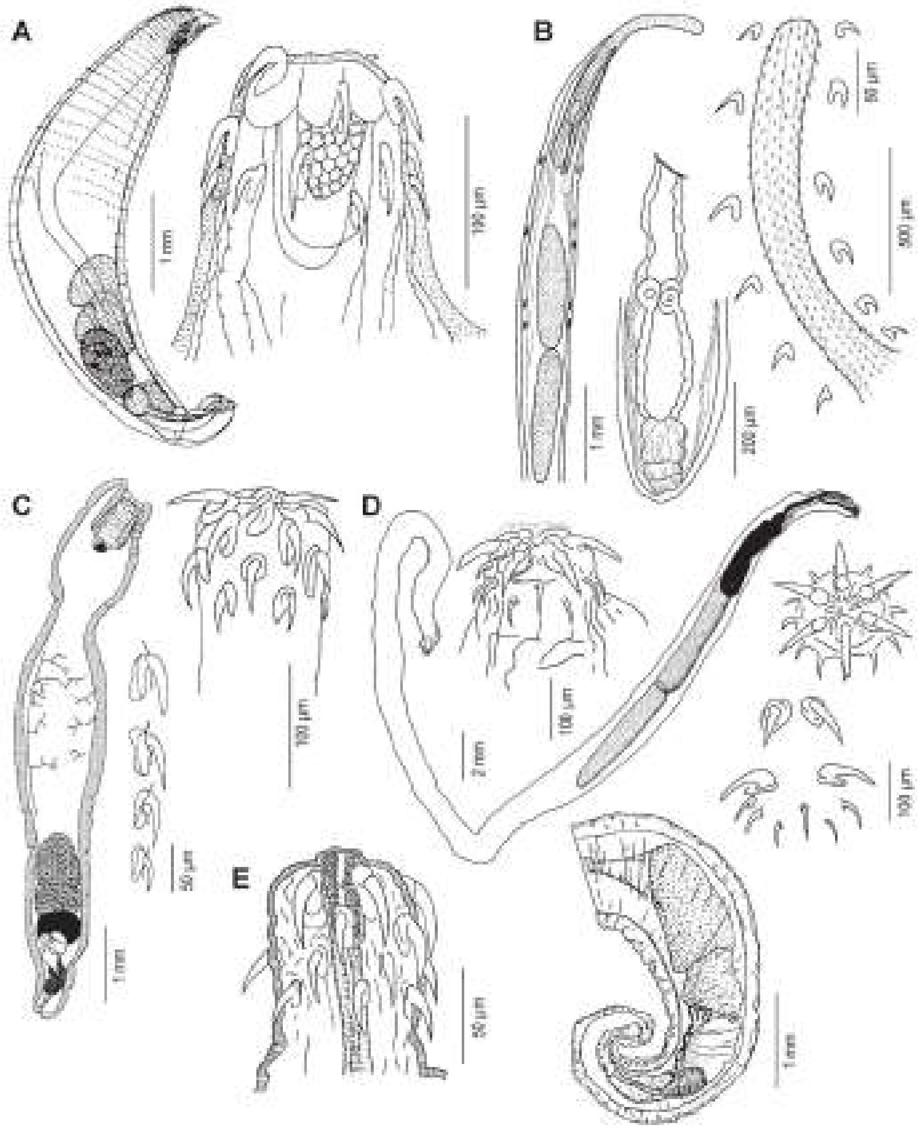

Fig. 4.7.3. Acanthocephala. **A.** *Acanthogyrus malawiensis* Amin et Hendrix, 1999 from *Labeo cylindricus*; **B.** *Tenuisentis niloticus* (Meyer, 1932) from *Heterotis niloticus*; **C.** *Pararaosentis golvani* (Troncy et Vassiliadès, 1973) from *Synodontis batensoda*; **D.** *Neoechinorhynchus africanus* Troncy, 1970 from *Citharinops distichodoides*; **E.** *Hexaspiron nigericum* Dollfus and Golvan, 1956 from *Synodontis membranaceus*. (Modified from Meyer 1932; Dollfus & Golvan 1956; Yamaguti 1963; Troncy 1970; Troncy & Vassiliadès 1973; Amin & Hendrix 1999.)

List of adult spiny-headed worms (Acanthocephala) from African freshwater fishes

Species are listed alphabetically according to individual families; the system of Amin (2013) is followed. No monograph on the Acanthocephala with keys to the families and genera has been published since Petrochenko (1956). Type species and type host species are highlighted in bold. Country of the type locality is given.

Echinorhynchida Petrochenko, 1956

Key to the genera of the Echinorhynchida from African freshwater fishes

1 (2) Trunk unarmed, cylindrical to fusiform; proboscis cylindrical, claviform or spherical, terminal; transition between true proboscis hooks and basal spiniform hooks gradual; neck and proboscis receptacle short; cement glands 6 or 8, usually pyriform to spherical and compact; eggs fusiform or elliptical (family Echinorhynchidae) [Fig. 4.7.2A]……… ***Acanthocephalus***

2 (1) Trunk armed with tegument spines; proboscis subcylindrical; proboscis receptacle long; brain ganglion anterior; cement glands 4-6, elongate and tubular or short and pyriform (family Rhadinorhynchidae)…………………3

3 (4) Middle-sized worms; trunk armed with tegument spines anteriorly; spines more numerous on the ventral side [Fig. 4.7.2B]…………………………………………..***Paragorgorhynchus***

4 (3) Body large, 1.5-2 cm in length; trunk covered with giant spines in anterior part. Parasites of mormyrid fish [Fig. 4.7.2C]………………***Megistacantha***

List of the Echinorhynchida from African freshwater fishes

Echinorhynchidae Cobbold, 1876

Acanthocephalus Koelreuther, 1771

Acanthocephalus lucii (Müller, 1776) from *Oreochromis niloticus* [Fig. 4.7.2A]

Rhadinorhynchidae Travassos, 1923

Megistacantha Golvan, 1960

Megistacantha horridum (Lühe, 1912) from *Gnathonemus petersii*, *Hyperopisus bebe*, *Hippopotamyrus pictus*, ***Marcusenius cyprinoides*** (Egypt) [Fig. 4.7.2C]

Megistacantha sanghaensis Kvach, Jirků et Scholz, 2016 from ***Mormyrops anguilloides*** (Central African Republic)

Paragorgorhynchus Golvan 1957

Paragorgorhynchus albertianus Golvan, 1957 from *Alestes dentex*, *Bagrus bajad*, *Hydrocynus forskahlii*, ***Lates niloticus*** (Congo), *Schilbe mystus* [Fig. 4.7.2B]

Paragorgorhynchus aswanensis Saoud et Wanas, 1990 from *Bagrus bajad*, *B. docmak*, *Clarias gariepinus*, ***Lates niloticus*** (Egypt), *Tetraodon lineatus*

Paragorgorhynchus chariensis Troncy, 1970 from ***Lates niloticus*** (Chad)

Paragorgorhynchus sp. from *Clarias gariepinus*

Polyacanthorhynchida Amin, 1987

List of the Polyacanthorhynchida from African freshwater fishes

Polyacanthorhynchidae Golvan, 1926

Polyacanthorhynchus Travassos, 1920

Polyacanthorhynchus kenyensis Schmidt et Canaris, 1967 (juvenile) from *Coptodon zillii*, *Enteromius paludinosus*, *Micropterus salmoides*, *Oreochromis leucostictus*, *O. niloticus*, ***Tilapia* sp.** (Kenya) [Fig. 4.7.2D]

Polymorphida Petrochenko, 1956

List of the Polymorphida from African freshwater fishes

Polymorphidae Meyer, 1931

Arhythmorhynchus Lühe, 1911

Arhythmorhynchus siluricola Dollfus, 1929 (juvenile) from *Chrysichtys* sp., ***Gephyroglanis* sp.** (Cameroon) [Fig. 4.7.2E]

Gyracanthocephala Van Cleave, 1936

Key to the genera of the Gyracanthocephala from African freshwater fishes

1 (2) Trunk armed only anteriorly with circles of spines; circles usually incomplete dorsally (subfamily Quadrigyrinae) [Fig. 4.7.3A]……………………… ……………………………………………………………….. ***Acanthogyrus***

2 (1) Trunk armed anteriorly with complete circles of spines in one or two regions separated by an unarmed zone; spines in the second region may extend over the rest of the trunk in circles or in longitudinal rows (subfamily Pallisentinae) [Fig. 4.7.3C]…………………………………… ***Pararaosentis***

List of the Gyracanthocephala from African freshwater fishes

Quadrigyridae Van Cleave, 1920

Acanthogyrus Thapar, 1927

Acanthogyrus malawiensis Amin et Hendrix, 1999 from ***Labeo cylindricus*** (Malawi) [Fig. 4.7.3A]

Acanthogyrus maroccanus (Dollfus, 1951) from ***Luciobarbus setivimensis*** (Morocco)

Acanthogyrus nigeriensis (Dollfus et Golvan, 1956) from ***Labeo coubie*** (Mali)

Acanthogyrus phillipi (Mashego, 1988) from ***Enteromius neefi*** (South Africa)

Acanthogyrus tilapiae (Baylis, 1948) from *Aristochromis christyi*, *Bagrus meridionalis*, *Coptodon guineensis*, *C. rendalli*, *C. zillii*, *Ctenopharynx pictus*, *Genyochromis mento*, *Haplochromis squamipinnis*, *Haplochromis* sp., *Hemichromis bimaculatus*, *H. fasciatus*, *Labeotropheus fuelleborni*, *Lichnochromis acuticeps*, *Maylandia emmiltos*, *M. zebra*, *Mchenga thinos*, *Melanochromis auratus*, *M. heterochromis*, *M. vermivorus*, *Nimbochromis polystigma*, *Oreochromis andersonii*, *O. esculentus*, *O. leucostictus*, ***O. lidole*** (Tanzania), *O. macrochir*, *O. niloticus*, *O. tanganicae*, *Oreochromis* sp., *Petrotilapia genalutea*, *Placidochromis johnstoni*, *Protomelas annectens*, *P. taeniolatus*, *Pseudotropheus elongatus*, *Sarotherodon galilaeus*, *S. melanotheron*, *Stigmatochromis woodi*, *Tetraodon lineatus*, *Trematocranus placodon*, *Tropheops microstoma*, *T. tropheops*, *Tyrannochromis macrostoma*

Pararaosentis Amin, Heckmann, Ha, Luc et Doanh, 2000

Pararaosentis golvani (Troncy et Vassiliadès, 1973) from *Schilbe mystus*, ***Synodontis batensoda*** (Senegal), *S. frontosus*, *S. membranaceus*, *Tetraodon lineatus* [Fig. 4.7.3C]

Neoechinorhynchida Ward, 1917

Keys to the genera of the Neoechinorhynchida from African freshwater fishes

1 (2) Trunk without dendritic nuclei; proboscis short and subglobular or subcylindrical, armed with a small number of hooks arranged in spiral, circular, or diagonal rows (family Neoechinorhynchidae)……………………………3

2 (1) Proboscis relatively long and cylindrical, armed with many hooks arranged quincuncially, in longitudinal rows. Parasites of fishes (family Tenuisentidae) [Fig. 4.7.3B]……………………………………………….***Tenuisentis***

3 (4) Six hooks in each of three circles of hooks on the proboscis [Fig. 4.7.3D] ………………………………………………………..***Neoechinorhynchus***

4 (3) Proboscis armed with 6 hooks per circle; anterior trunk with 7 circles of tiny spines [Fig. 4.7.3E]……………………………………………..***Hexaspiron***

List of the Neoechinorhynchida from African freshwater fishes

Tenuisentidae Van Cleave, 1936

Tenuisentis Van Cleave, 1936

Tenuisentis niloticus (Meyer, 1932) from ***Heterotis niloticus*** (Egypt), *Hydrocynus brevis*, *Lates niloticus* [Fig. 4.7.3B]

Neoechinorhynchidae Van Cleave, 1919

Hexaspiron Dollfus et Golvan, 1956

Hexaspiron nigericum Dollfus et Golvan, 1956 from ***Synodontis membranaceus*** (Nigeria) [Fig. 4.7.3E]

Neoechinorhynchus Stiles et Hassall, 1905

Neoechinorhynchus africanus Troncy, 1970 from ***Citharinops distichodoides*** (Chad), *Citharinus citharus*, *Hydrocynus forskahlii*, *Synodontis membranaceus* [Fig. 4.7.3D]

Neoechinorhynchus ichthyobori Saoud, El-Naffar et Abu-Sinna, 1974 from *Chelon ramada*, ***Ichthyborus besse*** (Sudan)

Neoechinorhynchus rutili (Müller, 1780) from *Clarias gariepinus*

Neoechinorhynchus sp. from *Citharinus citharus*, *Clarias gariepinus*

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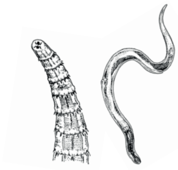

Chapter 4.8.

NEMATODA

Šárka MAŠOVÁ & Roman KUCHTA

Nematodes or roundworms (Nematoda) – basic characteristics, life cycles, classification and principal diagnostic features

- roundworms (Ecdysozoa: Nematoda)
- nearly 25,000 species described
- only some groups parasitic in vertebrates
- endoparasites of different organs and tissues
- mostly long narrow cylindrical body, round in cross section, tapered towards both ends
- pseudocoelom
- body surface covered with cuticle
- well-developed digestive tract
- gonochoristic (separate sexes)
- direct or indirect life cycles (Fig. 4.8.1) (monoxeny or heteroxeny)
- four changes of cuticle (moulting)
- causative agents of human fish-borne diseases (*e.g.*, anisakiasis, gnathostomiasis)

Generally, life cycles differ depending upon the species of nematode (Yanong 2002). Development of almost all species of fish nematodes requires an intermediate host with presumable exceptions in some groups such as Capillariidae (see Moravec 2013). A complete nematode life cycle consists of four larval stages (L), separated from one another by a moult (or ecdysis) followed by an immature adult (or subadult): egg → L1 → L2 → L3 → L4 → adult. Larval stages of monoxenous species (with direct life cycle) undergo two transformations after hatching (*e.g.*, Pharyngodonidae). Infective juveniles (L3) may be ingested and mature in the intestine or infect via penetrating the skin and migrate through tissues.

Heteroxenous nematodes (with indirect life cycle) involve one or more intermediate hosts (*e.g.*, *Camallanus* sp. – see Fig. 4.8.1). The first two moults usually occur in the intermediate host. Infection of the definitive host by the L3 is either via ingestion of the intermediate host or inoculation by the intermediate host (Gaugler & Bilgrami 2004).

The classification of nematodes presented here is based on a combination of Moravec (2013), Hodda (2011) and the traditional consensus of nematode relationships according to Blaxter et al. (1998). This presentation includes only parasitic nematode families reported from Africa. Higher-level classification (classes, orders and families) of nematodes is based mainly on the type and morphology of the oesophagus, the anterior part of the digestive tract, the structure of the anterior end (type of oral opening, lips, interlabia, teeth, buccal cavity, cuticular structures), presence/absence of phasmids, presence and number of caudal papillae, presence/absence of stichosome or trophosome, presence/absence of lateral canals in the excretory system and structure of its terminal duct, type of eggs, morphology of the reproductive system (type of uterus, presence/absence of caudal bursa in males), presence/absence of the precloacal sucker, position of the vulva, etc. (Fig. 4.8.2; see Anderson et al. 2009; Gibbons 2010 for keys up to the genus level).

Generic classification and species identification are based on the details of the anterior end (*e.g.*, deirids, structure of the buccal capsule), the morphology of the male copulatory organs (*e.g.*, spicules, gubernaculum, copulatory bursa), detailed structure of the digestive system (*e.g.*, ventriculus, intestinal caecum, oesophagus, pharynx). Other morphological characteristics used for identification include body size and the proportional size of individual parts (*e.g.*, oesophagus, caeca, tail, position of the vulva) to body length; the position of the excretory pore and the nerve ring, structure of the cuticle; the structures or projections of the eggs and the number and position of caudal papillae. For identification, the infection site and host taxon are also very often important.

Fig. 4.8.1. Heteroxenous (indirect) life cycle of *Camallanus* sp.; copepods serve as intermediate hosts. (Illustration by M. Luo.)

Glossary of taxonomically important morphological characteristics of nematodes

ala (plural alae): thin (flat) cuticular protrusion or fin, running longitudinally, usually lateral or sublateral, frequently paired; cervical alae (see below) on the anterior end; caudal alae on the posterior end of males

amphid: complex sensilla at the anterior body end with chemoreceptory function; its primitive position is lateral and postlabial; opening to the exterior usually as a simple pore [Fig. 4.8.2A-C]

bacillary bands: modification of hypodermis, consisting of longitudinal rows of columnar cells that have pore-like openings to the surface of cuticle (in Trichuroidea) (Anderson *et al.* 2009)

boss (plural bosses): any protruberant part, prominence or swelling of cuticle (*e.g.*, *Galeiceps*, *Philometroides*, *Nilonema*)

bulb or bulbus: bulbous inflation usually in the posterior part of the oesophagus in certain nematodes (*e.g.*, Pharyngodonidae), forces the food into the intestine [Fig. 4.8.2E]

cervical alae: in some parasitic nematodes, wide lateral anterior alae (single, bifid or trifid, often with internal supporting struts)

cheilostome: anterior region of the mouth (stoma), which is lined by external cuticle formation and is not surrounded by oesophageal tissues

collar: any of various structures comparable with a collar (*e.g.*, *Galeiceps*)

collarette: usually anterior cuticular extensions forming an annular ring in the neck region (*e.g.*, Physalopteroidea or *Pseudoproleptus*)

copulatory bursa: copulatory accessorial organ, formed by greatly expanded caudal alae in male tail end of certain parasitic nematodes

corpus: anterior part of oesophagus, acts as a suction pump

deirids (cervical papillae): peripheral somatic sense organs in the anterior part of the body, very often near nerve ring, considered to act as a mechanoreceptor [Fig. 4.8.2N-P]

didelphic: having two uteri

gubernaculum: sclerotised dorsal and lateral walls of the distal cloaca form the gubernaculum that guides the spicules when protracting or retracting and protects the underlying tissue [Fig. 4.8.2R-T]

interlabium: small lobe situated between the lips in certain nematodes [Fig. 4.8.2C]

intestinal caecum: appendage of the intestine extending anteriorly to the oesophagus [Fig. 4.8.2G-J]

isthmus: constriction of the oesophagus, region between corpus and bulb [Fig. 4.8.2E]

labium: lip on the cephalic region [Fig. 4.8.2C]

monodelphic: having one uterus

mucron: a small pointed projection, or spine-like ending on a terminus (*e.g.*, on tail tip of *Galeiceps*)

oesophagus (pharynx): part of the digestive tract that starts behind the buccal cavity or oral opening [Fig. 4.8.2D-J]

oesophastome: bulbous pseudobuccal capsule formed by an expansion of the oesophagus at its anterior end [Fig. 4.8.2D]

phasmids: peripheral somatic sense organs usually on the tail, probably with a chemoreceptory function [Fig. 4.8.2Q,U]

platymyarian: having fibres of the muscle cells adjacent and perpendicular to the hypodermis

spicule: sclerotised male copulatory organ of various shapes, usually paired, located immediately dorsal to the cloaca

stichocyte: glandular unicellular cell forming the stichosome [Fig. 4.8.2M]

stichosome: series of protein synthesising gland cells arranged in a row along the posterior portion of the oesophagus [Fig. 4.8.2L]

trophosome: structure which probably represents a modified intestine serving as a nutrient storage area of certain parasitic nematodes arranged along the posterior portion of the oesophagus

ventricular appendix: appendage of the ventriculus extending posteriorly to the intestine [Fig. 4.8.2H-K]

ventriculus: short region at the anterior end of the intestine [Fig. 4.8.2G-K]

Classification of nematodes infecting African fishes

A single asterisk (*) before a nematode's scientific name denotes that the taxon has been recorded only in its larval stage in the given fish host(s). A double asterisk (**) denotes records of both larvae and adults in the given host(s), whilst an unmarked taxon denotes records of only adults in the given host(s).

Class Adenophorea von Linstow, 1905
Subclass Enoplia Pearse, 1942
- Order Enoplida Filipjev, 1929
  - Suborder Enoplina Chitwood et Chitwood, 1937
- Order Trichocephalida Spasski, 1954
  - Suborder Trichinellina Hodda, 2007
  - Superfamily Trichinelloidea Ward, 1907
    - Family Capillariidae Railliet, 1915
    - Family Cystoopsidae Skrjabin, 1923
  - Suborder Dioctophymatina Skrjabin, 1927
  - Superfamily *Dioctophymatoidea Castellani et Chalmers, 1910
    - Family *Dioctophymatidae Castellani et Chalmers, 1910

Class Secernentea von Linstow, 1905
- Order Oxyurida Skrjabin, 1923
  - Suborder Oxyurina Railliet, 1916
  - Superfamily Oxyuroidea Cobbold, 1864
    - Family Pharyngodonidae Travassos, 1920
- Order Ascaridida Skrjabin et Schulz, 1940
  - Superfamily Cosmocercoidea Railliet, 1916
    - Family Cosmocercidae Railliet, 1916
    - Family Kathlaniidae Lane, 1914
    - Family Atractidae Railliet, 1917
  - Superfamily Seuratoidea Hall, 1916
    - Family Quimperiidae Gendre, 1928
    - Family Cucullanidae Cobbold, 1864
  - Superfamily Ascaridoidea Baird, 1853
    - Family **Anisakidae Railliet et Henry, 1912
    - Family *Ascarididae Baird, 1853
    - Family Heterocheilidae Railliet et Henry, 1915
- Order Spirurida Chitwood, 1933
  - Suborder Camallanina Chitwood, 1937
  - Superfamily Camallanoidea Railliet et Henry, 1915
    - Family **Camallanidae Railliet et Henry, 1915
  - Superfamily Dracunculoidea Cameron, 1934
    - Family Philometridae Baylis et Daubney, 1926
    - Family Daniconematidae Moravec et Køie, 1987
  - Suborder Spirurina Chitwood, 1933
  - Superfamily **Anguillicoloidea Yamaguti, 1935
    - Family **Anguillicolidae Yamaguti, 1935

Superfamily *Gnathostomatoidea Railliet, 1895
Family *Gnathostomatidae Railliet, 1895
Superfamily Physalopteroidea Railliet, 1893
Family Physalopteridae Railliet, 1893
Superfamily Thelazioidea Skryabin, 1915
Family ** Rhabdochonidae Travassos, Artigas et Pereira, 1928
Superfamily Habronematoidea Chitwood et Wehr, 1932
Family Cystidicolidae Skryabin, 1946
Superfamily Acuarioidea Railliet, Henry et Sissoff, 1912
Family **Acuariidae Railliet, Henry et Sissoff, 1912

Identification keys and a systematic survey of nematodes (Nematoda) from African freshwater fish

The keys presented below are designed according to Moravec (2006, 2013), Thatcher (2006), Anderson *et al.* (2009), and Arai & Smith (2016) to allow identification of larval and/or adult nematodes up to the genus level. Species are listed alphabetically within the respective higher-order taxa. The type species of each genus and the type host of each species are highlighted in bold. The country where the type locality lies is given if known. Host names follow Froese & Pauly (2017).

Key to the classes of the Nematoda *sensu* Blaxter *et al.* (1998)

1 (2) Amphids always post-labial. Phasmids absent. Caudal papillae absent or few in number. Oesophagus cylindrical or with oesophageal glands free in pseudocoel and forming stichosome or trophosome. Excretory system with out lateral canals and terminal duct not lined with cuticle. Males with one spicule or spicule absent. Eggs usually unsegmented with plug at either pole or hatching *in utero*. First-larval stage often with stylet and usually infective to final host..…………**Adenophorea** (**subclass Enoplia**)

2 (1) Amphid apertures on lips, often difficult to see. Phasmids present. Oesophagus never in form of stichosome. Excretory system with lateral canals and terminal canal lined with cuticle. Caudal papillae almost always numerous in males. Spicules two, exceptionally spicules absent. Eggs without polar plugs, rarely operculate at one or both poles, or hatching *in utero*. Early third larval stage infective to the final host………………**Secernentea**

ENOPLIA Pearse, 1942

Key to the superfamilies of the Enoplia from African freshwater fishes

1 (2) Well-developed oesophagus cylindrical; stichosome or trophosome absent. Male tail modified to form ventral sucker-like muscular bursa. Monodelphic. Vulva near anus. Body thick, massive………**Dioctophymatoidea**

2 (1) Stichosome present. Male tail without muscular bursa. Vulva anterior or near the end of oesophagus. Body small, thin, mostly filiform. Only in *Cystoopsis* is posterior part of body globular………………**Trichinelloidea**

TRICHINELLOIDEA Ward, 1907

Key to the families of the Trichinelloidea from African freshwater fishes

1 (2) Digestive tract incomplete, intestine dilated into a sac, anus absent. Vulva near nerve ring. Female body with thread-like anterior region and poste-

riorly expanded to form vesicles. Parasites of the skin of sturgeons and gars, in Africa known from Cichlidae…………………………**Cystoopsidae**

2 (1) Digestive tract complete including anus. Vulva near mid-region or end of oesophagus. Posterior region of female somewhat expanded but not cylindrical, not vesicle-like. Adults in the digestive tract or liver .....……………………………………………………………**Capillariidae**

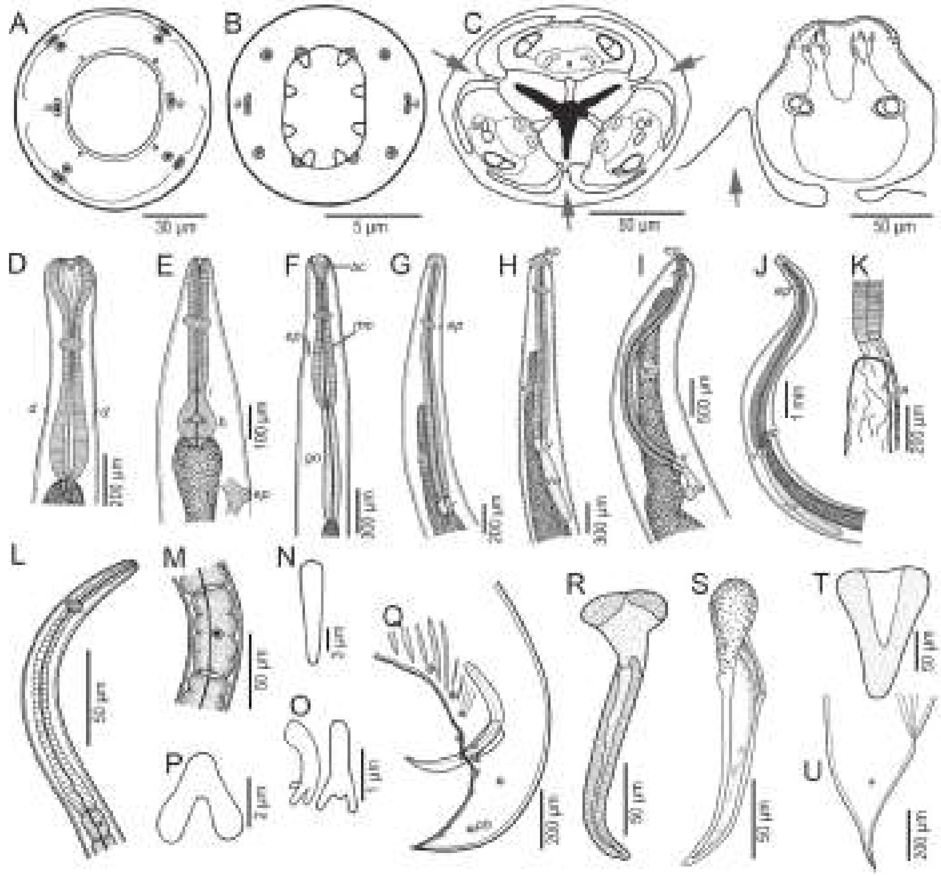

Fig. 4.8.2. Selected morphological characters important for identification. **A-C.** Amphids (*a*) of *Procamallanus daleneae* (Boomker, 1933) (A); *Rhabdochona tricuspidata* Moravec et Jirků, 2014 (B); *Multicaecum heterotis* Petter, Vassiliadès et Marchand, 1979 (C); **D.** Muscular oesophagus with oesophastome and deirids (*d*) of *Cucullanus mormyri* Moravec et Scholz, 2017; **E.** Cylindrical oesophagus of *Cithariniella khalili* Petter, Vassiliadès et Troncy, 1972, ending in a globular bulb (*b*) with valvular apparatus and separated from the corpus by a constriction (isthmus – *i*); **F.** Buccal capsule (*bc*) and oesophagus of *Procamallanus daleneae* (Boomker, 1933) divided into muscular (*mo*) and glandular (*go*) parts; **G-J.** Intestinal caecum (*ic*) of *Dujardinascaris mormyropsis* Moravec et Jirků, 2014 (G); third-stage larva of *Galeiceps* sp. with ventricular appendix (*va*) situated below ventral cephalic tooth (H); third-stage larva of *Contracaecum* sp. (I); *Hysterothylacium anguillae* Moravec, Taraschewski, Appelhoff et Weyl 2012 (J); **K.** Ventriculus with ventricular appendix of *Raphidascaroides bishaii* Khalil, 1961; **L, M.** Capillariidae gen. sp., muscular oesophagus (L); stichocyte in the middle of the stichosome (M); **N-P.** Deirids of *Rhabdochona* spp.; **Q.** Two equal spicules, gubernaculum, eleven pairs of caudal papillae (posterior lateral pair represents phasmids – *ph*) and one additional unpaired median papilla on the anterior cloacal lip of *Falcaustra similis* Moravec et Van As, 2004; **R-T.** Gubernaculum of *Dujardinascaris mormyropsis* Moravec et Jirků, 2014 (R); *Multicaecum heterotis* Petter, Vassiliadès et Marchand, 1979 (S); *Falcaustra similis* Moravec et Van As, 2004 (T); **U.** Phasmid of *Falcaustra piscicola* (von Linstow, 1907). (Modified from Moravec *et al*. 1999, 2012; Moravec & Van As 2004, 2015; Mašová *et al.* 2010; Moravec & Jirků 2014a,b, 2015, 2017; Moravec & Scholz 2017.) ex - excretory pore

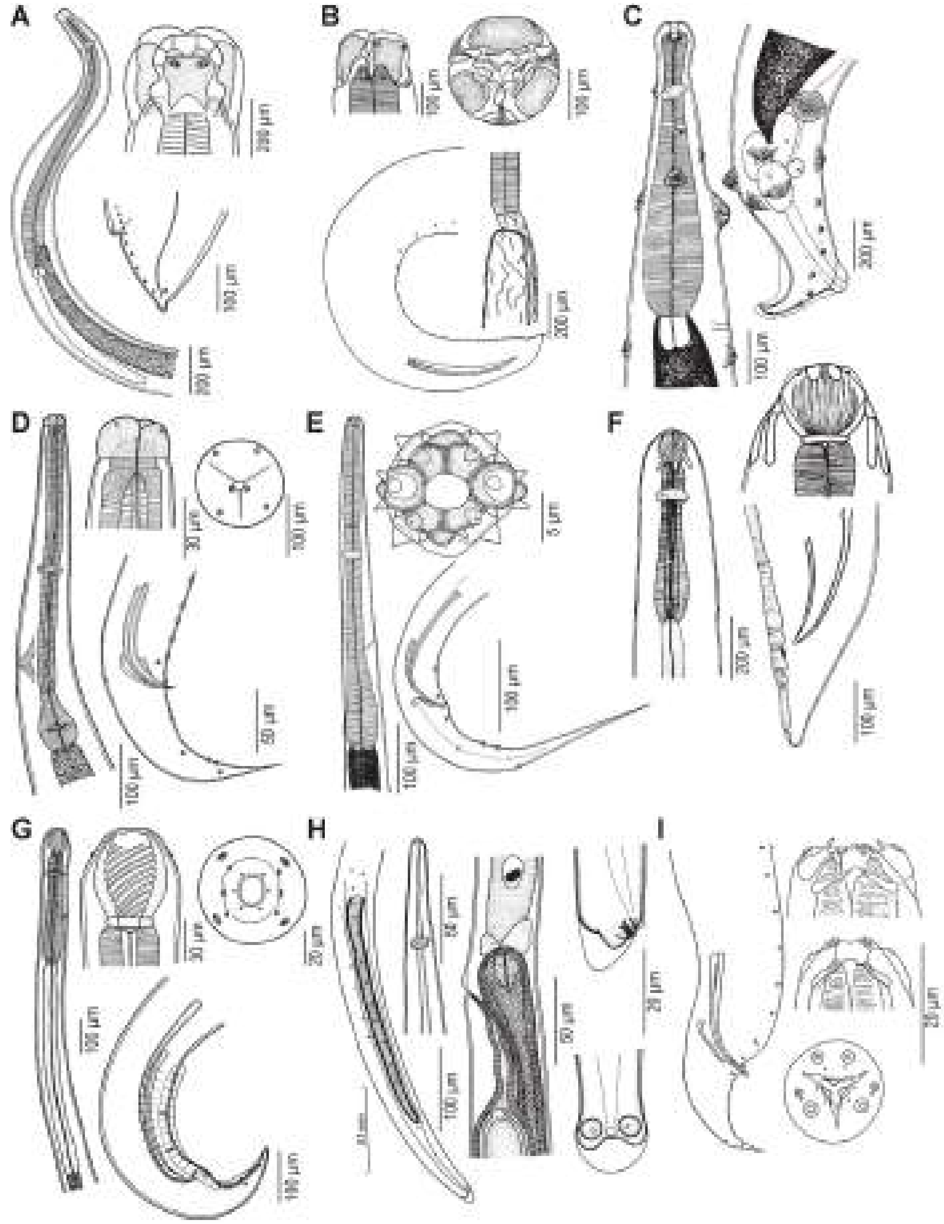

Fig. 4.8.3. Nematoda. **A.** *Hysterothylacium anguillae* Moravec, Taraschewski, Appelhoff et Weyl, 2012 from *Anguilla marmorata*; **B.** *Raphidascaroides bishaii* Khalil, 1961 from *Gymnarchus niloticus*; **C.** *Anguillicoloides papernai* (Moravec et Taraschewski, 1988) from *Anguilla mossambica*; **D.** *Labeonema africanum* Moravec et Van As, 2004 from *Synodontis nigromaculatus*; **E.** *Orientatractis brycini* González-Solís et Mariaux, 2017 from *Brycinus macrolepidotus*; **F.** *Camallanus longicaudatus* Moravec, 1973 from *Labeo horie*; **G.** *Procamallanus* (*Spirocamallanus*) *spiralis* Baylis, 1923 from *Clarias theodorae*; **H.** *Capillostrongyloides fritschi* (Travassos, 1914) from *Bagrus docmak*; **I.** *Aplectana chamaeleonis* (Baylis, 1929) from *Oreochromis niloticus.* (Modified from Khalil 1961; Chen 1966; Moravec 1973, 2001; Moravec & Taraschewski 1988; Moravec & Van As 2004, 2015; Moravec *et al*. 2012; González-Solís & Mariaux 2017.)

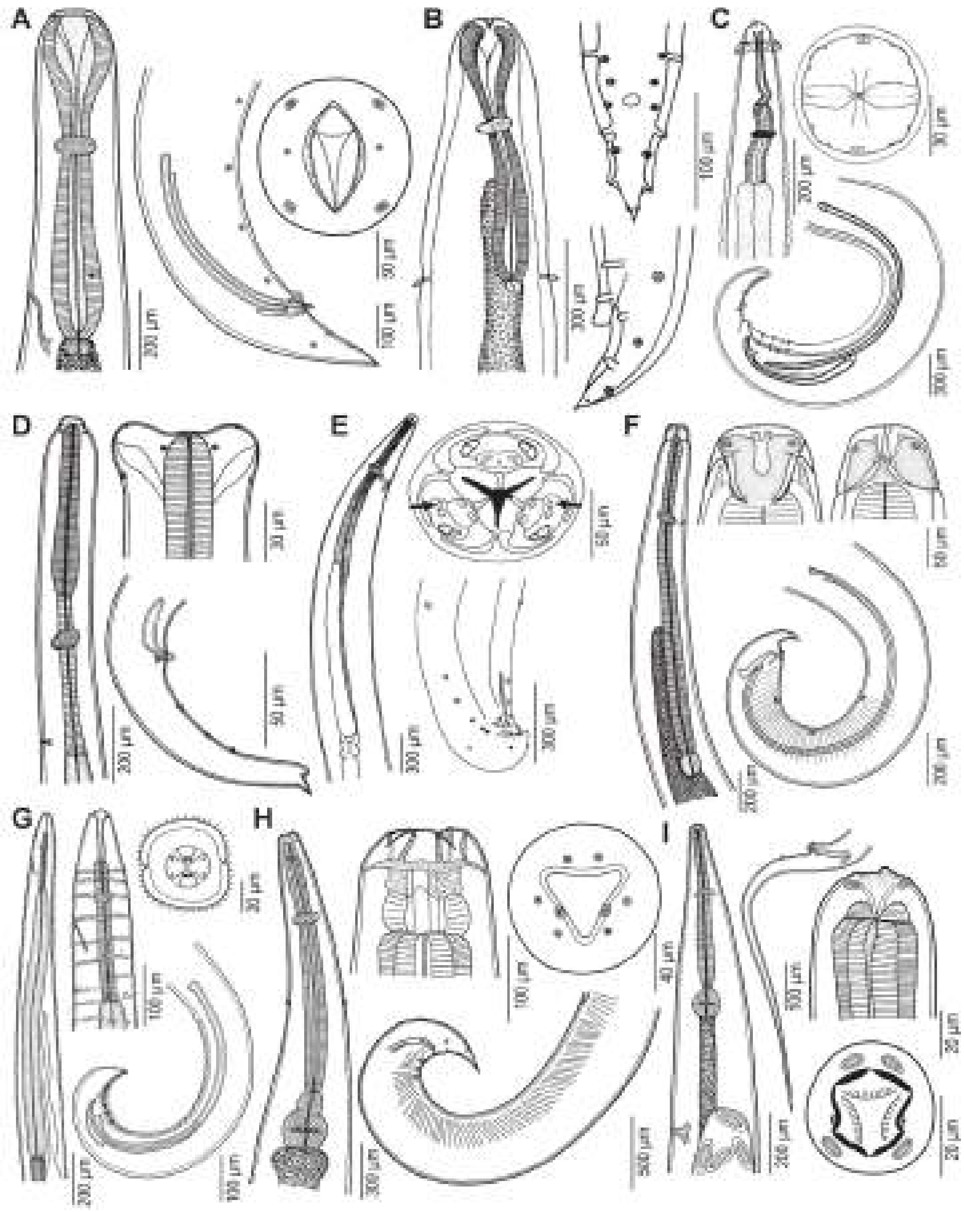

Fig. 4.8.4. Nematoda. **A.** *Cucullanus baylisi* Campana-Rouget, 1961 from *Synodontis schall*; **B.** *Dichelyne fossor* Jägerskiöld, 1902 from *Lates niloticus*; **C.** *Pseudoproleptus africanus* Khalil, 1973 from *Mormyrus* sp.; **D.** *Mexiconema africanum* Moravec, Jirků, Charo-Karisa et Mašová, 2009 from *Auchenoglanis occidentalis*; **E.** *Multicaecum heterotis* Petter, Vassiliadès et Marchand, 1979 from *Heterotis niloticus*; **F.** *Dujardinascaris mormyropsis* Moravec et Jirků, 2014 from *Mormyrops anguilloides*; **G.** *Spinitectus polli* Campana-Rouget, 1961 from *Synodontis decorus*; **H.** *Falcaustra similis* Moravec et Van As, 2004 from *Synodontis nigromaculatus*; **I.** *Cithariniella longicaudata* Moravec et Van As, 2015 from *Schilbe intermedius.* (Modified from Moravec 1974; Moravec & Van As 2004; Moravec *et al.* 2009a; Mašová *et al.* 2010; Moravec & Jirků 2014a, 2017; Moravec & Scholz 2017.)

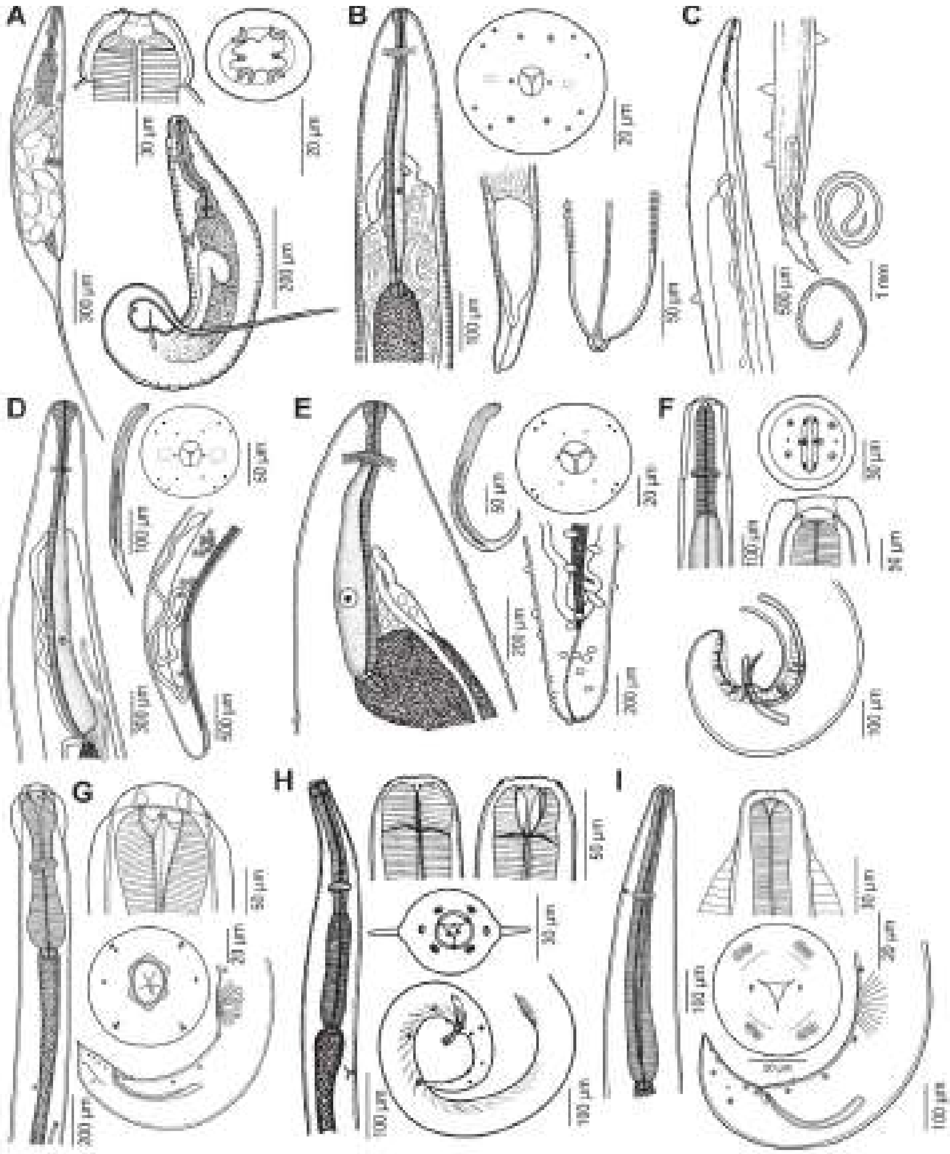

Fig. 4.8.5. Nematoda. **A.** *Synodontisia okavangoensis* Moravec et Van As, 2004 from *Synodontis nigromaculatus*; **B.** *Afrophilometra hydrocyoni* (Fahmy, Mansour et El-Naffar, 1976) from *Hydrocynus forskahlii*; **C.** *Nilonema gymnarchi* Khalil, 1960 from *Gymnarchus niloticus*; **D.** *Philometra lati* Moravec, Charo-Karisa et Jirků, 2009 from *Lates niloticus*; **E.** *Philometroides khalili* Moravec, Halajian, Tavakol, Nyagura et Luus-Powell, 2015 from *Labeo rosae*; **F.** *Heliconema africanum* (von Linstow, 1899) from *Anguilla mossambica*; **G.** *Gendria sanghaensis* Moravec et Jirků, 2017 from *Schilbe marmoratus*; **H.** *Paraquimperia africana* Moravec, Boomker et Taraschewski, 2000 from *Anguilla mossambica*; **I.** *Quimperia lanceolata* Gendre, 1926 from *Ctenopoma kingsleyae.* (Modified from Khalil 1960; Moravec *et al.* 2000; Moravec & Van As 2004; Moravec *et al.* 2009b, 2013, 2015; Moravec & Jirků 2017.)

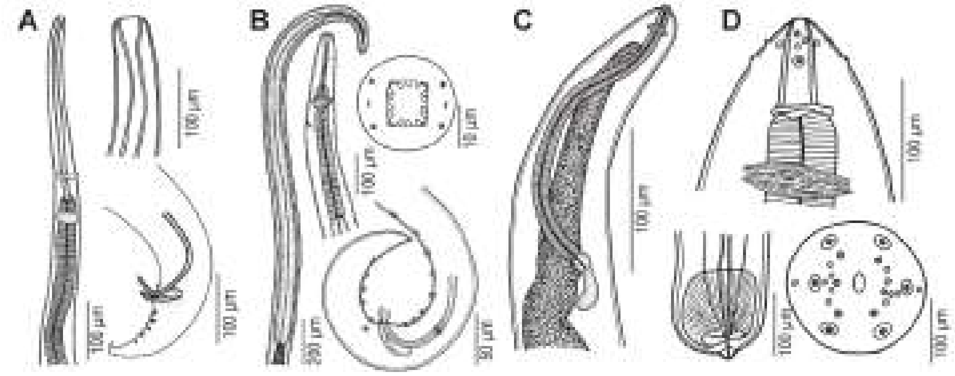

Fig. 4.8.6. Nematoda. **A.** *Prosungulonema africanum* (Moravec et Puylaert, 1970) from *Thoracochromis schwetzi*; **B.** *Rhabdochona* (*Rhabdochona*) *centroafricana* Moravec et Jirků, 2014 from *Enteromius miolepis*; **C.** *Contracaecum* sp. from *Hydrocynus vittatus*; **D.** *Eustrongylides* sp. from *Hydrocynus vittatus*. (Modified from Moravec & Puylaert 1970; Moravec & Jirků 2014b; Moravec & Van As 2015.)

List of the Enoplia from African freshwater fishes

Capillariidae Railliet, 1915

Capillaria Zeder, 1800

Capillaria sp. from *Clarias gariepinus*, *Clarotes laticeps*, *Malapterurus electricus*, *Synodontis zambezensis*

Capillostrongyloides Freitas et Lent, 1935

Capillostrongyloides fritschi (Travassos, 1914) [syns *Capillaria fritschi* Travassos, 1914; *Capillaria yamagutii* Tadros et Mahmoud, 1968] from *Bagrus bajad*, *B. docmak*, *Malapterurus electricus* [Fig. 4.8.3H]

Capillariidae gen. sp. from *Auchenoglanis* sp., *Gnathonemus petersii*

Note: representatives of *Capillaria* are not known from fishes. This very probably concerns a misidentification. For generic key to fish capillariids – see Moravec (2001).

Cystoopsidae Skryabin, 1923

Cystoopsis Wagner, 1867

Cystoopsis sp. from *Tropheus moorii* (see Moravec and Salgado-Maldonado 2003 who reported unpublished data of G.L. Hoffman)

*****Dioctophymatidae Railliet, 1915

Eustrongylides Jägerskiöld, 1909

Eustrongylides africanus (Jägerskiöld, 1909) from *Clarias anguillaris*, *C. gariepinus*

Eustrongylides sp. from *Bagrus docmak*, *Clarias camerunensis*, *C. gariepinus*, *C. theodorae*, *Clarias* sp., *Clarotes laticeps*, *Dinotopterus cunningtoni*, *Enteromius humilis*, *Haplochromis angustifrons*, *H. argenteus*, *H. eduardii*, *H. graueri*, *H. guiarti*, *H. labiatus*, *H. nubilus*, *H. pappenheimi*, *Haplochromis* sp., *Hydrocynus vittatus*, *Labeobarbus altianalis*, *L. tsanensis*, *Lepidiolamprologus cunningtoni*, *Mormyrus caschive*, *Oreochromis niloticus*, *Protopterus aethiopicus* [Fig. 4.8.6D]

Note: *Eustrongylides africanus* was designated as *species inquirenda* by Measures (1988). *Eustrongylides* sp. from *Clarias* sp. was originally misidentified as *Philometra congolensis* Schuurmans-Stekhoven, 1937 – see Moravec (2006).

SECERNENTEA von Linstow, 1905

Key to the orders of the Secernentea from African fishes

1 (2) Male with a reduced number of caudal papillae. Generally only one spicule. Body short and stout. Oesophagus with a bulb. Pre-anal sucker absent. Female with large embryonated eggs often flattened on one side. Monoxenous with two moults in egg. Parasites of colon or rectum…………………………………………**Oxyurida** (**Pharyngodonidae**)

2 (1) Nematodes lacking most of the above characters…………………………3

3 (4) Anterior extremity triradiate (except in some members of the Seuratoidea). Head end with three lips (one dorsal and two ventrolateral). Lateral, external labial papillae present. With 2-3 pairs of caudal papillae in dorsolateral position. Oesophagus variable in form but not divided into short muscular and long glandular parts. Pre-anal sucker present or absent in males. Usually found in the intestine of the final host. Larval stages preinfective for final host do not develop entirely in an intermediate host……………………………………………………………….**Ascaridida**

4 (3) Anterior extremity bilaterally symmetrical. Lateral, external labial papillae absent. Head end with two lateral lips or lips reduced or absent. Stoma usually well developed, sometimes reduced. Oesophagus divided into shorter anterior muscular part and longer posterior glandular part; division sometimes indistinct. Caudal papillae always ventral or ventrolateral in position. Pre-anal sucker not present. Parasites of anterior part of gut (oesophagus, stomach, rarely duodenum) or tissues and tissue voids. Larval stages preinfective for final host develop entirely in an intermediate host ……………………………………………………………………….. **Spirurida**

Pharyngodonidae Travassos, 1920

Key to the genera of the family Pharyngodonidae from African freshwater fishes

(for keys to the species of *Cithariniella* – see Koubková *et al.* 2010; for keys to pharyngodonid genera from fishes – see Moravec 1994)

1(2) Tail long, slender and sharply pointed. Oral aperture hexagonal. Spicule present…………………………………………………………..***Synodontisia***

2 (1) Tail long, slender and sharply pointed. Oral aperture triangular. Buccal cavity present; vulva close to anus; eggs with polar filaments.……………………………………………………………***Cithariniella***

List of the Pharyngodonidae from African freshwater fishes

Cithariniella Khalil, 1964

Cithariniella citharini Khalil, 1964 from ***Citharinus citharus*** (Sudan), *Distichodus brevipinnis*, *Synodontis schall*, *S. serratus*

Cithariniella khalili Petter, Vassiliadès et Troncy, 1972 [syn. *Cithariniella gonzalesi* van Waerebeke, Chabaud, Bain et Georges, 1988] from *Auchenoglanis biscutatus*, *Paradistichodus dimidiatus*, *Synodontis acanthomias*, *S. batensoda*, *S. frontosus*, *S. greshoffi*, *S. longirostris*, *S. membranaceus*, *S. nigrita*, *S. ocellifer*, ***S. schall*** (Chad), *S. serratus*, *S. sorex*

Cithariniella koubkovae Moravec et Van As, 2015 from ***Paradistichodus dimidiatus*** (Senegal)

Cithariniella longicaudata Moravec et Van As, 2015 from ***Schilbe intermedius*** (Botswana) [Fig. 4.8.4I]

Cithariniella petterae Khalil, 1974 from ***Distichodus schenga*** (Zambia), *Synodontis nigrita*, *S. schall*

Synodontisia Petter, Vassiliadès et Troncy, 1972

Synodontisia annulata Moravec et Van As, 2015 from ***Schilbe intermedius*** (Botswana)

Synodontisia okavangoensis Moravec et Van As, 2004 from ***Synodontis nigromaculatus*** (Botswana), *S. vanderwaali* [Fig. 4.8.5A]

Synodontisia thelastomoides Petter, Vassiliadès et Troncy, 1972 from *Synodontis acanthomias*, *S. decorus*, ***S. greshoffi***, *S. nigrita*, *S. nigriventris*, *S. pleurops*, *S. ocellifer*, *S. schall*, ***S. sorex*** (Senegal), *S. zambezensis*, *Xenocharax spilurus*

ASCARIDIDA Skrjabin et Schulz, 1940

Key to the superfamilies of the Ascaridida from African freshwater fishes

1 (2) Lips present or absent, when present variable in number and form. Platymyarian (*i.e.*, having all muscle cells lying next to the hypodermis, their sarcoplasm being uncovered on three sides next to the body cavity). Eggs hatching *in utero* or eggs with delicate shells deposited by females. First moult generally outside eggs. Usually small worms, less than 1 cm long……………………………………………………………………………3

2 (1) With three well-defined lips usually of large size, sometimes separated by interlabia. Coelomyarian (*i.e.*, musculature in which the muscle fibres are next to the hypodermis and perpendicular to it; myofibrils extend varying distances up the side of the muscle cell). Eggs thick-shelled and not embryonated when deposited. First moult inside eggs. Generally large nematodes, more than 1 cm long…………………………………………………5

3 (4) Oesophagus cylindrical, anteriorly differentiated into distinct pharyngeal part, a subspherical or elongate isthmus and a valved bulb possessing uninucleate gland cells. Viviparous nematodes…………**Cosmocercoidea**

4 (3) Oesophagus short, simple and cylindrical, or short and divided into two parts of the same or different diameters. Pharyngeal part of oesophagus present or absent. Oviparous nematodes……………………..**Seuratoidea**

5 (6) Pre-anal sucker present, surrounded by cuticularised ring. Oesophagus with claviform corpus, short isthmus, and valved bulb with binucleated subventral oesophageal glands or oesophagus cylindrical. Caeca absent……………………………………………………………**Heterakoidea**

6 (5) Pre-anal sucker absent. Oesophagus simple and cylindrical or terminated by swelling, without valves, containing uninucleate gland cells. Caeca pre-

sent or absent…………………………………………………...**Ascaridoidea**

COSMOCERCOIDEA Railliet, 1916

Key to the families of the Cosmocercoidea from African freshwater fishes

1 (2) Oviparous, or if viviparous, larvae laid in the first stage. Didelphic…………3

2 (1) Viviparous with larvae laid in an advanced stage of development and capable of endogenous development. Generally monodelphic……………………………………………………...**Atractidae**

3 (4) Oesophageal isthmus elongate, not spherical. Male without a pre-anal sucker………………………………………………………**Cosmocercidae**

4 (4) Oesophageal isthmus not elongate, generally spherical. Male generally with one or several pre-anal suckers……………………………**Kathlanidae**

Atractidae Railliet, 1917

Key to the genera of the Atractidae from African freshwater fishes

1 (2) Oral opening surrounded by three lips………………………….***Labeonema***

2 (1) Oral opening surrounded by 6 (2 lateral and 4 submedian) poorly developed lips……………………………………………………….. ***Orientatractis***

List of the Atractidae (adults) from African freshwater fishes

Labeonema Puylaert, 1970

Labeonema africanum Moravec et Van As, 2004 from ***Synodontis nigromaculatus*** (Botswana), *S. vanderwaali* [Fig. 4.8.3D]

Labeonema bainae Baker, 1982 from ***Schilbe mandibularis*** (Gabon)

Labeonema bakeri van Waerebeke, Chabaud, Bain et Georges, 1988 from ***Distichodus fasciolatus***, ***D. sexfasciatus*** (type host not explicitly mentioned; Central African Republic)

Labeonema intermedium Puylaert, 1970 from ***Labeo* sp.** (Democratic Republic of the Congo)

Labeonema longispiculatum Moravec et Jirků, 2017 from ***Synodontis acanthomias*** (Democratic Republic of the Congo)

Labeonema synodontisi (Vassiliadès, 1973) [syn. *Raillietnema synodontisi* Vassiliades, 1973] from *Synodontis eupterus*, *S. frontosus*, *S. nigrita*, ***S. ocellifer*** (Senegal), *S. schall*, *S. zambezensis*

Orientatractis Petter, 1966

Orientatractis brycini González-Solís et Mariaux, 2017 from ***Brycinus macrolepidotus*** (Gabon), *Xenocharax spilurus* [Fig. 4.8.3E]

Atractidae gen. sp. from *Schilbe intermedius*

List of the Atractidae (larvae) from African freshwater fishes

Atractidae gen. sp. from *Clarias gariepinus*

Cosmocercidae Railliet, 1916

List of the Cosmocercidae from African freshwater fishes

Aplectana Railliet et Henry, 1916

Aplectana chamaeleonis (Baylis, 1929) [syn. *Oxysomatium chamaeleonis* Baylis, 1929] from *Oreochromis niloticus* [Fig. 4.8.3I]

Note: Chen (1966) found this species common in reptiles also in the frog *Amietia angolensis* (Bocage) and a freshwater fish (*Oreochromis niloticus*).

Kathlaniidae Lane, 1914

List of the Kathlanidae from African freshwater fishes

Falcaustra Lane, 1914

Falcaustra hexapapillata (Khalil, 1962) [syns *Spironoura hexapapillata* Khalil, 1962; *Falcaustra guiersi* Vassiliadès, 1973, *Spironoura guiersi* (Vassiliadès, 1973)] from *Distichodus brevipinnis*, ***D. nefasch*** (Sudan), *D. rostratus*

Falcaustra petrei (Khalil, 1970) from *Distichodus nefasch*, ***D. rostratus*** (Ghana)

Falcaustra piscicola (von Linstow, 1907) [syns *Nematoxys piscicola* von Linstow, 1907; *Spironoura congolense* Taylor, 1925] from *Distichodus lusosso*, ***Distichodus* sp.** (Cameroon)

Falcaustra similis Moravec et Van As, 2004 from *Schilbe intermedius*, *Synodontis acanthomias*, *S. frontosus*, *S. nigrita*, ***S. nigromaculatus*** (Botswana), *S. schall*, *S. serratus*, *S. vanderwaali* [Fig. 4.8.4H]

Falcaustra straeleni Campana-Rouget, 1961 from ***Labeobarbus altianalis***, ***L. intermedius*** (type host not explicitly mentioned; Democratic Republic of the Congo)

Falcaustra sudanensis (Khalil, 1962) from ***Distichodus brevipinnis*** (Sudan), *D. nefasch*

Falcaustra tchadi Vassiliadès et Troncy, 1973 from ***Distichodus brevipinnis*** (Chad), *D. rostratus*

Falcaustra therezieni Petter, 1979 from ***Arius madagascariensis***, ***Ptychochromoides betsileanus*** (type host not explicitly mentioned; Madagascar)

Falcaustra verbekei Campana-Rouget, 1961 from ***Labeobarbus altianalis***, ***L. intermedius*** (type host not explicitly mentioned; Democratic Republic of the Congo)

SEURATOIDEA Hall, 1916

Key to the families of the Seuratoidea from African freshwater fishes

1 (2) Buccal cavity absent, or if present, derived from cheilostome; cuticle of walls of cheilostome has same structure and staining reactions as external body cuticle……………………………………………………..**Quimperiidae**

2 (1) Buccal cavity formed from modifications of the anterior end of oesophagus (oesophastome); walls of oesophastome surrounded by oesophageal tissue…………………………………………………………….**Cucullanidae**

Cucullanidae Cobbold, 1864

Key to the genera of the Cucullanidae from African freshwater fishes

1 (2) Intestinal caecum absent………………………………………***Cucullanus***

2 (1) Intestinal caecum present……………………………………….***Dichelyne***

List of the Cucullanidae from African freshwater fishes

Cucullanus Müller, 1777

Cucullanus barbi Baylis, 1923 from *Enteromius perince*, ***Labeobarbus bynni*** (Egypt)

Cucullanus baylisi Campana-Rouget, 1961 from ***Synodontis schall*** (Democratic Republic of the Congo), *Synodontis* sp. [Fig. 4.8.4A]

Cucullanus clarotis Baylis, 1923 from *Clarotes laticeps*, ***Synodontis schall*** (Sudan), *Synodontis* sp.

Cucullanus congolensis Moravec et Jirků, 2017 from ***Auchenoglanis occidentalis*** (Democratic Republic of the Congo)

Cucullanus djilorensis Ndew, Diouf, Bâ et Morand, 2014 from *Labeobarbus bynni*, ***Mugil curema*** (Senegal), *Tilapia sparrmanii*

Cucullanus egyptae Abdel-Ghaffar, Bashtar, Abdel-Gaber, Morsy, Mehlhorn, Al Quraishy et Mohammed, 2014 from ***Anguilla anguilla*** (Egypt)

Cucullanus mormyri Moravec et Scholz, 2017 from *Marcusenius cyprinoides*, ***Mormyrus caschive*** (Sudan), *Mormyrus* sp.

Cucullanus sp. from *Tilapia sparrmanii*

Dichelyne Jägerskiöld, 1902

Dichelyne fossor Jägerskiöld, 1902 from *Bagrus bajad*, ***Lates niloticus*** (Sudan) [Fig. 4.8.4B]

Dichelyne sp. from *Lates niloticus*

Quimperiidae Gendre, 1928

Key to the genera of the Quimperiidae from African freshwater fishes

1 (2) Pre-anal sucker absent in juvenile males, present in fully developed males. Distinct ventral muscle bands in pre-anal region present in males. Parasites of eels……………………………………………….***Paraquimperia***

2 (1) Pre-anal sucker present. Ventral oblique muscle bands in preanal region absent or inconspicuous in males. Parasites of African fishes…………….3

3 (4) Cephalic vesicle absent. Cervical alae well developed. Buccal capsule absent. Oral opening triangular……………………………………***Quimperia***

4 (3) Cephalic vesicle present. Cervical alae usually absent. Buccal cavity with three teeth, the two-ventrolateral ones sometimes reduced. Oral opening oval to circular……………………………………………………….***Gendria***

List of the Quimperiidae from African freshwater fishes

Gendria Baylis, 1930 [syn. *Chabaudus* Inglis et Ogden, 1965]

Gendria chabaudi (Inglis et Ogden, 1965) from ***Heterobranchus bidorsalis*** (Sierra Leone)

Gendria longispiculata Moravec et Jirků, 2017 from ***Schilbe grenfelli*** (Democratic Republic of the Congo)

Gendria polypteri Vassiliadès et Chevalier, 1973 from *Erpetoichthys calabaricus*, ***Polypterus senegalus*** (Senegal)

Gendria thysi (Puylaert, 1970) from ***Parauchenoglanis punctatus*** (Democratic Republic of the Congo)

Gendria tilapiae Baylis, 1930 from ***Sarotherodon galilaeus*** (Mali)

Gendria sanghaensis Moravec et Jirků, 2017 from ***Schilbe marmoratus*** (Central African Rep.) [Fig. 4.8.5G]

Gendria sp. from *Pantodon buchholzi*

Paraquimperia Baylis, 1934

Paraquimperia africana Moravec, Boomker et Taraschewski, 2000 from ***Anguilla mossambica*** (South Africa) [Fig. 4.8.5H]

Quimperia Gendre, 1926

Quimperia lanceolata Gendre, 1926 from ***Ctenopoma kingsleyae*** (Guinea) [Fig. 4.8.5I]

ASCARIDOIDEA Baird, 1853

Key to the families of the Ascaridoidea from African freshwater fishes

1 (2) Oesophagus cylindrical, slightly enlarged posteriorly. Ventriculus absent. Long intestinal caecum present……………………………**Heterocheilidae**

2 (1) Oesophagus with oblong to cylindrical posterior ventriculus, anterior intestinal caecum and posterior ventricular appendix present or both absent…………………………………………………………………………3

3 (2) If only intestinal caecum present, then excretory pore situated between subventral lips or at base of ventral interlabium………………**Anisakidae**

4 (3) Anterior intestinal caecum present, excretory pore approximately at level of nerve ring………………………………………………….. ***Ascarididae**

****Anisakidae Railliet et Henry, 1912

Key to genera of the Anisakidae from African freshwater fishes

1 (2) Anterior intestinal caecum absent and ventricular appendix present ……………………………………………………….***..Raphidascaroides***

2 (1) Both anterior intestinal caecum and ventricular appendix present…………3

3 (2) Anterior end with distinct cuticular collar. Body covered by many small cuticular bosses…………………………………………………………***Galeiceps***

4 (3) Anterior end without distinct cuticular collar. Body without cuticular projecti ons………………………………………………………………………….5

5 (4) Excretory pore situated at level of nerve ring or somewhat posterior, always distant from head end. Tail of fourth-stage larvae with minute cuticular projections at tip. Adults parasitic in fishes …………………***Hysterothylacium***

6 (5) Excretory pore located at base of ventral interlabium. Tail of larvae conical or rounded, without cuticular projections at tip. Adults parasitic in fish-eating birds and marine mammals…………………………***.Contracaecum***

List of the Anisakidae (adults) from African freshwater fishes

Hysterothylacium Ward et Magath, 1917

Hysterothylacium anguillae Moravec, Taraschewski, Appelhoff et Weyl, 2012 from ***Anguilla marmorata*** (South Africa) [Fig. 4.8.3A]

Raphidascaroides Yamaguti, 1941

Raphidascaroides bishaii Khalil, 1961 from *Chrysichthys nigrodigitatus*, ***Gymnarchus niloticus*** (Sudan) [Fig. 4.8.3B]

List of Anisakidae (larvae) from African freshwater fishes

Contracaecum Railliet et Henry, 1912

Contracaecum microcephalum (Rudolphi, 1809) from *Synodontis batensoda*

Contracaecum sp. from *Anguilla mossambica*, *Bagrus bajad*, *B. docmak*, *Boulengerella cuvieri*, *Brycinus imberi*, *B. macrolepidotus*, *B. nurse*, *Campylomormyrus tamandua*, *Chetia flaviventris*, *Clarias gariepinus*, *C. liocephalus*, *C. ngamensis*, *C. platycephalus*, *C. stappersii*, *C. theodorae*, *Clarias* sp., *Clarotes laticeps*, *Coptodon rendalli*, *Cyprinus carpio*, *Decapterus russelli*, *Enteromius humilis*, *E. mattozi*, *E. paludinosus*, *E. trimaculatus*, *E. unitaeniatus*, *Gnathonemus petersii*, *Haplochromis astatodon*, *H. eduardii*, *H. guiarti*, *H. ishmaeli*, *H. mahagiensis*, *H. pappenheimi*, *H. paucidens*, *H. placodus*, *H. serridens*, *Haplochromis* sp., *Hydrocynus brevis*, *H. forskahlii*, *H. vittatus*, *Hydrocynus* sp., *Hyperopisus bebe*, *Labeobarbus altianalis*, *L. marequensis*, *Lates niloticus*, *Lepidiolamprologus cunningtoni*, *Malapterurus electricus*, *Marcusenius stanleyanus*, *Micropterus salmoides*, *Mormyrops anguilloides*, *Oreochromis andersonii*, *O. leucostictus*, *O. macrochir*, *O. mossambicus*, *O. niloticus*, *Pomadasys olivaceus*, *P. commersonnii*, *Pseudocrenilabrus philander*, *Sandelia capensis*, *Sargochromis carlottae*, *S. codringtonii*, *Schilbe intermedius*, *S. mystus*, *Serranochromis angusticeps*, *S. macrocephalus*, *S. robustus*, *Synodontis nigromaculatus*, *Thoracochromis wingatii*, *Tilapia sparrmanii* [Fig. 4.8.6C]

Galeiceps Railliet, 1916

Galeiceps sp. from *Clarias gariepinus*, *Hydrocynus vittatus*, *Thoracochromis wingatii*

Hysterothylacium Ward et Magath, 1917

Hysterothylacium sp. from *Chrysichthys nigrodigitatus*, *Thoracochromis wingatii*

*****Ascarididae Baird, 1853

Key to the genera of the Ascarididae from African freshwater fishes

1 (2) Oesophagus with spherical or elongate ventriculus……….****Porrocaecum***

2 (1) Oesophagus without ventriculus……………………………. ****Amplicaecum***

List of the Ascarididae from African freshwater fishes

Amplicaecum Baylis, 1920

Amplicaecum sp. (type I) from *Alestes baremoze*, *A. dentex*, *Brycinus nurse*, *Bagrus bajad*, *B. docmak*, *Clarias anguillaris*, *C. gariepinus*, *Coptodon zillii*, *Hydrocynus brevis*, *H. forskahlii, H. vittatus*, *Labeo niloticus*, *Malapterurus electricus*, *Oreochromis niloticus*, *Polypterus endlicheri*, *P. senegalus*, *Sarotherodon galilaeus*, *Schilbe mystus*, *S. uranoscopus*, *Synodontis schall*

Amplicaecum sp. (type II) from *Coptodon zillii*, *Oreochromis niloticus*, *Sarotherodon galilaeus*

Porrocaecum Railliet et Henry, 1912

Porrocaecum sp. from *Clarias buthupogon*, *C. dumerilii*, *Micropterus salmoides*, *Periophthalmus barbarus*, *Tilapia* sp.

**Heterocheilidae Railliet et Henry, 1915

Key to the genera of the Heterocheilidae from African freshwater fishes

1 (2) Oesophagus with small posterior ventriculus with two anteriorly and three posteriorly directed appendices of different sizes……………***Multicaecum***

2 (1) Ventriculus without appendices…………………………..***Dujardinascaris***

List of the Heterocheilidae (adults) from African freshwater fishes

Dujardinascaris Baylis, 1947

Dujardinascaris malapteruri (Baylis, 1923) from ***Malapterurus electricus*** (Sudan)

Dujardinascaris mormyropsis Moravec et Jirků, 2014 from ***Mormyrops anguilloides*** (Central African Rep.) [Fig. 4.8.4F]

Dujardinascaris sp. from *Lates microlepis*

Multicaecum Baylis, 1923

Multicaecum heterotis Petter, Vassiliadès et Marchand, 1979 from ***Heterotis niloticus*** (Senegal) [Fig. 4.8.4E]

SPIRURIDA Chitwood, 1933

Key to the suborders and superfamilies of the Spirurida from African freshwater fishes

1 (2) Pseudolabia always absent. Buccal capsule well developed, reduced or absent. Oesophagus divided into muscular and glandular portions or muscular throughout. Oesophageal glands usually uninucleate. Larvae without cephalic hooks, tail generally long and pointed, usually with conspicuous phasmids containing broad cavities and prominent pores. Parasites of gut of cold-blooded vertebrates or other organs of all classes of vertebrates. Intermediate hosts mostly copepods, rarely branchiurids or ostracods (suborder **Camallanina**)……………………………………………………………3

2 (1) Head end with pseudolabia, sometimes rudimentary. Buccal capsule (stoma) usually elongate, moderately sclerotised tube. Oesophagus divided into well differentiated muscular and glandular parts. Oesophageal glands

usually multinucleate. Larvae usually with cephalic hooks or spines and inconspicuous pore-like phasmids, containing broad cavities and prominent pores. Parasites of gut or tissues of all classes of vertebrates. Intermediate host invertebrates other than copepods (except Gnathostomatoidea) (suborder **Spirurina**)……………………………………………………………7

3 (4) Buccal capsule well developed, orange-brown in colour. Buccal cavity well developed; internal labial papillae tiny; parasitic in the digestive tract……………………………………………………………**Camallanoidea**

4 (3) Buccal capsule reduced or absent (except for Anguillicoloidea). If buccal cavity present then simple, rounded, not separated into two valves, tridents absent; internal labial papillae prominent. Not usually parasitic in the digestive tract……………………………………………………………………5

5 (6) Oviparous. Buccal capsule well developed. Oesophagus short, undivided. Sclerotised copulatory organs absent. Vulva functional. Parasites of swim bladder of eels………………………………………………**Anguillicoloidea**

6 (5) Viviparous. Buccal capsule usually reduced or absent. Oesophagus divided into muscular and glandular portions or muscular throughout. Spicules, copulatory plate or sclerotised genital cone present. Anus and vulva in gravid worms sometimes atrophied. Parasites of tissues, body cavity or closed cavities and organs of vertebrates…………………**Dracunculoidea**

7 (8) Buccal capsule well cuticularised, elongate or short. Pseudolabia present or absent…………………………………………………………………………9

8 (7) Buccal capsule weakly cuticularised; two massive lateral trilobed pseudolabia present. Cuticle on the inner face of each pseudolabium thick, generally folded into rounded tooth-like formations that fit into corresponding folds on adjacent pseudolabium. Anterior extremity sometimes swollen into a bulb. Intermediate hosts are copepods or molluscs. Adults in fishes, reptiles and mammals, only larvae in fishes. Note: from Africa unpublished data…………………………………………………..*__Gnathostomatoidea__

9 (10) Pseudolabia absent. Buccal capsule variable, sometimes long and cylindrical. Mouth opening hexagonal or oval. Caudal papillae not arranged as in typical spirurid. Adults in the intestine of fishes……………**Thelazioidea**

10 (9) Pseudolabia present……………………………………………………… 11

11 (12) Two small pseudolabia present. Cephalic and outer labial papillae not fused. Adults in various organs of fishes………………..**Habronematoidea**

12 (11) Two large pseudolabia present. Cephalic and outer labial papillae fused………………………………………………………………………13

13 (14) Pseudolabia involving entire cephalic surface. Cuticular cephalic ornamentation present, in form of cordons, collarettes or ptilina derived from ante-

rior cuticular structures. Larvae parasitic in arthropods and fishes, adults in birds………………………………………………………........... **Acuarioidea**

14 (13) Pseudolabia with a variable number of teeth on their free borders. Body cuticle immediately behind pseudolabia often expanded to form collarette. Male with ornamented cuticle in posteroventral region (area rugosa). Intermediate hosts insects…………………………………….**Physalopteroidea**

**ANGUILLICOLOIDEA Yamaguti, 1935

**Anguillicolidae Yamaguti, 1935

List of the Anguillicolidae from African freshwater fishes

***Anguillicoloides* Moravec et Taraschewski, 1988

Anguillicoloides crassus (Kuwahara, Niimi et Hagaki, 1974) from *Anguilla* spp.

Anguillicoloides papernai (Moravec et Taraschewski, 1988) from ***Anguilla mossambica*** (South Africa) [Fig. 4.8.3C]

CAMALLANOIDEA Railliet et Henry, 1915

Camallanidae Railliet et Henry, 1915

Key to the subfamilies, genera and subgenera of the Camallanidae from African freshwater fishes

1 (2) Buccal capsule round, continuous and not separated into two valves (*i.e.*, single cup-shaped object) (subfamily **Procamallaninae**)…………………3

2 (1) Buccal capsule divided into two valves united by a posterior hinge (subfamily **Camallaninae**)……………………………………………………………5

3 (1) Buccal capsule interior smooth and without markings in both males and females………………………………...***Procamallanus* (*Procamallanus*)**

4 (3) Buccal capsule interior with ridges arranged in a spiral…………………………………***Procamallanus* (*Spirocamallanus*)**

5 (2) Buccal capsule composed of two chambers; buccal cavity behind valves large……………………………………………………….***.Paracamallanus***

6 (5) Buccal capsule composed of two chambers; buccal cavity behind valves reduced to basal ring……………………………………………***.Camallanus***

List of the Camallanidae (adults) from African freshwater fishes

Camallanus Railliet et Henry, 1915

Camallanus (*Zeylanema*) *ctenopomae* Vassiliadès et Petter, 1972 [syn. *Camallanus ctenopomae* Vassiliadès et Petter, 1972] from ***Ctenopoma kingsleyae*** (Senegal), *Ctenopoma* sp.

Camallanus kirandensis Baylis, 1928 from ***Barbus* sp.** (Tanzania), *Labeo altivelis*, *L. niloticus*

Camallanus longicaudatus Moravec, 1973 from ***Labeo horie*** (Egypt), *Labeo niloticus* [Fig. 4.8.3F]

Camallanus polypteri Kabré et Petter, 1997 from *Clarias anguillaris*, ***Polypterus bichir*** (Burkina Faso), *Synodontis schall*

Camallanus sp. from *Clarias gariepinus*, *Clarotes laticeps*, *Coptodon zillii*, *Enteromius paludinosus*, *Labeo molybdinus*

Paracamallanus Yorke et Maplestone, 1926

Paracamallanus cyathopharynx Baylis, 1923 [syn. *Paracamallanus senegalensis* Vassiliadès, 1970] from *Clariallabes laticeps*, *Clarias anguillaris*, ***C. gariepinus*** (Egypt), *C. stappersii*, *C. theodorae*, *C. werneri*, *Clarias* sp., *Clarotes laticeps*, *Heterobranchus longifilis*, *Hydrocynus vittatus*, *Schilbe intermedius*, *Synodontis zambezensis*

Paracamallanus sp. from *Clarias gariepinus*

Procamallanus Baylis, 1923

Procamallanus (*Procamallanus*) *armatus* Campana-Rouget et Therezien, 1965 from ***Anguilla* sp.** (Madagascar)

Procamallanus (*Procamallanus*) *laeviconchus* (Wedl, 1861) [syn. *Cucullanus laeviconchus* Wedl, 1861] from *Astatotilapia desfontainii*, *Auchenoglanis biscutatus*, *A. occidentalis*, *Bagrus bajad*, *B. docmak*, *Campylomormyrus tamandua*, *Citharinus citharus*, *C. gibbosus*, *Clarias anguillaris*, *C. gariepinus*, *Distichodus brevipinnis*, *D. nefasch*, *D. rostratus*, *Malapterurus electricus*, *Marcusenius cyprinoides*, *Mormyrops anguilloides*, *Mormyrus caschive*, *M. rume*, *Schilbe intermedius*, *S. mystus*, *Synodontis batensoda*, *S. clarias*, *S. membranaceus*, *S. nigrita*, *S. nigromaculatus*, *S. ocellifer*, ***S. schall*** (Egypt), *S. sorex*, *S. thamalakanensis*, *S. vanderwaali*, *Tetraodon lineatus*

Procamallanus (*Procamallanus*) *pseudolaeviconchus* Moravec et Van As, 2014 from *Clarias alluaudi*, *C. anguillaris*, ***C. gariepinus*** (Egypt), *C. stappersii*, *C. theodorae*

Procamallanus (*Procamallanus*) *siluranae* (Jackson et Tinsley, 1995) from *Erpetoichthys calabaricus* (accidental infection)

Procamallanus (*Spirocamallanus*) *daleneae* (Boomker, 1993) from *Synodontis acanthomias*, *S. afrofischeri*, *S. batensoda*, *S. eupterus*, *S. haugi*, *S. membranaceus*, *S. ocellifer*, *S. schall*, *S. tessmanni*, *S. vanderwaali*, *S. victoriae*, ***S. zambezensis*** (South Africa)

Procamallanus (*Spirocamallanus*) *olseni* (Campana-Rouget et Razarihelissoa, 1965) from ***Rhabdosargus sarba*** (inland brackish-water lake) (South Africa)

Procamallanus (*Spirocamallanus*) *parachannae* Moravec et Jirků, 2015 from ***Parachanna insignis*** (Sudan)

Procamallanus (*Spirocamallanus*) *pseudospiralis* Moravec et Jirků, 2017 from *Synodontis frontosus*, *S. nigrita*, ***S. schall*** (Democratic Republic of the Congo)

Procamallanus (*Spirocamallanus*) *serranochromis* Moravec et Van As, 2015 from *Serranochromis angusticeps*, ***S. macrocephalus*** (Botswana), *S. robustus*

Procamallanus (*Spirocamallanus*) *spiralis* Baylis, 1923 [syn. *Spirocamallanus mazabukae* Yeh, 1957] from *Auchenoglanis occidentalis*, ***Clarias anguillaris*** (Egypt), *C. gariepinus*, *C. stappersii*, *C. theodorae*, *Hepsetus odoe*, *Polypterus endlicheri*, *Synodontis eupterus*, *S. tessmanni* [Fig. 4.8.3G]

Procamallanus (*Spirocamallanus*) sp. from *Synodontis afrofischeri*, *S. membranaceus*, *S. ocellifer*, *S. schall*

Procamallanus sp. from *Clarias gariepinus*, *C. pachynema*, *Clarias* sp., *Mormyrus* sp., *Synodontis schall*

List of the Camallanidae (larvae) from African freshwater fishes

Camalanidae gen. sp. from *Nothobranchius furzeri*, *N. kadleci*, *N. orthonotus*, *N. pienaari*

DRACUNCULOIDEA Cameron, 1934

Key to the families and genera of the Dracunculoidea from African freshwater fishes

1 (2) Spicules absent. Oesophagus distinctly divided into short anterior muscular part and posterior, longer glandular part with two large cell nuclei. Vulva anterior or pre-equatorial, well developed in mature female……………………………….family **Daniconematidae** (***Mexiconema***)

2 (1) Spicules present. Oesophagus short, undivided or with markedly large unicellular dorsal oesophageal gland with large cell nucleus; anterior part of oesophagus often bulbously inflated. Vulva posterior or equatorial, more or less completely atrophied in gravid female (family **Philometridae**)……3

3 (4) Cuticle of adult females smooth………………………………….***Philometra***

4 (3) Cuticle of adult females with ornamentation…………………………………5

5 (6) Cuticle of adult females covered with numerous narrow transverse bands of raised cuticle, interrupted by narrow smooth lateral fields………………………………………………………..***Afrophilometra***

6 (5) Cuticle of adult females with bosses…………………………………………7

7 (6) Bosses rounded…………………………………………….***Philometroides***

8 (7) Bosses conical…………………………………………………….***Nilonema***

Daniconematidae Moravec et Køie, 1987

List of the Daniconematidae from African freshwater fishes

Mexiconema Moravec, Vidal et Salgado-Maldonado, 1992

Mexiconema africanum Moravec, Jirků, Charo-Karisa et Mašová, 2009 from ***Auchenoglanis occidentalis*** (Kenya) [Fig. 4.8.4D]

PHILOMETRIDAE Baylis et Daubney, 1926

List of the Philometridae from African freshwater fishes

Afrophilometra Moravec, Charo-Karisa et Jirků, 2009

Afrophilometra hydrocyoni (Fahmy, Mansour et El-Naffar, 1976) [syn. *Philometroides hydrocyonae* Fahmy, Mandour et El-Nafar, 1976] from ***Hydrocynus forskahlii*** (Egypt), *H. vittatus* [Fig. 4.8.5B]

Nilonema Khalil, 1960

Nilonema gymnarchi Khalil, 1960 from ***Gymnarchus niloticus*** (Sudan) [Fig. 4.8.5C]

Philometra Costa, 1845

Philometra bagri (Khalil, 1965) [syn. *Thwaitia bagri* Khalil, 1965] from ***Bagrus bajad*** (Sudan)

Philometra lati Moravec, Charo-Karisa et Jirků, 2009 from ***Lates niloticus*** (Kenya) [Fig. 4.8.5D]

Philometra spiriformis Moravec, Charo-Karisa et Jirků, 2009 from ***Lates niloticus*** (Kenya)

Philometroides Yamaguti, 1935

Philometroides africanus Moravec et Van As, 2001 from ***Hepsetus odoe*** (Botswana)

Philometroides khalili Moravec, Halajian, Tavakol, Nyagura et Luus-Powell, 2015 from *Labeo altivelis*, ***L. rosae*** (Zimbabwe) [Fig. 4.8.5E]

Philometridae gen. sp. from *Schilbe intermedius*, *Serranochromis meridianus*, *Synodontis zambezensis*

GNATHOSTOMATOIDEA Railliet, 1895

*Gnathostomatidae Railliet, 1895

List of the Gnathostomatidae (larvae) from African freshwater fishes

Gnathostomatidae gen. sp. from *Nothobranchius furzeri*, *N. kadleci*, *N. orthonotus*, *N. pienaari*

PHYSALOPTEROIDEA Railliet, 1893

Physalopteridae Railliet, 1893

List of the Physalopteridae from African freshwater fishes

Heliconema Travassos, 1919

Heliconema africanum (von Linstow, 1899) [syn. *Spiropterina africana* von Linstow, 1899] from ***Anguilla mossambica*** (South Africa) [Fig. 4.8.5F]

Physalopteridae gen. sp. from *Clarias gariepinus*, *Serranochromis angusticeps*

THELAZIOIDEA **Skryabin, 1915

Rhabdochonidae **Travassos, Artigas et Pereira, 1928

Key to the genera/subgenera of the Rhabdochonidae from African freshwater fishes

1(2) Two small unarmed pseudolabia, without teeth. Oesophagus long, not clearly divided into muscular and glandular portions ……………………………………………………genus ***Prosungulonema***

2(1) Pseudolabia rudimentary, prostom with teeth. Oesophagus divided into muscular and glandular portions (genus ***Rhabdochona***)…………………3

3(2) Prostom with 8 or 12 anterior teeth…………………subgenus ***Globochona***

4(3) Prostom with 14 anterior teeth……………………subgenus ***Rhabdochona***

List of the Rhabdochonidae (adults) from African freshwater fishes

Prosungulonema Roytman, 1963

Prosungulonema africanum (Moravec et Puylaert, 1970) [syn. *Johnstonmawsonia africana* Moravec et Puylaert, 1970] from ***Thoracochromis schwetzi*** (Angola) [Fig. 4.8.6A]

Prosungulonema campanae (Puylaert, 1973) [syn. *Johnstonmawsonia campanae* Puylaert, 1973] from ***Aphyosemion cameronense*** (Cameroon), *Thoracochromis schwetzi*

Rhabdochona Railliet, 1916

Rhabdochona (*Rhabdochona*) *centroafricana* Moravec et Jirků, 2014 from ***Enteromius miolepis*** (Central African Republic) [Fig. 4.8.6B]

Rhabdochona (*Rhabdochona*) *esseniae* Mashego, 1990 from *Enteromius lineomaculatus*, *E. paludinosus*, *E. trimaculatus*, *Labeobarbus marequensis* (South Africa)

Rhabdochona (*Rhabdochona*) *gendrei* Campana-Rouget, 1961 from ***Barbus* sp.** (Gambia), *Enteromius camptacanthus*, *E. lineomaculatus*, *E. paludinosus*, *E. trimaculatus*, *Labeobarbus altianalis*, *L. bynni*, *L. intermedius*, *L. marequensis*

Rhabdochona (*Rhabdochona*) *marcusenii* Moravec et Jirků, 2014 from ***Marcusenius greshoffii*** (Central African Republic)

Rhabdochona (*Rhabdochona*) *moraveci* Puylaert, 1973 [syn. *Afrochona camerounensis* Puylaert, 1973] from ***Aphyosemion cameronense*** (Cameroon)

Rhabdochona (*Rhabdochona*) *srivastavai* Chabaud, 1970 from ***Sicyopterus fasciatus*** (Madagascar)

Rhabdochona (*Globochona*) *gambiana* Gendre, 1922 [syn. *Cystidicola minuta* Rodhain et Vuylsteke, 1934] from *Enteromius eutaenia*, *Raiamas moorii*

Rhabdochona (*Globochona*) *paski* Baylis, 1928 [syns *Rhabdochona aegyptica* El-Nafar et Saoud, 1974; *Rhabdochona congolensis* Campana-Rouget, 1961; *Rhabdochona versterae* Boomker et Petter, 1993] from *Alestes baremoze*, *A. dentex*, ***A. macrophthalmus*** (Tanzania), *Anguilla anguilla*, *Bagrus bajad*, *B. docmak*, *Brycinus imberi*, *B. nurse*, *Chrysichthys nigrodigitatus*, *Clarias gariepinus*, *Coptodon zillii*, *Ctenopoma kingsleyae*, *Haplochromis eduardii*, *H. elegans*, *H. graueri*, *H. ishmaeli*, *H. nigripinnis*, *H. nubilus*, *H. serridens*, *H. squamipinnis*, *Haplochromis* sp., *Hydrocynus brevis*, *H. forskahlii*, *Labeobarbus altianalis*, *L. bynni*, *Oreochromis niloticus*, *Phenacogrammus aurantiacus*, *Polypterus senegalus*, *Schilbe mystus*, *Synodontis batensoda*, *S. nigromaculatus*, *S. notatus*, *S. schall*, *Thoracochromis wingatii*

Rhabdochona (*Globochona*) *tricuspidata* Moravec et Jirků, 2014 from ***Raiamas christyi*** (Central African Republic)

Rhabdochona (*Globochona*) sp. from *Epiplatys multifasciatus*

Rhabdochona sp. from *Anguilla anguilla*, *Clarias gariepinus*, *Coptodon rendalli*, *Hydrocynus forskahlii*, *Labeobarbus bynni*, *Schilbe intermedius*, *Serranochromis meridianus*, *Synodontis zambezensis*

List of the Rhabdochonidae (larvae) from African freshwater fishes

Rhabdochona Railliet, 1916

Rhabdochona (*Globochona*) *paski* Baylis, 1928 [syn. *R. congolensis* Campana-Rouget, 1961] from *Auchenoglanis occidentalis*, *Synodontis nigromaculatus*, *Thoracochromis wingatii*

Rhabdochona sp. from *Brycinus macrolepidotus*, *Labeo niloticus*, *Mormyrus caschive*

HABRONEMATOIDEA Chitwood et Wehr, 1932

Cystidicolidae Skryabin, 1946

Key to the genera of the Cystidicolidae from African freshwater fishes

1(2) Cephalic cuticle forming collarette similar to that of physalopterids. Parasitic in the alimentary tract of marine and freshwater fishes……………………………………………………..***Pseudoproleptus***

2 (1) Cuticle with marked ornamentation in form of numerous transverse rings with spines. Parasitic in the alimentary tract of fishes, generally in freshwater……………………………………………………….***Spinitectus***

List of the Cystidicolidae from African freshwater fishes

Pseudoproleptus Khera, 1953

Pseudoproleptus africanus Khalil, 1973 from ***Mormyrus* sp.** (Democratic Republic of the Congo) [Fig. 4.8.4C]

Spinitectus Fourment, 1883

Spinitectus allaeri Campana-Rouget, 1961 [syns *Spinitectus macheirus* Boomker et Puylaert, 1994; *Spinitectus macilentus* Boomker et Puylaert, 1994; *Spinitectus minusculus* Boomker et Puylaert, 1994; *Spinitectus moraveci* Boomker et Puylaert, 1994] from ***Alestes dentex***, ***Bagrus bajad***, *B. docmak*, *Clarias gariepinus*, *Clarias* sp., *Heterobranchus isopterus*, ***Lates niloticus***, ***Malapterurus electricus***, *Mormyrus caschive*, *Pantodon buchholzi*, ***Schilbe mystus*** (type host not explicitly mentioned, all from Democratic Republic of the Congo), *Synodontis schall*, *Xenoclarias eupogon*

Spinitectus macilentus Boomker et Puylaert, 1994 from ***Heterobranchus isopterus*** (Ivory Coast)

Spinitectus maleficus Boomker et Puylaert, 1994 from ***Mastacembelus flavidus*** (Democratic Republic of the Congo)

Spinitectus micropectus Boomker et Puylaert, 1994 from ***Mastacembelus micropectus*** (Democratic Republic of the Congo)

Spinitectus monstrosus Boomker et Puylaert, 1994 from ***Mormyrops boulengeri*** (Democratic Republic of the Congo)

Spinitectus mormyri Campana-Rouget, 1961 from ***Mormyrus caschive*** (Democratic Republic of the Congo), *M. rume*

Spinitectus mucronatus Boomker et Puylaert, 1994 from ***Mormyrops anguilloides*** (Democratic Republic of the Congo), *Mormyrops boulengeri*, *M. zanclirostris*

Spinitectus petterae Boomker, 1993 from ***Clarias gariepinus*** (South Africa)

Spinitectus polli Campana-Rouget, 1961 [syn. *Spinitectus zambezensis* Boomker, 1993] from *Synodontis decorus*, *S. nigromaculatus*, ***S. schall*** (Democratic Republic of the Congo), *S. zambezensis* [Fig. 4.8.4G]

Spinitectus thurstonae Ogden, 1967 from ***Mormyrus* sp.** (Uganda)

Spinitectus sp. from *Alestes macrolepidotus*, *Hydrocynus forskahlii*, *H. vittatus*, *Schilbe intermedius*, *Synodontis nigromaculatus*

** ACUARIOIDEA Railliet, Henry et Sisoff, 1912

**Acuariidae Railliet, Henry et Sissoff, 1912

List of the Acuariidae from African freshwater fishes

***Chordocephalus* Alegret, 1941

Chordocephalus sp. [syn. *Skrjabinocara* sp.] from *Clarias gariepinus*

Note: one adult female, three fourth-stage larvae and one unidentifiable nematode found in one catfish; they were very probably ingested by the catfish after regurgitation by a white-breasted cormorant whilst feeding chicks (Boomker 1982).

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Chapter 4.9.

CRUSTACEA

Nico SMIT & Kerry HADFIELD

Crustaceans (Crustacea) – basic characteristics, life cycles, classification and principal diagnostic features

- phylum Arthropoda: subphylum Crustacea
- approximately 67,000 species classified in 65 orders (including free-living and parasitic)
- no internal skeleton – only hard chitinous exoskeleton
- old exoskeleton shed by moulting
- two pairs of antennae
- three pairs of mouthparts
- head with two compound eyes
- biramous appendages (split into two)
- gaseous exchange through gills
- abdominal segments with swimming legs (swimmerets)
- tail segment fan-shaped, ending in a telson and uropods
- sexual reproduction (most have separate sexes but some are hermaphrodites)
- several larval stages (including the nauplius larva)
- circulatory system open (no heart, blood does not circulate in blood vessels)
- two part-nervous system with a ventral nerve cord and system of ganglia

The general classification of free-living and parasitic crustaceans is based on well-defined orders (65 at present). In almost every order of the Crustacea there are species in some kind of association with other species. This ranges from facultative to highly specialised parasitism where the parasite undergoes total morphological adaptation, becoming metabolically completely reliant on the host for its survival. In freshwater, crustaceans are mostly associated with fish, but there are a few examples of lernaeid copepods and branchiurans associated with tadpoles and invertebrates.

Life cycles and strategies differ among the different groups of parasitic Crustacea and range from temporary parasites that move between the hosts and substrate (*e.g.*, *Argulus* spp.) and permanently attached parasites (*e.g.*, *Chonopeltis* spp.) that complete their life cycle on the hosts. The number of life stages also differs between groups where, for example, the larvae of species from the copepod genera *Ergasilus* and *Lamproglena* are free swimming and only females become parasitic after a few moults (as copepodites) [Fig. 4.9.1]. All copepod appendages can exhibit sexual dimorphism, but typically this is most commonly found in the antennulae, maxillipeds and fifth swimming legs. The precise pattern of sexual dimorphism is highly variable and thus the taxonomy of parasitic copepods is based on the adult female in most cases.

The parasitic Crustacea are characterised mainly by the morphology of the body, mouthparts and appendages. Generic classification and species identification is also based on the size and shape of the various appendages and many other characteristics (see below).

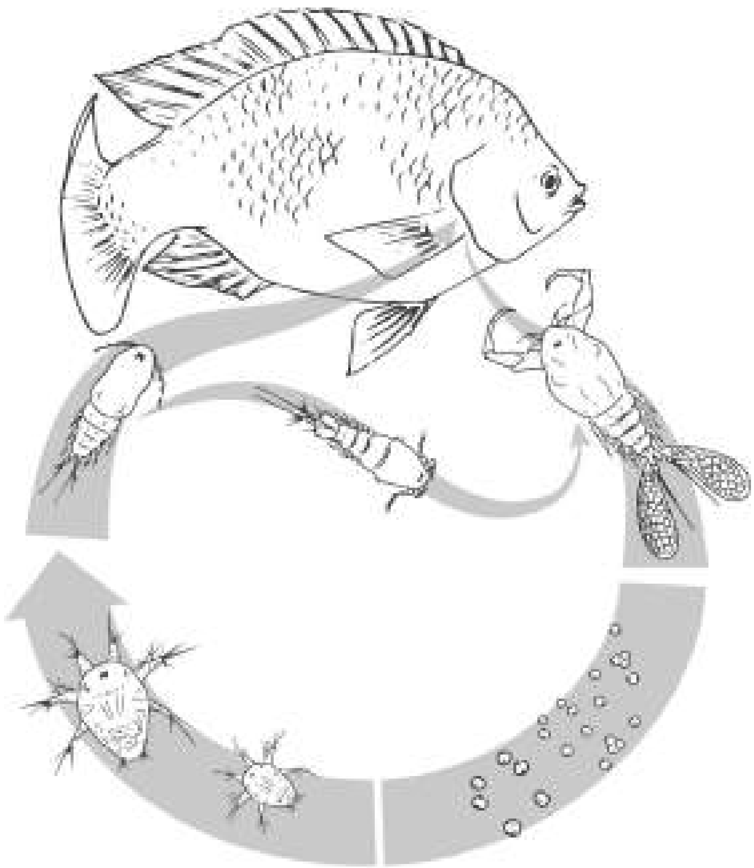

Fig. 4.9.1. Life cycle of *Ergasilus* sp. showing the free-living naupliar and copepodid stages as well as the parasitic adult female. (Illustration by M. Luo.)

Key to the orders of crustaceans (Crustacea) from African freshwater fishes

1 (2) Body dorsoventrally flattened; 4-7 pairs of legs……………………………3

2 (1) Body elongate and not flattened…………………………………………5

3 (4) Suction discs or hooks present; cephalic shield present; second maxillae terminating with large hooks; egg-sacs present; four pairs of legs on the thorax [Fig. 4.9.2A,B]…………………………………………… **Arguloida**

4 (3) Suction discs and shield absent; no cephalic shield; mouthparts form a tightly sealed mouth cone; egg sacs absent (young in brood pouch); seven pairs of legs on the thorax [Fig. 4.9.4A]……………………………**.Isopoda**

5 (6) Worm-like body shape, body clearly segmented; anteriorly there is a singular mouth and two pairs of hooks; egg-sacs absent [Fig. 4.9.5F] …………………………………………………………**.Porocephalida**

6 (5) Body cylindrical or irregularly shaped, not always clearly segmented; egg-sacs present (from female abdomen)………………………………………7

7 (8) Oral cone present with stylet-like mandibles [Fig. 4.9.6B] ………………………………………………………**Siphonostomatoida**

8 (7) Oral cone absent; often falcate mandibles………………………………..9

9 (10) Long, cylindrical body; antennulae long (but shorter than the length of the body); uniramous antennae [Fig. 4.9.3D]……………………….**Cyclopoida**

10 (9) Body cylindrical or abdomen narrower than the thorax; antennulae reduced in size; antennae modified into hooks [Fig. 4.9.4C]…………………………………………**Poecilostomatoida**

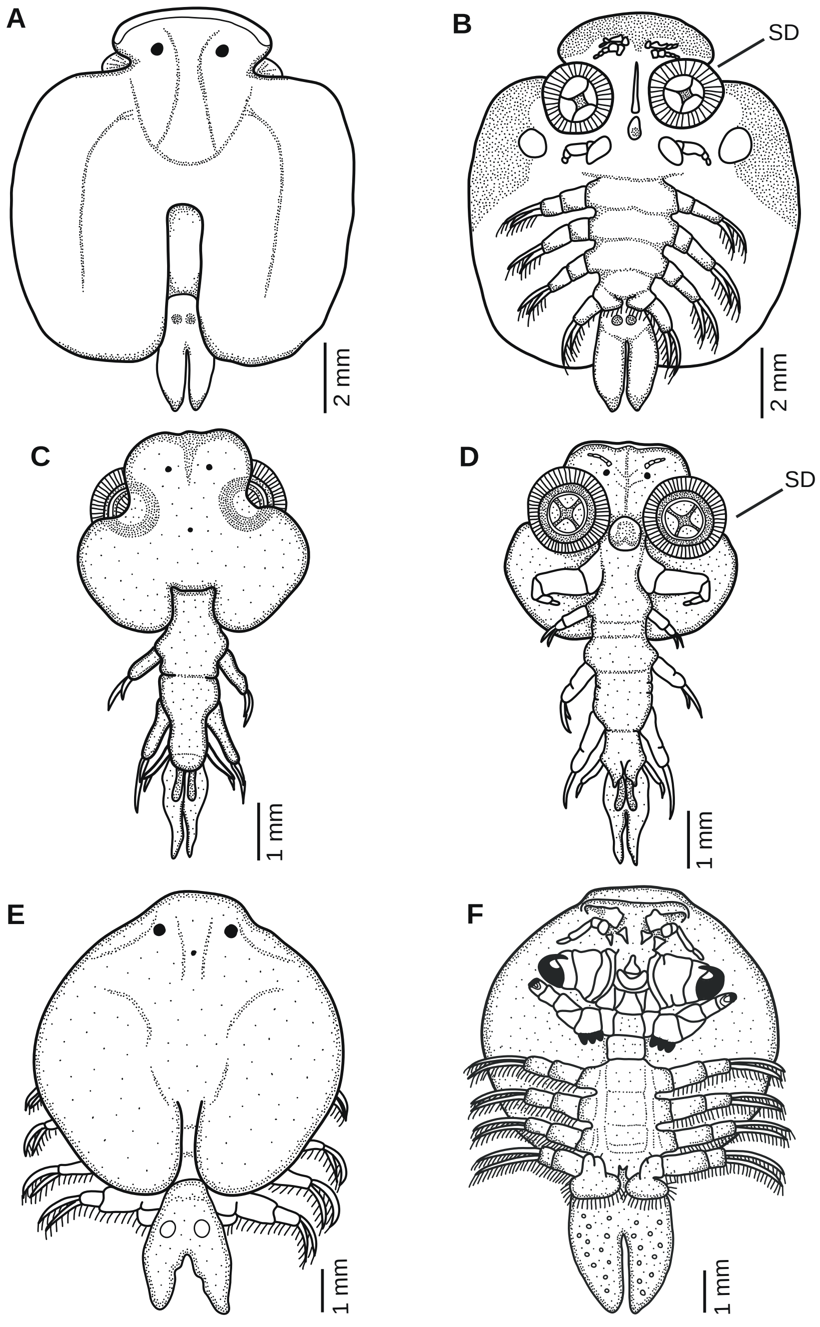

Fig. 4.9.2. Arguloida. **A, B.** *Argulus multipocula* Barnard, 1955, dorsal and ventral views; **C, D.** *Chonopeltis flaccifrons* Fryer, 1960, dorsal and ventral views; **E, F.** *Dolops ranarum* (Stuhlmann, 1892), dorsal and ventral views. (Modified from Fryer 1960; Avenant *et al.* 1989; Smit *et al.* 2005.) SD = suction discs.

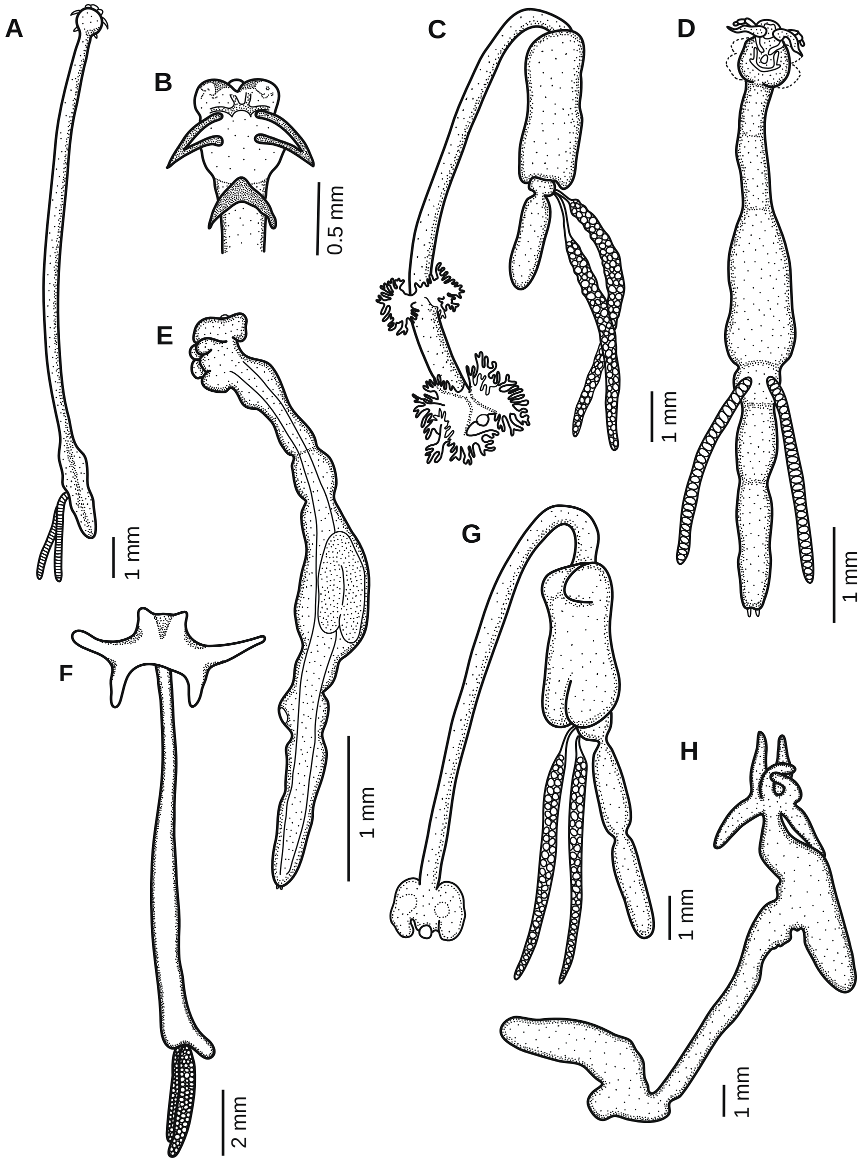

Fig. 4.9.3. Cyclopoida. **A, B.** *Afrolernaea longicollis* Fryer, 1956, entire body and cephalon; **C.** *Dysphorus torquatus* Kurtz, 1924, entire body; **D.** *Lamproglena clariae* Fryer, 1956; **E.** *Lamproglenoides vermiformis* Fryer, 1964; **F.** *Lernaea cyprinacea* Linnaeus, 1758; **G.** *Lernaeogiraffa heterotidicola* Zimmermann, 1922; **H.** *Opistholernaea laterobrachialis* (Fryer, 1959). (Modified from Kurtz 1924; Fryer 1956, 1959, 1964; Marx & Avenant-Oldewage 1996.)

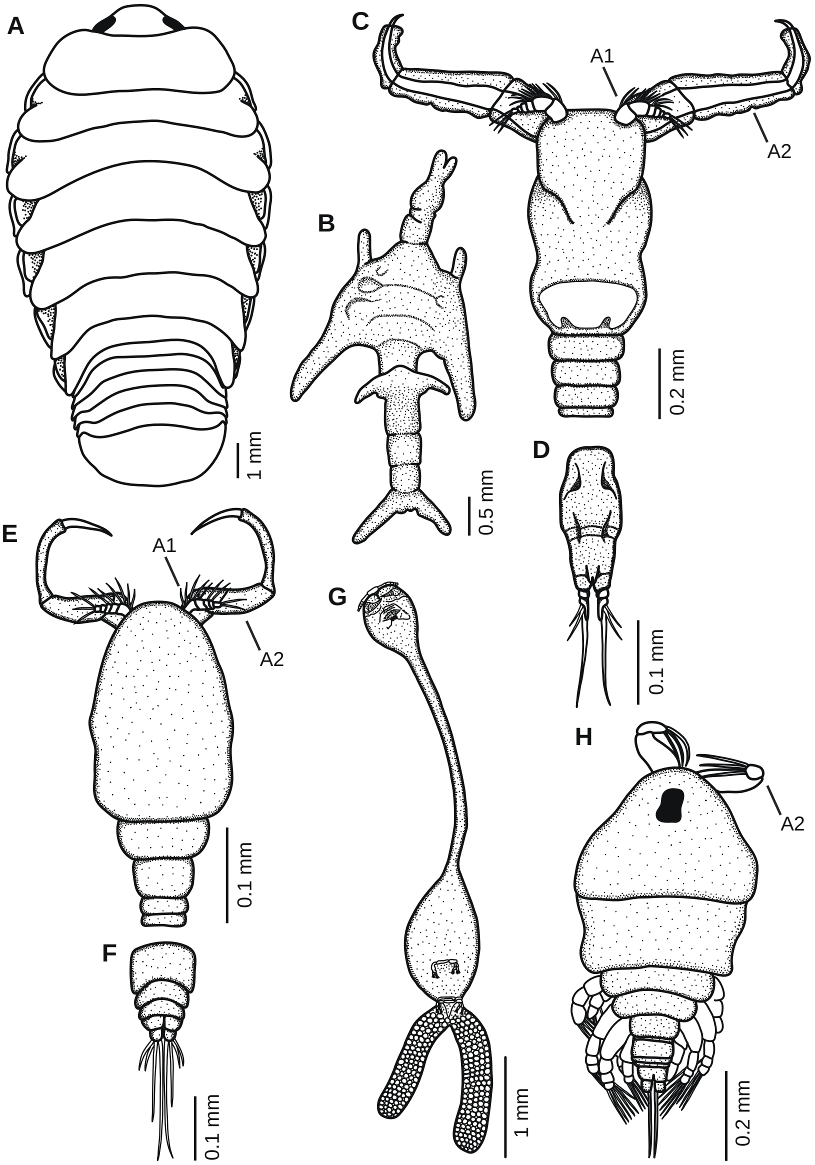

Fig. 4.9.4. Isopoda. **A.** *Ichthyoxenos tanganyikae* (Fryer, 1965); Poecilostomatoida. **B.** *Colobomatus mugilis* Raibaut, Caillet et Ben Hassine, 1978; **C, D.** *Dermoergasilus mugilis* Oldewage et Van As, 1988, dorsal view of antennae, cephalothorax and thorax, and ventral view of genital segment and abdomen; **E, F.** *Ergasilus ilani* Oldewage et Van As, 1988, dorsal view of antennae, cephalothorax and thorax, and ventral view of genital segment and abdomen; **G.** *Mugilicola smithae* Jones et Hine, 1978; **H.** *Paraergasilus minutus* (Fryer, 1956). (Modified from Fryer 1956, 1965; Oldewage & Van As 1988b; Radujković & Raibaut 1990; Kruger *et al.* 1998.) A1 = first antenna; A2 = second antenna.

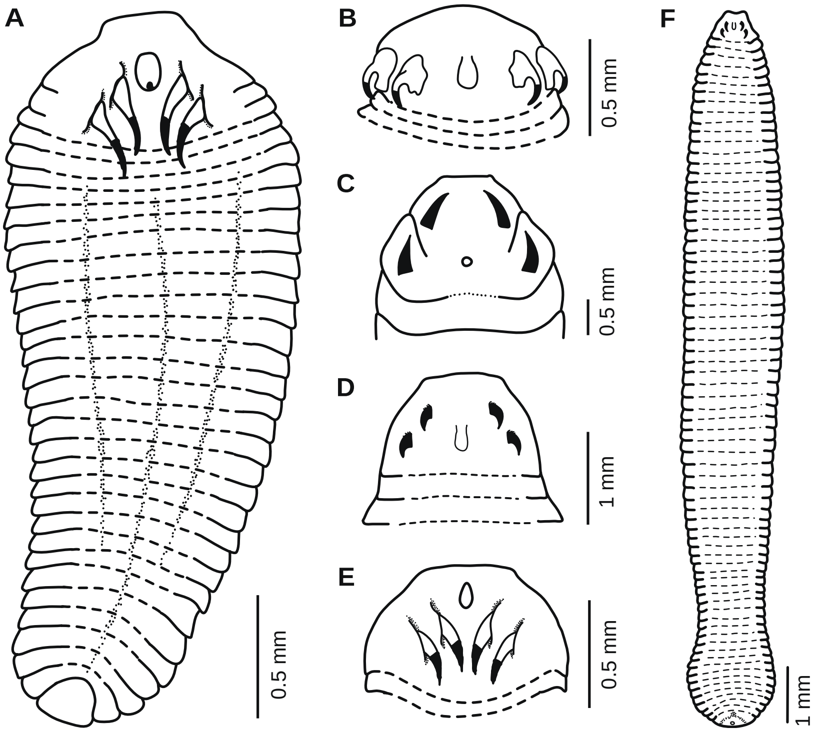

Fig. 4.9.5. Porocephalida. **A.** *Subtriquetra rileyi* Junker, Boomker et Booyse, 1998, entire body of infective larva; **B.** *Alofia* Giglioli in Sambon, 1922, cephalon; **C.** *Leiperia* Sambon, 1922, cephalon; **D.** *Sebekia* Sambon, 1922, cephalon; **E.** *Subtriquetra rileyi* Junker, Boomker et Booyse, 1998, cephalon; **F.** *Alofia merki* Giglioli in Sambon, 1922, entire body of female adult. (Modified from Riley 1994; Junker *et al.* 1998; Christoffersen & De Assis 2013.)

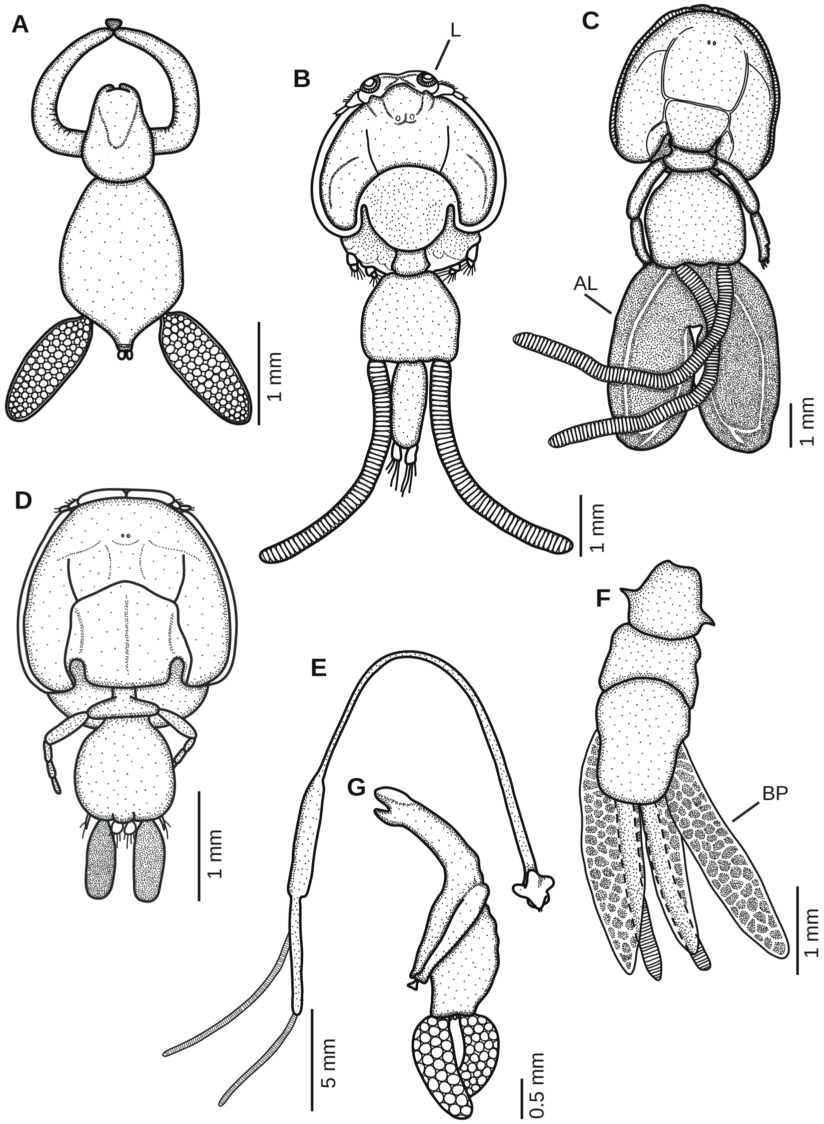

Fig. 4.9.6. Siphonostomatoida. **A.** *Achtheres micropteri* Wright, 1882; **B.** *Caligus apodus* (Brian, 1924); **C.** *Dartevellia bilobata* Brian, 1939; **D.** *Lepeophtheirus plotosi* Barnard, 1948; **E.** *Lernaeenicus neglectus* Richiardi, 1877; **F.** *Lernanthropsis mugilis* (Brian, 1898); **G.** *Parabrachiella mugilis* (Kabata, Raibaut et Ben Hassine, 1971). (Modified from Barnard 1948, 1955; Kabata *et al.* 1971; Raibaut *et al.* 1971; Radujković & Raibaut 1990; Dojiri & Ho 2013.) AL = abdominal alae; BP = bilobate processes; L = lunules.

A systematic survey of crustaceans (Crustacea) in African freshwater fishes

Keys to genera are provided where applicable. Species are listed alphabetically according to individual crustacean orders. Type species of genera and type host of species are highlighted in bold where available. When no type host is indicated, it means that no type host was originally designated from Africa, and if multiple hosts are indicated it signifies that the original description included more than one host with no specific type host designated. If known, the country where the type locality is situated is also provided. Host names are according to Froese & Pauly (2017) and *Catalog of Fishes* (Eschmeyer *et al.* 2017). Some of the species listed here include those parasitising mullets (Mugilidae) and other fish families that can cross over from marine and brackish regions into the freshwater environment.

ARGULOIDA Yamaguti, 1963 (subclass Branchiura Thorell, 1864)

Key to the genera of the Arguloida (adults) from African freshwater fishes (for a key to the species of *Argulus* see Rushton-Mellor 1994; for a key to the species of *Chonopeltis* see Fryer 1977)

1 (2) Ventral suction discs absent (stout hooks on the first maxilla) [Fig. 4.9.2E,F] ………………………………………………………………………….***Dolops***

2 (1) Ventral suction discs present (modified first maxilla)………………………3

3 (4) One pair of antennae (antennula absent); carapace anteriorly constricted; thorax elongate (approximately the same length as the carapace); retractable poison stylet absent [Fig. 4.9.2C,D]……………………..***Chonopeltis***

4 (3) Two pairs of antennae; carapace subcircular; thorax short (approximately half the length of the carapace); retractable poison stylet in front of the mouth tube [Fig. 4.9.2A,B]…………………………………………***Argulus***

List of the Arguloida (adults) from African freshwater fishes

(* indicates marine or brackish water species that might cross over into freshwater)

Argulus Müller, 1785

Argulus africanus Thiele, 1900 from *Anguilla labiata*, *Bagrus degeni*, *B. docmak*, *B. meridionalis*, *Chrysichthys brachynema*, *Clarias anguillaris*, *C. gariepinus*, ***Clarias* sp.** (Lake Malawi), *Haplochromis guiarti*, *H. obesus*, *H. obliquidens*, *H. retrodens*, *Heterobranchus bidorsalis*, *H. longifilis*, *Heterobranchus* sp., *Hydrocynus vittatus*, *Labeo* sp., *Lates microlepis*, *Mormyrops anguilloides*, *M. longirostris*, *Oreochromis esculentus*, *O. macrochir*, *O. niloticus*, *O. variabilis*, *Polypterus* sp., *Protopterus aethiopicus*, *Schilbe banguelensis*, *Schilbe* sp., *Tilapia* sp.

Argulus alexandrensis* Wilson, 1923 from ***Zeus* sp. (Angola)

Argulus amblopIites Wilson, 1920 from *Clarias gariepinus*, *Hydrocynus vittatus*, ***Parachanna obscura*** (Democratic Republic of the Congo)

Argulus angusticeps Cunnington, 1913 (Lake Tanganyika) – no hosts recorded

**Argulus arcassonensis* Cuénot, 1912 [syns *Argulus otolithi* Brian, 1927; *Argulus zei* Brian, 1924] from *Pseudotolithus typus*, *Zeus faber*

**Argulus belones* van Kampen, 1909 from *Sphyraena barracuda*

Argulus brachypeltis Fryer, 1959 from ***Hydrocynus vittatus*** (Zimbabwe)

Argulus capensis Barnard, 1955 from ***Sandelia capensis*** (South Africa)

Argulus confusus Rushton-Mellor, 1994 [this species is the male originally described as *A. amblopIites* by Wilson in 1920] – no hosts recorded

Argulus cunningtoni Fryer, 1965 from ***Auchenoglanis occidentalis***, ***Bagrus bajad***, ***Clarias gariepinus***, ***Distichodus nefasch***, ***Lates niloticus***, *Lates* sp., *Serranochromis robustus*, ***Synodontis schall*** (all Lake Albert)

**Argulus dactylopteri* Thorell, 1865 from *Dactylopterus volitans*

Argulus dageti Dollfus, 1960 from ***Clarias anguillaris***, ***Heterobranchus bidorsalis***, ***Tetraodon lineatus*** (all Mali)

Argulus dartevellei Brian, 1940 from ***Polydactylus quadrifilis*** (Angola), *Promicrops distalis*

Argulus exiguus Cunnington, 1913 from ***Lamprichthys tanganicanus***, ***Simochromis diagramma*** (both Lake Tanganyika)

Argulus fryeri Rushton-Mellor, 1994 from *Coptodon zillii*

Argulus gracilis Rushton-Mellor, 1994 from ***Auchenoglanis occidentalis*** (Lake Tanganyika)

Argulus incisus Cunnington, 1913 from ***Auchenoglanis occidentalis*** (Lake Tanganyika)

Argulus izintwala Van As et Van As, 2001 from ***Hilsa kelee*** (South Africa)

Argulus japonicus Thiele, 1900 [invasive species] [syns *Argulus matritensis* Arevalo, 1921; *Argulus pelucidus* Wagler, 1935] from *Clarias gariepinus*, *Cyprinus carpio*, *Enteromius mattozi*, *Labeo capensis*, *L. umbratus*, *Labeobarbus aeneus*, *L. kimberleyensis*, *L. marequensis*, *Oncorhynchus mykiss*, *Oreochromis mossambicus*, *Tilapia sparrmanii*

Argulus jollymani Fryer, 1956 from ***Haplochromis* sp.**, ***Protomelas fenestratus*** (both Lake Malawi)

Argulus kosus* Avenant-Oldewage, 1994 [syn. *Argulus smalei* Avenant-Oldewage et Oldewage, 1995] from *Aluterus monoceros*, *Elops machnata*, *Liza luciae*, *Mugil cephalus*, *Oreochromis mossambicus*, *Otolithes ruber*, *Pomadasys commersonnii*, *P. multimaculatus*, *Rhabdosargus holubi*, ***Sarpa salpa* (South Africa)

Argulus melita* van Beneden, 1891 from **shark (Senegal)

Argulus monodi Fryer, 1959 from *Coptodon zillii*, ***Hydrocynus vittatus*** (Zimbabwe)

Argulus multipocula Barnard, 1955 from *Chelon richardsonii* [Fig. 4.9.2A,B]

Argulus personatus Cunnington, 1913 from *Bathybates fasciatus*, ***B. ferox*** (Lake Tanganyika)

Argulus reticulatus Wilson, 1920 from ***Hydrocynus goliath*** (Democratic Republic of the Congo)

Argulus rhipidiophorus Monod, 1931 from *Alestes baremoze*, *Bagrus bajad*, *Barbus* sp., *Clarias gariepinus*, *Clarias* sp., *Haplochromis pappenheimi*, *Haplochromis* sp., *Hydrocynus forskahlii*, *H. vittatus*, ***Hydrocynus* sp.** (Lake Albert), *Labeo* sp., *Lates niloticus*, *Lates* sp., *Oreochromis leucostictus*, *O. niloticus*, *Synodontis schall*, *Tilapia* sp.

Argulus rijckmansii Brian, 1940 – no hosts recorded

Argulus rubescens Cunnington, 1913 from ***Chrysichthys brachynema*** (Lake Tanganyika)

Argulus rubropunctatus Cunnington, 1913 from *Lates angustifrons*, ***Lates microlepis*** (Lake Tanganyika)

Argulus schoutedeni Monod, 1928 from *Citharinus gibbosus*, *Distichodus fasciolatus*

Argulus striatus Cunnington, 1913 from ***Auchenoglanis occidentalis***, ***Chrysichthys brachynema***, ***Clarias gariepinus***, ***Dinotopterus cunningtoni*** (all Lake Tanganyika), *Heterobranchus* sp., *Oreochromis tanganicae*

Argulus trachynoti* Brian, 1927 from ***Trachinotus blochii* (Cameroon)

Argulus vittatus* (Rafinesque-Schmaltz, 1814) [syns *Agenor purpureus* Risso, 1826; *Argulus giganteus* Lucas, 1845; *Argulus purpureus* (Risso, 1826); *Diprosia vittata* Rafinesque, 1814] from *Boops boops*, ***Pagellus erythrinus*, *Pagrus pagrus*, *Sparus aurata*

Argulus wilsoni Brian, 1940 from ***Hydrocynus goliath*** (Democratic Republic of the Congo)

Chonopeltis Thiele, 1900

Chonopeltis australis Boxshall, 1976 from ***Labeo capensis***, ***L. rosae*** (both South Africa), *L. umbratus*, *Labeobarbus aeneus*

Chonopeltis brevis Fryer, 1961 from *Amphilius grandis*, *Amphilius* sp., *Chrysichthys nigrodigitatus*, *Garra* sp., *Labeo cylindricus*, ***L. victorianus*** (Lake Victoria), ***Labeobarbus altianalis*** (Nile River)

Chonopeltis congicus Fryer, 1959 [syn. *Chonopeltis inermis* var. *schoutedeni* Brian, 1940 *partim*] from ***Marcusenius monteiri*** (Zimbabwe)

Chonopeltis elongatus Fryer, 1974 from ***Synodontis longirostris*** (Democratic Republic of the Congo)

Chonopeltis flaccifrons Fryer, 1960 from ***Cyphomyrus discorhynchus*** (Lake Mweru), ***Hippopotamyrus wilverthi*** (Democratic Republic of the Congo), ***Marcusenius* sp.** (Malagarasi River Swamps) [Fig. 4.9.2C,D]

Chonopeltis fryeri Van As, 1986 from ***Clarias gariepinus***, ***C. theodorae*** (both South Africa)

Chonopeltis inermis Thiele, 1900 from *Bathyclarias nyasensis*, ***Chromis* sp.** (East Africa), *Clarias gariepinus*, *C. theodorae*

Chonopeltis lisikili Van As et Van As, 1996 from ***Synodontis leopardinus*** (Botswana), *S. macrostigma*, *S. nigromaculatus*, *S. thamalakanensis*, *S. vanderwaali*

Chonopeltis liversedgi Van As et Van As, 1999 from ***Mormyrus lacerda*** (Botswana)

Chonopeltis meridionalis Fryer, 1964 [syns *Chonopeltis koki* Van As, 1992; *Chonopeltis victori* Avenant-Oldewage, 1991] from *Labeo congoro*, *L. cylindricus*, ***L. rosae*** (Zimbabwe), *L. ruddi*, *Labeobarbus marequensis*

Chonopeltis minutus Fryer, 1977 [syn. *Chonopeltis australissimus* Fryer, 1977] from *Pseudobarbus burgi*, ***P. calidus***, ***P. erubescens*** (both South Africa)

Chonopeltis schoutedeni Brian, 1940 [syn. *Chonopeltis inermis* var. *schoutedeni* Brian, 1940 *partim*] from *Cyphomyrus discorhynchus*, *Marcusenius macrolepidotus*, *M. monteiri*, *Mormyrus longirostris*, *Mormyrus* sp.

Dolops Audouin, 1837

Dolops ranarum (Stuhlmann, 1892) [syn. *Gyropeltis ranarum* Stuhlmann, 1892] from *Astatoreochromis alluaudi*, *Auchenoglanis occidentalis*, *Bagrus bajad*, *B. degeni*, *B. docmak*, *Chetia flaviventris*, *Chrysichthys* sp., *Clarias anguillaris*, *C. gariepinus*, *C. stappersii, Clarias* sp., *Coptodon rendalli*, *C. zillii*, *Enteromius mattozi*, **frog tadpoles** (Lake Malawi), *Hepsetus cuvieri*, *H. odoe*, *Heterobranchus bidorsalis*, *Heterobranchus* sp., *Labeo altivelis*, *L. congoro*, *Labeobarbus marequensis*, *Lates microlepis*, *L. niloticus*, *Micropterus dolomieu*, *M. salmoides*, *Mormyrops anguilloides*, *M. longirostris*, *Oreochromis andersonii*, *O. esculentus*, *O. macrochir*, *O. mortimeri*, *O. mossambicus*, *O. niloticus*, *O. variabilis*, *Parachanna obscura*, *Protopterus aethiopicus*, *Pseudocrenilabrus philander*, *Sargochromis carlottae*, *S. codringtonii*, *S. giardi*, *Serranochromis macrocephalus, S. robustus, Serranochromis* sp., *Schilbe intermedius*, *S. mystus*, *Schilbe* sp., *Synodontis nigromaculatus*, *S. zambezensis*, *Tetraodon lineatus*, *Tilapia* sp. [Fig. 4.9.2E,F]

CYCLOPOIDA Burmeister, 1834

Key to the genera of the Cyclopoida (adults) from African freshwater fishes
(for a key to the species of *Lernaea* see Harding 1950; for a key to the species of *Lamproglena* see Fryer 1964)

1 (2) Body clearly segmented; with antennulae and antennae…………………3

2 (1) Body not clearly segmented; no antennulae or antennae…………………9

3 (4) Body elongate and cylindrical; caudal rami short or produced (extended)……………………………………………………………………5

4 (3) Body sub-cylindrical, irregularly swollen; caudal rami reduced; maxilla and maxilliped united laterally [Fig. 4.9.3E]…………………***Lamproglenoides***

5 (6) Body bent (twisted) up to 180° angle; thorax elongate; posterior part of the body (abdomen) abruptly thickened……………………………….............7

6 (5) Body not bent, with robust maxillipeds terminating in 1-5 claws; thorax with legs, first two segments forming a distinct short neck; abdomen not thickened, with three segments [Fig. 4.9.3D]……………………….***Lamproglena***

7 (8) Head horns dendritic (head and neck with branched appendages); two abdominal segments [Fig. 4.9.3C]……………………………….***Dysphorus***

8 (7) Head horns rounded, short and soft; no branched neck appendages; three abdominal segments [Fig. 4.9.3G]…………………………..***Lernaeogiraffa***

9 (10) Very long and narrow neck; head with short protuberances anteriorly; pair of robust maxillae terminating in a recurved spine [Fig. 4.9.3A,B]………… ……………………………………………………………...***Afrolernaea***

10 (9) Shorter neck; head with noticeable horn-like structures…………………11

11 (12) Head with 2-3 (rarely 4) large horns (pointed or swollen); body may gradually thicken towards the posterior end [Fig. 4.9.3F]……………..***Lernaea***

12 (11) Head with 4 horns, 2 posterior horns directed backwards to form 90° angle; lateral outgrowth extends from the upper neck region [Fig. 4.9.3H] ……………………………………………………………***Opistholernaea***

List of the Cyclopoida (adults) from African freshwater fishes

Afrolernaea Fryer, 1956

Afrolernaea annemari Oldewage, 1994 from ***Clarias gariepinus*** (Namibia)

Afrolernaea brevicollis Fryer, 1982 from ***Stomatorhinus corneti*** (Gabon)

Afrolernaea edi Oldewage, 1994 from ***Marcusenius macrolepidotus***, ***Mormyrops anguilloides***, ***Petrocephalus catostoma*** (all Namibia)

Afrolernaea longicollis Fryer, 1956 from *Cyphomyrus discorhynchus*, *Marcusenius macrolepidotus*, ***Mormyrops anguilloides***, ***M. longirostris*** (both Lake Malawi), *Mormyrops* sp. [Fig. 4.9.2A,B]

Afrolernaea mormyroides Van As, 1983 from ***Marcusenius macrolepidotus*** (South Africa)

Afrolernaea nigeriensis (Dollfus, 1960) [syn. *Delamarina nigeriensis* Dollfus, 1960] from ***Mormyrus rume*** (Niger River)

Dysphorus Kurtz, 1924

Dysphorus torquatus Kurtz, 1924 from ***Heterotis niloticus*** (Sudan) [Fig. 4.9.3C]

Lamproglena von Nordmann, 1832

Lamproglena angusta Wilson, 1924 from ***Malapterurus electricus*** (Nile River)

Lamproglena barbicola Fryer, 1961 from ***Labeobarbus altianalis*** (Lake Victoria)

Lamproglena clariae Fryer, 1956 from *Clarias anguillaris*, ***C. gariepinus***, *C. ngamensis*, ***Clarias* sp.** (both Lake Malawi)**,** *Lates niloticus* [Fig. 4.9.3D]

Lamproglena cleopatra Humes, 1957 from ***Labeo forskalii*** (Nile River)

Lamproglena cornuta Fryer, 1965 from *Clarias gariepinus*, ***Heterobranchus bidorsalis*** (Nile River)

Lamproglena elongata Capart, 1956 from ***Citharinus citharus*** (Sudan), *C. latus*, *Hydrocynus vittatus*

Lamproglena hemprichii von Nordmann, 1832 [syn. *Lamproglena aubentoni* Dollfus, 1960] from *Alestes dentex*, *Brycinus nurse*, *Clarotes laticeps*, *Hepsetus odoe*, *Hydrocynus brevis*, *H. forskahlii*, *H. vittatus*, ***Hydrocynus* sp.** (Nile River)

Lamproglena hepseti Van As et Van As, 2007 from ***Hepsetus cuvier*** (previously *H. odoe*) (Botswana)

Lamproglena hoi Dippenaar, Luus-Powell et Roux, 2001 from ***Labeobarbus marequensis*** (South Africa), ***L. polylepis*** (South Africa)

Lamproglena intercedens Fryer, 1964 from *Citharinus* sp.

Lamproglena monodi Capart, 1944 [syn. *Lamproglena nyasae* Fryer, 1956] from *Coptodon rendalli*, *C. zillii*, *Haplochromis bicolor*, *H. degeni*, *H. eduardii*, *H. guiarti*, *H. macrops*, *H. nubilus*, *H. pappenheimi*, *H. retrodens*, *H. schubotzi*, *H. serridens*, *H. squamipinnis*, *Haplochromis* sp., *Hemichromis bimaculatus*, *H. fasciatus*, *Oreochromis esculentus*, *O. macrochir*, *O. niloticus*, *O. variabilis*, *Orthochromis polyacanthus*, *O. stormsi*, *Pseudocrenilabrus philander*, *Pterochromis congicus*, *Sargochromis codringtonii*, *Sarotherodon galilaeus*, *Schwetzochromis neodon*, *Serranochromis macrocephalus*, *S. thumbergi*, *Thoracochromis callichromus*, *T. moeruensis*, *T. schwetzi*, *T. wingatii*, *Tylochromis labrodon*

Lamproglena werneri Zimmermann, 1922 from *Auchenoglanis occidentalis*, ***Bagrus bajad*** (Nile River)

Lamproglena wilsoni Capart, 1955 – no hosts reported

Lamproglenoides Fryer, 1964

Lamproglenoides vermiformis Fryer, 1964 from ***Labeo cylindricus*** ("African Eastern Rivers") [Fig. 4.9.3E]

Lernaea Linnaeus, 1758

Lernaea bagri Harding, 1950 from ***Bagrus meridionalis*** (Lake Malawi)

Lernaea barbicola Leigh-Sharpe, 1930 from ***Barbus* sp.** (South Africa), *Malapterurus electricus*

Lernaea barilius Harding, 1950 from ***Opsaridium microlepis*** (Lake Malawi), *Raiamas steindachneri*

Lernaea barnimiana (Hartmann, 1865) [syns *Lernaea temnocephala* (Cunnington, 1914); *Lernaeocera barnimii* Hartmann, 1870; *Lernaeocera temnocephala* Cunnington, 1914] from *Coptodon zillii*, *Haplochromis nubilus*, *Labeo altivelis*, *L. capensis*, *L. congoro*, *L. cylindricus*, *L. forskalii*, *L. rosae*, *L. umbratus*, *L. victorianus*, *Labeobarbus aeneus*, *L. altianalis*, *L. bynni*, *L. intermedius*, *L. marequensis*, *Lates niloticus*, *Oreochromis esculentus*, *O. leucostictus*, *O. macrochir*, *O. mossambicus*, *O. niloticus*, *O. variabilis*, *Oreochromis* sp., *Tylochromis mylodon*, *Tylochromis* sp.

Lernaea bistricornis Harding, 1950 from *Boulengerochromis microlepis*, *Callochromis pleurospilus*, ***Cardiopharynx schoutedeni*** (Lake Tanganyika), *Cyathopharynx furcifer*

Lernaea composita Wilson, 1924 [syns *Lernaea werneri* (Kurtz, 1922); *Lernaeocera werneri* Kurtz, 1922] from *Clarias gariepinus*, *Distichodus nefasch*, ***Malapterurus electricus*** (Sudan)

Lernaea cyprinacea Linnaeus, 1758 [invasive species] [syns *Lernaea carassii* Tidd, 1933; *Lernaea elegans* Leigh-Sharpe, 1925; *Lernaea esocina* (Burmeister, 1835); *Lernaea ranae* Stunkard et Cable, 1931; *Lernaea tentaculis* Linnaeus, 1746; *Lernaea tentaculis quatuor* Linnaeus, 1746; *Lernaeocera cyprinacea* (Linnaeus, 1746); *Lernaeocera gasterostei* Bruhl, 1860] from *Bagrus docmak*, *Coptodon rendalli*, *Labeo altivelis*, *L. capensis*, *L. congoro*, *L. cylindricus*, *L. rosae*, *L. ruddi*, *Labeobarbus kimberleyensis*, *L. marequensis*, *Oreochromis aureus*, *O. mossambicus*, *O. niloticus*, *O. placidus*, *Pseudocrenilabrus philander*, *Tilapia* sp. [Fig. 4.9.3F]

Lernaea diceracephala (Cunnington, 1914) [syn. *Lernaeocera diceracephala* Cunnington, 1914] from ***Clarias gariepinus*** (Lake Tanganyika), *Heterobranchus longifilis*

Lernaea haplocephala (Cunnington, 1914) [syns *Lernaea bichiri* (Kurtz, 1923); *Lernaeocera bichiri* Kurtz, 1923; *Lernaeocera haplocephala* Cunnington, 1914] from *Labeobarbus aeneus*, ***Polypterus bichir*** (White Nile), ***P. congicus*** (Lake Tanganyika), ***P. senegalus*** (White Nile)

Lernaea hardingi Fryer, 1956 [syn. *Lernaea* sp. cf. *lophiara* Harding, 1950] from *Brycinus nurse*, *Chrysichthys mabusi*, *Coptodon zillii*, ***Nyassachromis serenus***, *Oreochromis niloticus*, ***Rhamphochromis lucius*** (both Lake Malawi), *Sargochromis mellandi*, *Sarotherodon galilaeus*, *Synodontis nigromaculatus*

Lernaea inflata Fryer, 1961 from ***Enteromius argenteus*** (Victoria Nile)

Lernaea lophiara Harding, 1950 from *Copadichromis quadrimaculatus*, ***Coptodon zillii***, ***Diplotaxodon argenteus***, *Labeo cylindricus*, *Labeobarbus johnstonii*, ***Lethrinops lethrinus***, ***L. micrentodon***, *Mylochromis incola*, ***Nyassachromis breviceps***, *N. nigritaeniatus*, ***N. prostoma***, *Otopharynx argyrosoma*, *Placidochromis johnstoni*, ***Rhamphochromis lucius***, ***Taeniolethrinops praeorbitalis***, ***Tropheops tropheops*** (all Lake Malawi)

Lernaea palati Harding, 1950 from ***Copadichromis chrysonotus*** (Lake Malawi)

Lernaea senegali (Zimmermann, 1922) [syn. *Lernaeocera senegali* Zimmermann, 1922] from ***Polypterus senegalus*** (Sudan)

Lernaea tilapiae Harding, 1950 from ***Oreochromis lidole***, ***O. squamipinnis*** (both Lake Malawi)

Lernaea tuberosa Harding, 1950 from ***Engraulicypris sardella*** (Lake Malawi)

Lernaeogiraffa Zimmermann, 1922

Lernaeogiraffa heterotidicola Zimmermann, 1922 from ***Heterotis niloticus*** (Nile River) [Fig. 4.9.3G]

Opistholernaea Yin, 1960

Opistholernaea contorta Fryer, 1965 from ***Distichodus brevipinnis***, ***D. rostratus*** (both Niger River)

Opistholernaea laterobrachialis (Fryer, 1959) [syn. *Lernaea laterobrachialis* Fryer, 1959] from *Oreochromis andersonii*, ***O. macrochir*** (Zambia), *O. niloticus* [Fig. 4.9.3H]

Opistholernaea longa (Harding, 1950) [syn. *Lernaea longa* Harding, 1950] from *Lates angustifrons*, *L. microlepis*, ***L. niloticus*** (Lake Turkana)

ISOPODA Latreille, 1817

List of the Isopoda (adults) from African freshwater fishes

Ichthyoxenos Herklots, 1870

Ichthyoxenos africana (Lincoln, 1972) from ***Lepidiolamprologus attenuatus***, ***L. elongatus*** (both Lake Tanganyika)

Ichthyoxenos expansus Van Name, 1920 from ***Eugnathichthys eetveldii*** (Democratic Republic of the Congo)

Ichthyoxenos tanganyikae (Fryer, 1965) from ***Simochromis diagramma*** (Lake Tanganyika) [Fig. 4.9.4A]

POECILOSTOMATOIDA Thorell, 1859

Key to the genera of the Poecilostomatoida (adults) from African freshwater fishes (for a key to the species of Ergasilidae see Oldewage & Van As 1988a)

1 (2) Body form elongate…………………………………………………………….3

2 (1) Body generally short and teardrop-shaped………………………………….7

3 (4) Thorax 6-segmented, segments 3-4 always fused and enlarged, with two pairs of divergent lateral lobes in form of “X” [Fig. 4.9.4B]………………… ………………………………………………………………….***Colobomatus***

4 (3) Thorax without segmentation; long, slender neck with no appendages; three pairs of biramous legs on abdomen [Fig. 4.9.4G]……….. ***Mugilicola***

5 (6) Thick cuticular covering on second antennae [Fig. 4.9.4C]………………… ……………………………………………………………..***Dermoergasilus***

6 (5) No cuticular covering on second antennae…………………………………7

7 (8) Terminal segment of second antennae smooth and subdivided into three pointed processes [Fig. 4.9.4H]……………………………***Paraergasilus***

8 (7) Terminal segment of second antennae sclerotised and with a single point [Fig. 4.9.4E,F]……………………………………………………***Ergasilus***

List of the Poecilostomatoida (adults) from African freshwater fishes

(* indicates marine or brackish water species that might cross over into freshwater)

Colobomatus Hesse, 1873

**Colobomatus mugilis* Raibaut, Caillet et Ben Hassine, 1978 from *Chelon aurata*, *C. labrosus*, *C. ramada*, *C. saliens* [Fig. 4.9.4B]

Dermoergasilus Ho et Do, 1982

Dermoergasilus mugilis* Oldewage et Van As, 1988 from *Chelon richardsonii*, ***Mugil cephalus* (South Africa), *Pseudomyxus capensis* [Fig. 4.9.4C,D]

Ergasilus von Nordmann, 1832

Ergasilus cunningtoni Capart, 1944 from *Brycinus leuciscus*, *B. nurse*, *Campylomormyrus elephas*, *Distichodus atroventralis*, *D. rostratus*, *Enteromius macrops*, *Hippopotamyrus psittacus*, *Hydrocynus forskahlii*, *Marcusenius greshoffii*, *M. moorii*, *Mormyrops anguilloides*, *M. macrophthalmus*, *M. nigricans*, *Pellonula leonensis*, *Petrocephalus grandoculis*, *Phago loricatus*, *Pollimyrus isidori*, *Pterochromis congicus*, *Raiamas senegalensis*, *Schilbe laticeps*, *Synodontis nigriventris*, *Tylochromis lateralis*, *T. microdon*

Ergasilus flaccidus Fryer, 1965 from ***Oreochromis tanganicae*** (Lake Tanganyika)

Ergasilus ilani* Oldewage et Van As, 1988 from ***Mugil cephalus* (South Africa) [Fig. 4.9.4E,F]

Ergasilus inflatipes Cressey in Cressey et Collette, 1970 from ***Strongylura senegalensis*** (Ghana)

Ergasilus kandti van Douwe, 1912 from *Bagrus bajad*, *Citharinus citharus*, *Lamprologus lemairii*, *Lates niloticus*, *Limnotilapia dardennii*, *Oreochromis tanganicae*, *Plecodus paradoxus*, *Pseudosimochromis curvifrons*, *Pterochromis congicus*, *Synodontis membranaceus*, *Tilapia* sp., *Tylochromis bangwelensis*, *T. mylodon*, *T. polylepis*

Ergasilus lamellifer Fryer, 1961 from *Astatoreochromis alluaudi*, *Haplochromis bicolor*, *H. degeni*, *H. guiarti*, *H. longirostris*, *H. nuchisquamulatus*, *H. obesus*, *H. obliquidens*, *H. retrodens*, ***Haplochromis* sp.** (Uganda), *Parailia pellucida*

Ergasilus latus Fryer, 1960 from *Auchenoglanis occidentalis*, *Chrysichthys nigrodigitatus*, *Coptodon guineensis*, *C. zillii*, ***Oreochromis niloticus***, *Pelmatolapia cabrae*, ***Sarotherodon galilaeus*** (both Lake Turkana), *S. melanotheron*, *S. nigripinnis*, *Schilbe mystus*

**Ergasilus lizae* Krøyer, 1863 [syn. *Ergasilus nanus* van Beneden, 1870] from *Alosa fallax*, *Barbus barbus*, *Chelon ramada*, *C. saliens*, *Mugil cephalus*, *Solea solea*

Ergasilus macrodactylus (Sars, 1909) [syn. *Ergasiloides macrodactylus* Sars, 1909] from *Brycinus imberi, Haplochromis* sp., *Lethrinops* sp., *Pseudotropheus* sp., *Tilapia* sp.

Ergasilus megacheir (Sars, 1909) [syn. *Ergasiloides megacheir* Sars, 1909] from *Bathybates fasciatus*, *B. minor*, *Cyphotilapia frontosa*, *Haplotaxodon microlepis*, *Limnotilapia dardennii*, *Plecodus paradoxus*, *Pterochromis congicus*, *Simochromis* sp., *Synodontis granulosus*, *S. multipunctatus*

Ergasilus mirabilis Oldewage et Van As, 1987 from *Brycinus imberi*, *Clarias gariepinus, C. ngamensis, Cyphomyrus discorhynchus*, *Enteromius afrohamiltoni*, *Glossogobius giuris*, *Hemichromis elongatus*, *Hepsetus odoe*, *Hydrocynus vittatus, Labeo rosae*, *Marcusenius macrolepidotus*, *Petrocephalus catostoma*, *Schilbe intermedius*, *S. mystus*, ***Synodontis leopardinus*** (South Africa), *S. macrostigma*, *S. nigromaculatus*, *S. zambezensis*

Ergasilus nodosus Wilson, 1924 from ***Bagrus bajad*** (Sudan)*, Bagrus* sp., *Brycinus leuciscus*, *B. nurse*, *Campylomormyrus elephas*, *Distichodus atroventralis*, *Hippopotamyrus psittacus*, *Pellonula leonensis*, *Petrocephalus grandoculis*, *Phago loricatus*, *Pterochromis congicus*, *Synodontis nigriventris*, *Tylochromis lateralis*, *T. microdon*

Ergasilus sarsi Capart, 1944 from *Brycinus imberi*, *Clarias anguillaris*, *C. gariepinus*, *C. ngamensis*, *Heterobranchus bidorsalis, Lamprichthys tanganicanus*, *Marcusenius macrolepidotus*, *Pseudotropheus* sp., *Synodontis nigromaculatus*, *Thoracochromis moeruensis*, *Tylochromis bangwelensis*, *T. microdon*, *T. mylodon*

Ergasilus sieboldi von Nordmann, 1832 [syns *Ergasilus baikalensis* Messjatzeff, 1928; *Ergasilus depressus* Sars, 1862; *Ergasilus esocis* Sumpf, 1871; *Ergasilus hoferi* Borodin, 1915; *Ergasilus surbecki* Baumann, 1912; *Ergasilus trisetoceus* von Nordmann, 1832] from *Chelon aurata*, *C. ramada*, *C. saliens*, *Mugil cephalus*

Mugilicola Tripathi, 1960

Mugilicola smithae* Jones et Hine, 1978 from ***Anguilla mossambica* (South Africa), *Crenimugil seheli*, *Planiliza alata*, *P. macrolepis*, *Pseudomyxus capensis* [Fig. 4.9.4G]

Paraergasilus Markevich, 1937

Paraergasilus lagoonaris Paperna, 1969 from ***Aplocheilichthys* sp.**, ***Awaous lateristriga***, ***Coptodon guineensis***, ***Pellonula leonensis***, ***Sarotherodon melanotheron***, ***Sierrathrissa leonensis*** (all Ghana)

Paraergasilus minutus (Fryer, 1956) [syn. *Trigasilus minutus* Fryer, 1956] from ***Petrotilapia tridentiger***, ***Tropheops tropheops*** (both Lake Malawi) [Fig. 4.9.4H]

POROCEPHALIDA Heymons, 1935

List of the Porocephalida from African freshwater fishes

Only larval Porocephalida stages are known to infect fish hosts [Fig. 4.9.5A], however, the morphological information available for the different species on this infective stage is limited. As adults are usually found in the definitive piscivorous vertebrate host (usually a reptile), life-cycle studies or molecular identification may need to be completed for accurate species identification at nymph level.

Alofia Giglioli in Sambon, 1922

Alofia sp. from *Oreochromis mossambicus* [Fig. 4.9.5B]

Leiperia Sambon, 1922

Leiperia cincinnalis (Sambon in Vaney et Sambon, 1910) [syns *Porocephalus nematoides* Beauchamp, 1918; *Reighardia cincinnalis* Sambon in Vaney et Sambon, 1910] from *Alestes macrophthalmus*, *Bathybates ferox*, *Chrysichthys brachynema*, *C. mabusi*, *Coptodon rendalli*, *Lates microlepis*, *L. niloticus*, *Mastacembelus* sp., *Oreochromis mossambicus*, *O. niloticus*, *Sargochromis giardi*, *Serranochromis meridianus* [Fig. 4.9.5C]

Leiperia gracilis (Diesing, 1836) [syns *Leiperia neotropica* Heymons et Vitzthum, 1935; *Pentastoma gracile* Diesing, 1836; *Porocephalus crocodili* Wheeler, 1915] from *Alestes macrophthalmus*, *Chrysichthys mabusi*

Sebekia Sambon, 1922

Sebekia minor (Wedl, 1861) [syns *Pentastoma oxycephalum minor* Wedl, 1861; *Sebekia wedli* Giglioli in Sambon, 1922] from *Coptodon rendalli*, *Marcusenius macrolepidotus*, *Oreochromis mossambicus*, *Poecilia reticulata*

Sebekia okavangoensis Riley et Huchzermeyer, 1995 from *Clarias gariepinus* [Fig. 4.9.5D]

Subtriquetra Sambon, 1922

Subtriquetra rileyi Junker, Boomker et Booyse, 1998 from ***Coptodon rendalli***, ***Oreochromis mossambicus*** (type host not explicitly mentioned; South Africa) [Fig. 4.9.5A,E]

SIPHONOSTOMATOIDA Thorell, 1859

Key to the genera of the Siphonostomatoida (adults) from African freshwater fishes

1 (2) Caudal ramus and posterior processes absent……………………………3

2 (1) Caudal ramus or posterior processes present……………………………5

3 (4) Conical abdomen; lunules present; short and stout egg sacs; genital process absent [Fig. 4.9.6A]……………………………………….***Achtheres***

4 (3) Large abdominal alae (wing-like structures); lunules absent; long and slender egg-sacs; 4th somite free and without dorsal plates [Fig. 4.9.6C]……………………………………………………***Dartevellia***

5 (6) Body elongate and cylindrical ……………………………………………7

6 (5) Body oval, round or irregularly shaped; lacking dorsal plates……………..9

7 (8) Body not clearly segmented, with long thoracic neck; legs 1-2 uniramous, legs 3-4 biramous [Fig. 4.9.6E]……………………………..***Lernaeenicus***

8 (7) Body segmented; distinct groove between the cephalothorax and trunk; pits and grooves absent; biramous antennae [Fig. 4.9.6G]…………………
…………………………………………………………….***Parabrachiella***

9 (10) Lunules present; head and first three thoracic segments fused (4th free) [Fig. 4.9.6B]…………………………………………………………....***Caligus***

10 (9) Frontal lunules absent……………………………………………………...11

11 (12) Head and first three thoracic segments fused (4th free); antennula 2-segmented, antenna 3-segmented; 4th leg uniramous [Fig. 4.9.6D]………… ……………………………………………………………***Lepeophtheirus***

12 (11) Antennula 9-segmented, antenna 4-segmented; 4th leg with a pair of long, bilobate processes attached to posterolateral corner of trunk [Fig. 4.9.6F] ……………………………………………………………***Lernanthropsis***

List of the Siphonostomatoida (adults) from African freshwater fishes

(* indicates marine or brackish water species that might cross over into freshwater)

Achtheres von Nordmann, 1832

Achtheres micropteri Wright, 1882 from *Micropterus salmoides* [Fig. 4.9.6A]

Caligus Müller, 1785

**Caligus apodus* (Brian, 1924) [syns *Pseudocaligus apodus* Brian, 1924; *Pseudolepeophtheirus mediterraneus* Paperna, 1964] from *Chelon aurata*, *C. labrosus*, *C. ramada*, *C. saliens*, *Mugil cephalus* [Fig. 4.9.6B]

Caligus engraulidis* Barnard, 1948 from *Chelon tricuspidens*, *Mugil cephalus*, ***Stolephorus holodon* (South Africa)

**Caligus mugilis* Brian, 1935 [syns *Caligus minimus mugilis* Brian, 1935; *Caligus minimus* var. *mugilis* Brian, 1935] from *Chelon labrosus*

Caligus pageti* Russell, 1925 [syn. *Caligus argilasi* Brian, 1931] from *Chelon aurata,* ***C. ramada* (Egypt), *C. saliens*, *Mugil cephalus*

**Caligus pharaonis* von Nordmann, 1832 [syns *Lepeophtheirus pharaonis* (von Nordmann, 1832); *Sciaenophilus inopinus* Humes, 1957; *Sciaenophilus pharaonis* (von Nordmann, 1832)] – no hosts reported

Dartevellia Brian, 1939

Dartevellia bilobata Brian, 1939 from ***Arius* sp.** (Democratic Republic of the Congo) [Fig. 4.9.6C]

Lepeophtheirus von Nordmann, 1832

Lepeophtheirus monacanthus Heller, 1865 from *Arius latiscutatus, Carlarius heudelotii*

Lepeophtheirus plotosi Barnard, 1948 from ***Plotosus lineatus*** (South Africa) [Fig. 4.9.6D]

Lernaeenicus Le Sueur, 1824

**Lernaeenicus neglectus* Richiardi, 1877 from *Chelon ramada*, *C. saliens* [Fig. 4.9.6E]

Lernanthropsis Ho et Do, 1985

Lernanthropsis mugilis (Brian, 1898) [syn. *Lernanthropus mugilis* Brian, 1898] from *Chelon aurata* [Fig. 4.9.6F]

Parabrachiella Wilson, 1915

Parabrachiella mugilis (Kabata, Raibaut et Ben Hassine, 1971) [syns *Eubrachiella mugilis* Kabata, Raibaut et Ben Hassine, 1971; *Neobrachiella mugilis* (Kabata, Raibaut et Ben Hassine, 1971)] from *Chelon aurata*, *C. saliens* [Fig. 4.9.6G]

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Chapter 4.10.

HIRUDINEA

Kerry HADFIELD & Nico SMIT

Leeches (Hirudinea) – basic characteristics, life cycles, classification and principal diagnostic features

- phylum Annelida, class Clitellata (segmented worms)
- approximately 680 species globally (480 freshwater)
- cylindrical body
- posterior and anterior disc-shaped suckers
- 34 body segments (constant and do not correspond with the number of rings/annuli observed externally as is seen in other annelids)
- segments divided into rings or annuli (the number of annuli per segment in the middle of the body is taxon-specific)
- number of annuli per segment become progressively reduced towards either end of the body
- each segment with a single, transverse row of sensory structures called sensillae
- no internal skeleton
- often with elaborate colour patterns or brightly coloured
- hermaphrodites
- gaseous exchange through the skin
- eyes and oculiform spots to detect movement
- intermediate hosts for Digenea
- vectors for blood parasites (Haematozoa)

The subclass Hirudinea is divided into two orders based on the morphology of the proboscis and vascular system. Leeches of the order Arhynchobdellida Blanchard, 1894 have a non-protrusible muscular pharynx (with or without jaws) and a haemocoelomic system. Species of the order Rhynchobdellida Blanchard, 1894 have a protrusible proboscis and true vascular system. Sawyer (1986) comprehensively summarised leech biology, behaviour and systematics.

Leeches are hermaphroditic, having separate female and male reproductive systems in one organism. Cross-fertilisation occurs when mating leeches intertwine and spermatozoa are transferred (either by means of a protrusible penis, enclosed in hardened spermatophores, or injected into the body surface of the recipient leech). Internal fertilisation occurs followed by the production of a hardened cocoon (egg case). These cocoons are attached to a solid surface and contain all the eggs as well as a fluid to provide sustenance and energy to the eggs as they grow. Most leeches abandon the cocoons at this stage, but the glossiphoniid leeches show parental care during egg development and after the juveniles have hatched. Once the juveniles hatch from the cocoons they will seek out a potential host to feed on (Oosthuizen & Siddall 2003).

Leeches are often only temporary ectoparasites and will leave their host shortly after a blood meal. However, some leeches (particularly marine species) may spend most of their lives on a host. In large numbers some leeches can even kill their host (Cruz-Lacierda *et al.* 2000). Leeches can also act as intermediate hosts and vectors for other parasites. Hayes *et al.* (2014) noted a marine fish trypanosome from South Africa, *Trypanosoma nudigobii* Fantham, 1919, inside the marine fish host as well as in the marine leech, *Zeylanicobdella arugamensis* De Silva, 1963. Morphological and molecular techniques were used to confirm the identity of the same trypanosome in both the fish and the leech. The role of a leech as an intermediate host is also known. Recently, *Helobdella adiastola* Ringuelet, 1972, *Helobdella triserialis* (Blanchard, 1849), *Haementeria eichhorniae* Ringuelet, 1978, and *Haementeria* sp. were noted as secondary intermediate hosts for the digenean, *Australapatemon magnacetabulum* Dubois, 1988, in Argentina (see Davies & Ostrowski de Núñez 2012).

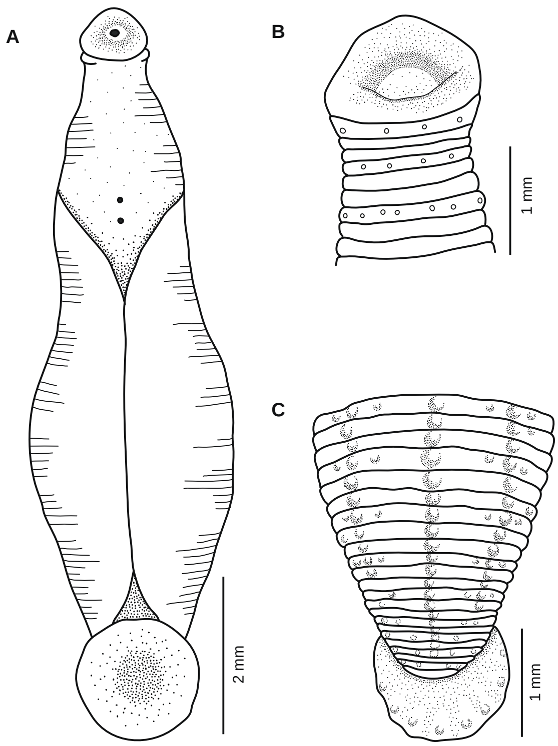

Fig. 4.10.1. Rhynchobdellida. *Batracobdelloides tricarinata* (Blanchard, 1897). **A.** Entire body; **B.** Head region; **C.** Posterior region. (Modified from Oosthuizen 1989.)

A systematic survey of leeches (Hirudinea) in African freshwater fishes

Only a single leech species, *Batracobdelloides tricarinata* (Blanchard, 1897), is currently a confirmed parasite of freshwater fishes from Africa (see Oosthuizen 1989) [Fig. 4.10.1]. However, two other leech species have been mentioned as possible African freshwater fish parasites but without specific hosts or localities recorded. *Hirudo michaelseni* Augener, 1936 (syn. *Aliolimnatis michaelseni* Sawyer, 1986) prefers mammalian hosts but according to Oosthuizen & Curtis (1990) also feeds on fishes, amphibians and snails. *Asiaticobdella buntonensis* (Meyer, 1951) (syn. *Limnatis buntonensis* Meyer, 1951) prefers hippopotami but according to Oosthuizen (1991) could possibly feed on freshwater fishes. These two species are excluded from the list of confirmed ectoparasites of African fishes. Fish names are according to Froese & Pauly (2017) and Eschmeyer *et al.* (2017).

RHYNCHOBDELLIDA Blanchard, 1894

List of the Rhynchobdellida (adults) from African freshwater fishes

Batracobdelloides Oosthuizen in Sawyer, 1986

Batracobdelloides tricarinata (Blanchard, 1897) [syns *Helobdella tricarinata* Blanchard, 1897; *Clepsine nilotica* Johansson, 1909; *Dundjibdella dartevellei* Sciacchitano, 1939; *Dundjibdella plurilineata* Sciacchitano, 1939; *Dundjibdella rubra* Sciacchitano, 1939; *Dundjibdella trilineata* Sciacchitano, 1939; *Dundjibdella triserialis* Sciacchitano, 1939; *Batrachobdella amnicola* Moore, 1958] from *Carassius auratus*, *Clarias gariepinus*, *Labeobarbus kimberleyensis*, *Oreochromis mossambicus*

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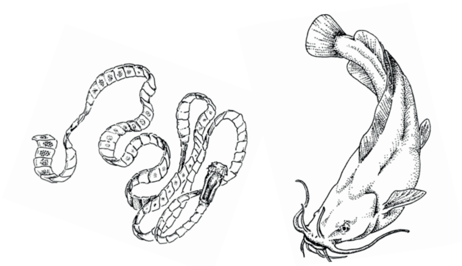
PART 5

HOST-PARASITE LIST



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Principal groups of parasites are indicated by the following abbreviations: Protista – Pr, Myxozoa – Mx, Monogenea – Mo, Trematoda – Tr, Cestoda – Ce, Acanthocephala – Ac, Nematoda – Ne, Crustacea – Cr. Larval stages are indicated by [L]. Accidental host records are indicated by (?) following the parasite name. Fish names follow Froese & Pauly (2017). Orders and families of fishes are listed alphabetically similarly as parasites of principal groups.

ORDER ANGUILLIFORMES

FAMILY ANGUILLIDAE

Anguilla anguilla (Linnaeus): Pr – *Trichodinella epizootica*, Tr – *Deropristis inflata*, *Nicolla gallica*, *Plagioporus niloticus*, Ce – *Bothriocephalus claviceps*, Ne – *Cucullanus egyptae*, *Rhabdochona* (*Globochona*) *paski*, *Rhabdochona* sp.

Anguilla labiata (Peters): Cr – *Argulus africanus*

Anguilla marmorata Quoy et Gaimard: Ne – *Hysterothylacium anguillae*

Anguilla mossambica (Peters): Pr – *Ichthyophthirius multifiliis*, Ne – *Anguillicoloides papernai*, Ne – *Contracaecum* sp. [L], *Heliconema africanum*, *Paraquimperia africana*, Cr – *Mugilicola smithae*

***Anguilla* sp.**: *Anguillicoloides crassus*, *Procamallanus* (*P.*) *armatus*

ORDER BELONIFORMES

FAMILY BELONIDAE

Strongylura senegalensis (Valenciennes): Cr – *Ergasilus inflatipes*

ORDER CHARACIFORMES

FAMILY ALESTIDAE

Alestes baremoze (Joannis): Mo – *Afrodiplozoon polycotyleus*, *Annulotrema curvipenis*, *A. elongata*, *A. longipenis*, *Characidotrema brevipenis*, *Paradiplozoon ghanense*, Tr – Cryptogonimidae gen. sp. [L], Ne – *Amplicaecum* sp. (type I) [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Argulus rhipidiophorus*

Alestes dentex (Linnaeus): Mx – *Myxobolus nyongana*, Mo – *Annulotrema elongata*, *Characidotrema nursei*, Ac – *Paragorgorhynchus albertianus*, Ne – *Amplicaecum* sp. (type I) [L], *Rhabdochona* (*Globochona*) *paski*, *Spinitectus allaeri*, Cr – *Lamproglena hemprichii*

Alestes macrolepidotus Valenciennes: Ne – *Spinitectus* spp.

Alestes macrophthalmus Günther: Ne – *Rhabdochona* (*Globochona*) *paski*, Cr – *Leiperia cincinnalis* [L], *L. gracilis* [L]

Brycinus imberi (Peters): Mo – *Annulotrema alestesimberi*, *A. allogravis*, *Characidotrema ruahae*, Ne – *Contracaecum* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Ergasilus macrodactylus*, *E. mirabilis*, *E. sarsi*

Brycinus jacksonii (Boulenger): Mo – *Annulotrema gravis*, *Characidotrema elongata*, *C. nzoiae*

Brycinus kingsleyae (Günther): Mo – *Annulotrema bouixi*, *A. combesi*, *A. maillardi*, *A. noyongensis*, *Characidotrema regia*

Brycinus lateralis (Boulenger): Pr – *Trichodina centrostrigeata*, *T. kwando*

Brycinus leuciscus (Günther): Mo – *Annulotrema robusta*, *Characidotrema elongata*, *C. nursei*, Cr – *Ergasilus cunningtoni*, *E. nodosus*

Brycinus longipinnis (Günther): Mx – *Myxobolus kribiensis*, Mo – *Annulotrema kribiensis*, *A. lamberti*, *A. moanko*

Brycinus macrolepidotus Valenciennes: Mx – *Myxobolus sangei*, Mo – *Annulotrema alberti*, *A. elongata*, *A. helicocirra*, *A. longipenis*, *A. tenuicirra*, *Paradiplozoon aegyptense*, *P. ghanense*, Tr – *Allocreadium voltanum*, Ne – *Contracaecum* sp. [L], *Orientatractis brycini*, *Rhabdochona* sp. [L]

Brycinus nurse Rüppell: Mo – *Afrogyrodactylus girgifae*, *Annulotrema alestesnursi*, *A. delta*, *A. gravis*, *A. spiropenis*, *Characidotrema brevipenis*, *C. nursei*, *C. spinivaginus*, *C. undifera, C. zelotes*, Ce – *Lytocestus marcuseni*, Ne – *Amplicaecum* sp. (type I) [L], *Contracaecum* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Ergasilus cunningtoni*, *E. nodosus*, *Lamproglena hemprichii*, *Lernaea hardingi*

Hemigrammopetersius pulcher (Boulenger): Mo – *Annulotrema amieti*, *A. gabrioni*, *Characidotrema spiropenis*

Hepsetus cuvieri (Castelnau): Pr – *Hemitrichodina robusta*, *Trichodina magna*, Mo – *Annulotrema hepseti*, Cr – *Dolops ranarum*

Hydrocynus brevis (Günther): Mo – *Annulotrema besalis*, *A. longipenis*, *A. nili*, *A. uncata*, Tr – *Siphodera ghanensis*, Ac – *Tenuisentis niloticus*, Ne – *Amplicaecum* sp. (type I) [L], *Contracaecum* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena hemprichii*

Hydrocynus forskahlii (Cuvier): Pr – *Trichodina heterodentata*, Mx – *Myxobolus hydrocyni*, *M. perforata*, Mo – *Annulotrema ansatum*, *A. besalis*, *A. bipatens*, *A. cryptophallus*, *A. cucullatum*, *A. curvipenis*, *A. gracilis*, *A. hydrocynusi*, *A. longipenis*, *A. magnihamula*, *A. nili*, *A. pikei*, *A. pontile*, *A. spiropenis*, Tr – *Dinurus gizae*, Didymozoidae gen. sp. [L], Ac – *Neoechinorhynchus africanus*, *Paragorgorhynchus albertianus*, Ne – *Afrophilometra hydrocyoni*, *Amplicaecum* sp. (type I) [L], *Contracaecum* sp. [L], *Rhabdochona* (*Globochona*) *paski*, *Rhabdochona* sp., *Spinitectus* spp., Cr – *Argulus rhipidiophorus*, *Ergasilus cunningtoni*, *Lamproglena hemprichii*

Hydrocynus goliath (Boulenger): Cr – *Argulus reticulatus*, *A. wilsoni*

Hydrocynus vittatus Castelnau: Mo – *Annulotrema bracteatum*, *A. longipenis*, *A. magna*, *A. nili*, *A. pikei*, *A. pikoides*, *A. pseudonili*, *A. ruahae*, Ce – *Schyzocotyle acheilognathi*, Ne – *Afrophilometra hydrocyoni*, *Amplicaecum* sp. (type I) [L], *Contracaecum* sp.

[L], *Eustrongylides* sp. [L], *Galeiceps* sp. [L], *Paracamallanus cyathopharynx*, *Spinitectus* spp., Cr – *Argulus africanus*, *A. ambloplites*, *A. brachypeltis*, *A. monodi*, *A. rhipidiophorus*, *Ergasilus mirabilis*, *Lamproglena elongata*, *L. hemprichii*

***Hydrocynus* sp.**: Mo – *Afrocleidodiscus hydrocynuous*, Ne – *Contracaecum* sp. [L], Cr – *Argulus rhipidiophorus*, *Lamproglena hemprichii*

Micralestes acutidens (Peters): Pr – *Apiosoma constricta*, *A. dermatum*, *A. micralesti*, *Hemitrichodina robusta*, *Trichodina heterodentata*, *T. kwando*, *Trichodinella crennulata*, *Tripartiella lechridens*, Mo – *Afrogyrodactylus kingi*

Micralestes humilis Boulenger: Mo – *Annulotrema sangmelinensis*

***Micralestes* sp.**: Mo – *Afrogyrodactylus characinis*, *Annulotrema edeensis*, *Gyrodactylus micralestes*

Phenacogrammus aurantiacus (Pellegrin): Ne – *Rhabdochona* (*Globochona*) *paski*

Phenacogrammus major (Boulenger): Mo – *Annulotrema amieti*, *A. gabrioni*, *Characidotrema spiropenis*

Phenacogrammus urotaenia (Boulenger): Mo – *Characidotrema spiropenis*

Rhabdalestes maunensis (Fowler): Mx – *Thelohanellus rhabdalestus*

Rhabdalestes septentrionalis (Boulenger): Mo – *Afrogyrodactylus ardae*

FAMILY CITHARINIDAE

Citharinops distichodoides (Pellegrin): Ac – *Neoechinorhynchus africanus*

Citharinus citharus (Geoffroy Saint-Hilaire): Mx – *Henneguya logonensis*, *Myxobolus charii*, *M. mbailaoi*, *Thelohanellus citharini*, *T. lagdoensis*, *T. taguii*, Mo – *Citharodactylus gagei*, *Nanotrema citharini*, *N. niokoloensis*, Tr – *Brevicaecum niloticum*, *Clinostomum* sp. [L], Ac – *Neoechinorhynchus africanus*, *Neoechinorhynchus* sp., Ne – *Cithariniella citharini*, *Procamallanus* (*P.*) *laeviconchus*, Cr – *Ergasilus kandti*, *Lamproglena elongata*

Citharinus gibbosus (Geoffroy Saint-Hilaire): Ne – *Procamallanus* (*P.*) *laeviconchus*, Cr – *Argulus schoutedeni*

Citharinus latus Müller et Troschel: Cr – *Lamproglena elongata*

***Citharinus* sp.**: Cr – *Lamproglena intercedens*

FAMILY CTENOLUCIIDAE

Boulengerella cuvieri (Spix et Agassiz): Ne – *Contracaecum* sp. [L]

FAMILY DISTICHODONTIDAE

Distichodus atroventralis Boulenger: Cr – *Ergasilus cunningtoni*, *E. nodosus*

Distichodus brevipinnis Günther: Ne – *Cithariniella citharini*, *Falcaustra hexapapillata*, *F. sudanensis*, *F. tchadi*, *Procamallanus* (*P.*) *laeviconchus*, Cr – *Opistholernaea contorta*

Distichodus engycephalus Günther: Mx – *Myxidium distichodi*, *Myxobolus distichodi*

Distichodus fasciolatus Boulenger: Ne – *Labeonema bakeri*, Cr – *Argulus schoutedeni*

Distichodus lusosso Schilthuis: Tr – *Brientrema malapteruri*, Ne – *Falcaustra piscicola*

Distichodus nefasch (Bonnaterre): Mo – *Afrocleidodiscus paracleidodiscus*, Tr – *Sandonia sudanensis*, Ne – *Falcaustra hexapapillata*, *F. petrei*, *F. sudanensis*, *Procamallanus* (*P.*) *laeviconchus*, Cr – *Argulus cunningtoni*, *Lernaea composita*

Distichodus rostratus Günther: Mo – *Afrocleidodiscus distichodis*, Tr – *Sandonia sudanensis*, Ne – *Falcaustra hexapapillata*, *F. petrei*, *F. tchadi*, *Procamallanus* (*P.*) *laeviconchus*, Cr – *Ergasilus cunningtoni*, *Opistholernaea contorta*

Distichodus schenga Peters: Ne – *Cithariniella petterae*

Distichodus sexfasciatus Boulenger: Ne – *Labeonema bakeri*

***Distichodus* sp.**: Ne – *Falcaustra piscicola*

Eugnathichthys eetveldii Boulenger: Cr – *Ichthyoxenos expansus*

Ichthyborus besse Joannis: Ce – *Ichthybothrium ichthybori*, *Proteocephalus* sp., Ac – *Neoechinorhynchus ichthyobori*

Ichthyborus quadrilineatus (Pellegrin): Ce – *Ichthybothrium ichthybori*

Nannaethiops unitaeniatus Günther: Mo – *Annulotrema nannaethiopis*

Nannocharax multifasciatus Boulenger: Pr – *Trichodina compacta*, *T. ngoma*

Neolebias ansorgii Boulenger: Mx – *Myxidium camerounense*

Neolebias trewavasae Poll et Gosse: Mo – *Annulotrema bilongi*, *A. endjami*, *A. fomenai*

Paradistichodus dimidiatus (Pellegrin): Ne – *Cithariniella khalili*, *C. koubkovae*

Phago loricatus Günther: Cr – *Ergasilus cunningtoni*, *E. nodosus*

Xenocharax spilurus Günther: Ne – *Orientatractis brycini*, *Synodontisia thelastomoides*

FAMILY HEPSETIDAE

Hepsetus cuvieri (Castelnau): Cr – *Lamproglena hepseti*

Hepsetus odoe (Bloch): Mx – *Henneguya nkamensis*, *Myxobolus africanus*, *Sphaerospora sangmelimaensis*, Mo – *Annulotrema biaensis*, *A. hepseti*, *A. macropenis*, Ne – *Philometroides africanus*, *Procamallanus* (*Spirocamallanus*) *spiralis*, Cr – *Dolops ranarum*, *Ergasilus mirabilis*, *Lamproglena hemprichii*

ORDER CLUPEIFORMES

FAMILY CLUPEIDAE

Alosa fallax (Lacepède): Cr – *Ergasilus lizae*

Hilsa kelee (Cuvier): Cr – *Argulus izintwala*

Limnothrissa miodon (Boulenger): Mo – *Ancyrocephalus limnotrissae*

Pellonula leonensis Boulenger: Mo – *Ancyrocephalus pellonulae*, Cr – *Ergasilus cunningtoni*, *E. nodosus*, *Paraergasilus lagoonaris*

Sierrathrissa leonensis Boulenger: Cr – *Paraergasilus lagoonaris*

FAMILY ENGRAULIDAE

Stolephorus holodon (Boulenger): Cr – *Caligus engraulidis*

ORDER CYPRINIFORMES

FAMILY CYPRINIDAE

Barbus barbus (Linnaeus): Cr – *Ergasilus lizae*

***Barbus* sp.**: Mx – *Myxobolus ovoidalis*, Mo – *Dactylogyrus afroruahae*, *D. barbus*, *D. gabonensis*, *D. macrocleithrum*, Tr – *Allocreadium indistinctum*, *Aspidogaster limacoides*, Ce – *Parvitaenia* sp. 2 [L], Ne – *Camallanus kirandensis*, *Rhabdochona gendrei*, Cr – *Lernaea barbicola*

***Barilius* sp.**: Mo – *Ancyrocephalus barilii*

Carasobarbus fritschii (Günther): Mo – *Dactylogyrus kulindrii*, *D. marocanus*, *D. oumiensis*, *D. volutus*, *D. zatensis*, Ce – *Schyzocotyle acheilognathi*

Carasobarbus harterti (Günther): Mo – *Dactylogyrus marocanus*, *D. oumiensis*

Carasobarbus moulouyensis (Pellegrin): Mo – *Dactylogyrus fimbriphallus*, *D. ksibioides*

Carassius auratus (Linnaeus): Pr – *Chilodonella hexasticha*, *Dermocystidium* sp., *Ichthyophthirius multifiliis*, *Ichthyophthirius* sp., *Trichodina mutabilis*, *T. reticulata*, *T. uniforma*, Hi – *Batracobdelloides tricarinata*

Chagunius nicholsi (Myers): Ce – *Ligula intestinalis* [L]

Ctenopharyngodon idella (Valenciennes): Pr – *Trichodinella epizootica*

Cyprinus carpio Linnaeus: Pr – *Chilodonella* sp., *Dermocystidium* sp., *Ichthyobodo necator*, *Ichthyobodo* sp., *Ichthyophthirius multifiliis*, *Tetrahymena pyriformis*, *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *T. nobilis*, *T. reticulata*, *Trichodina* sp., *Trichodinella epizootica*, *Trichodinella* sp., *Tripartiella lechridens*, Mx – *Myxobolus ovoidalis*, Tr – *Clinostomum tilapiae* [L], *Tylodelphys* sp. [L], Ce – *Atractolytocestus huronensis*, *Cyclustera* sp. [L], *Neogryporhynchus lasiopeius* [L], *Schyzocotyle acheilognathi*, Ne – *Contracaecum* sp. [L], Cr – *Argulus japonicus*

Engraulicypris sardella (Günther): Cr – *Lernaea tuberosa*

Enteromius ablabes (Bleeker): Mo – *Dactylogyrus afrobarbae*, *D. pokoase*

Enteromius afrohamiltoni (Crass): Cr – *Ergasilus mirabilis*

Enteromius annectens (Gilchrist et Thompson): Ce – *Schyzocotyle acheilognathi*

Enteromius anoplus (Weber): Pr – *Goussia anopli*

Enteromius apleurogramma (Boulenger): Mo – *Dactylogyrus nanocirrus*, *D. parviphallus*

Enteromius argenteus Günther: Ce – *Schyzocotyle acheilognathi*, Cr – *Lernaea inflata*

Enteromius aspilus (Boulenger): Mx – *Chloromyxum birgii*, *Myxidium nyongense*, *Myxobolus fobobi*, *M. nyongana*, *M. oloi*, *Thelohanellus valeti*, Mo – *Dactylogyrus aspili*, *D. mendehei*, *D. nyongensis*

Enteromius bifrenatus (Fowler): Ce – *Schyzocotyle acheilognathi*

Enteromius brevipinnis (Jubb): Ce – *Schyzocotyle acheilognathi*

Enteromius callipterus (Boulenger): Mx – *Myxobolus ngassami*, *M. sanagaensis*, *M. sessabai*

Enteromius camptacanthus (Bleeker): Mx – *Myxidium nyongense*, *Myxobolus fobobi*, *M. njinei*, *M. nyongana*, *M. oloi*, Mo – *Dactylogyrus amieti*, *D. njinei*, *D. valeti*, *Dogielius njinei*, Tr – *Allocreadium mazoensis*, Ne – *Rhabdochona gendrei*

Enteromius cercops (Whitehead): Mo – *Afrodiplozoon polycotyleus*

Enteromius eutaenia (Boulenger): Pr – *Trichodina compacta*, *T. heterodentata*, *Tripartiella macrosoma*, Ne – *Rhabdochona* (*Globochona*) *gambiana*

Enteromius fasciolatus (Günther): Pr – *Trichodina kalimbeza*

Enteromius guirali (Thominot): Mx – *Myxidium mendehi*, *M. nyongense*, *Myxobolus fobobi*, *M. njinei*, *M. nyongana*, *M. oloi*, Mo – *Dactylogyrus mendehei*, *D. nyongensis*

Enteromius holotaenia (Boulenger): Mo – *Gyrodactylus ivindoensis*

Enteromius humilis (Boulenger): Tr – *Apatemon barbusi* [L], *Diplostomum longicollis* [L], *D. montanum* [L], *Diplostomum* sp. [L], *Posthodiplostomum nanum* [L], Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L]

Enteromius jae (Boulenger): Mx – *Myxidium nyongense*, *Myxobolus fobobi*, *M. nkolyaensis*, *M. nyongana*, *Thelohanellus valeti*, Mo – *Dactylogyrus biradius*, *D. jaei*, *D. kii*

Enteromius kamolondoensis (Poll): Ce – *Ligula intestinalis* [L]

Enteromius kerstenii (Peters): Mo – *Afrodiplozoon polycotyleus*, Mo – *Dactylogyrus afrolongicornis*, *D. afropsilovaginus*, *D. afrotoxopous*, *D. brevicirrus*, *D. brevicornis*, *D. longionchus*, *D. longiphallus*, *D. parviphallus*

Enteromius lineomaculatus (Boulenger): Ce – *Ligula intestinalis* [L], Ne – *Rhabdochona esseniae*, *R. gendrei*

Enteromius lukusiensis (David et Poll): Ce – *Ligula intestinalis* [L]

Enteromius macrops (Boulenger): Mo – *Dactylogyrus mawli*, Ce – Parvitaenia sp. 2 [L], Cr – *Ergasilus cunningtoni*

Enteromius magdalenae (Boulenger): Mo – *Dactylogyrus afrosclerovaginus*

Enteromius martorelli (Roman): Mx – *Chloromyxum birgii*, *Myxidium mendehi*, *M. nyongense*, *Myxobolus fobobi*, *M. njinei*, *M. nyongana*, *M. oloi*, Mo – *Dactylogyrus birgii*, *D. bopeleti*, *D. insolitus*, *D. maillardi*, *Dogielius martorellii*

Enteromius mattozi (Guimarães): Ce – *Schyzocotyle acheilognathi*, Ne – *Contracaecum* sp. [L], Cr – *Argulus japonicus*, *Dolops ranarum*

Enteromius miolepis (Boulenger): Ne – *Rhabdochona* (*R.*) *centroafricana*

Enteromius multilineatus (Worthington): Tr – *Clinostomum complanatum* [L]

Enteromius neefi (Greenwood): Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus enidae*, Ac – *Acanthogyrus phillipi*

Enteromius neglectus (Boulenger): Mo – *Dactylogyrus afrofluviatilis*, *D. afrosclerovaginus*, *D. brevicirrus*, *D. nanocirrus*

Enteromius neumayeri (Fischer): Mo – *Afrodiplozoon polycotyleus*

Enteromius nyanzae (Whitehead): Mo – *Dactylogyrus clavatovaginus*, *D. spinicirrus*

Enteromius paludinosus (Peters): Pr – *Apiosoma phiala*, *A. piscicola*, *Chilodonella hexasticha*, *Cryptobia* sp., *Epistylis* sp., *Ichthyophthirius multifiliis*, *Tetrahymena pyriformis*, *Trichodina heterodentata*, *T. kazubski*, *T. nigra*, *Trichodina* sp., *Trichodinella epizootica*, *Trichodinella* sp., *Tripartiella lechridens*, Mx – *Myxobolus paludinosus*, Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus afrochelatus*, *D. afropsilovaginus*, *D. afrosclerovaginus*, *D. clavatovaginus*, *D. dominici*, *D. teresae*, *Dogielius intorquens*, *Paradiplozoon aegyptense*, Ce – *Ligula intestinalis* [L], *Paradilepis scolecina* [L], *Schyzocotyle acheilognathi*, *Valipora campylancristrota* [L], Ac – *Polyacanthorhynchus kenyensis* [L], Ne – *Camallanus* sp., *Contracaecum* sp. [L], *Rhabdochona esseniae*, *R. gendrei*

Enteromius perince (Rüppell): Mo – *Dactylogyrus afrofluviatilis*, *D. afrolongicornis*, *D. afropsilovaginus*, *D. afrosclerovaginus*, *D. allolongionchus*, *D. brevicirrus*, *D. myersi*, *D. nanocirrus*, *Gyrodactylus kyogae*, Ne – *Cucullanus barbi*

Enteromius pleurogramma (Boulenger): Tr – *Apatemon barbusi* [L]

Enteromius radiatus (Peters): Pr – *Trichodina compacta*, Mo – *Dactylogyrus spinicirrus*, Ce – *Ligula intestinalis* [L]

Enteromius sublineatus (Daget): Mo – *Dactylogyrus afrobarbae*, *D. nanocirrus*

Enteromius tanapelagius (Graaf, Dejen, Sibbing et Osse): Tr – *Apatemon barbusi* [L]

Enteromius thamalakanensis (Fowler): Mx – *Myxobolus etsataensis*

Enteromius treurensis (Groenewald): Ce - *Parvitaenia* sp. [L]

Enteromius trimaculatus (Peters): Pr – *Apiosoma constricta*, *A. dermatum*, *A. phiala*, *A. piscicola*, *Trichodina compacta*, *T. heterodentata*, *T. kazubski*, *T. minuta*, *T. uretra*, *Trichodinella epizootica*, *Tripartiella lechridens*, Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus afrolongicornis*, *D. allolongionchus*, *D. myersi*, *D. spinicirrus*, Tr – *Allocreadium mazoensis*, Ce – *Paradilepis scolecina* [L], *Parvitaenia* sp. 1 [L], *Parvitaenia* sp. 2 [L], *Schyzocotyle acheilognathi*, Ne – *Contracaecum* sp. [L], *Rhabdochona esseniae*, *R. gendrei*

Enteromius trispilopleura (Boulenger): Mo – *Dactylogyrus afrobarbae*

Enteromius trispilos (Bleeker): Mo – *Dactylogyrus nanocirrus*

Enteromius unitaeniatus (Günther): Pr – *Apiosoma phiala*, Ce – *Ligula intestinalis* [L], *Paradilepis scolecina* [L], Ne – *Contracaecum* sp. [L]

***Enteromius* sp.**: Pr – *Trichodina centrostrigeata*

Garra dembecha Getahun et Stiassny: Tr – *Diplostomum garrae* [L], *D. longicollis* [L], *D. montanum* [L], *Diplostomum* sp. [L], *Ichthyocotylurus* sp. [L], *Posthodiplostomum nanum* [L]

***Garra* sp. :** Cr – *Chonopettis brevis*

Labeo alluaudi (Pellegrin): Mo – *Dactylogyrus longiphalloides*, *Dogielius kabaensis*

Labeo altivelis (Peters): Ne – *Camallanus kirandensis*, *Philometroides khalili*, Cr – *Dolops ranarum*, *Lernaea barnimiana*, *L. cyprinacea*

Labeo capensis (Smith): Pr – *Apiosoma phiala*, Mo – *Dactylogyrus iwani*, *D. larindae*, *D. nicolettae*, *Dogielius intorquens*, *Paradiplozoon vaalense*, Cr – *Argulus japonicus*, *Chonopeltis australis*, *Lernaea barnimiana*, *L. cyprinacea*

Labeo congoro Peters: Mo – *Paradiplozoon krugerense*, Cr – *Chonopeltis meridionalis*, *Dolops ranarum*, *Lernaea barnimiana*, *L. cyprinacea*

Labeo coubie Rüppell: Mx – *Myxobolus burkinei*, *M. labeoi*, *Thelohanellus bicornei*, Mo – *Dactylogyrus cyclocirrus*, *D. decaspirus*, *D. dembae*, *D. digitalis*, *D. falcilocus*, *D. jaculus*, *D. labeous*, *D. leonis*, *D. oligospirophallus*, *D. retroversus*, *D. titus*, *D. yassensis*, *Dogielius anthocolpos*, *D. clavipenis*, *D. complicitus*, *D. flagellatus*, *D. harpagatus*, *D. tropicus*, *Paradiplozoon aegyptense*, Tr – *Clinostomum* sp. [L], *Nematobothrium labeonis*, *Nematobothrium* sp., Ac – *Acanthogyrus nigeriensis*

Labeo cylindricus Peters: Pr – *Apiosoma obliqua*, *A. phiala*, *A. piscicola*, *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *Tripartiella lechridens*, Mo – *Dactylogyrus brevicirrus*, *D. cyclocirrus*, *Dogielius dublicornis*, *Paradiplozoon aegyptense*, Ac – *Acanthogyrus malawiensis*, Cr – *Chonopeltis brevis*, *C. meridionalis*, *Lamproglenoides vermiformis*, *Lernaea barnimiana*, *L. cyprinacea*, *L. lophiara*

Labeo forskalii Rüppell: Mo – *Dactylogyrus brevicirrus*, *D. helicophallus*, *D. longiphallus*, *Paradiplozoon aegyptense*, Tr – *Ectenurus labeonis*, *Lecithochirium magnicaudatum*, *Nematobothrium labeonis*, Cr – *Lamproglena cleopatra*, *Lernaea barnimiana*

Labeo horie Heckel: Tr – *Nematobothrium labeonis*, Ce – *Cyclustera magna* [L], Ne – *Camallanus longicaudatus*

Labeo lukulae Boulenger: Ce – *Ligula intestinalis* [L]

Labeo molybdinus du Plessis: Ne – *Camallanus* sp.

Labeo niloticus (Linnaeus): Pr – *Apiosoma piscicola*, *Trichodina centrostrigeata*, Mx – *Myxidium shamama*, *Myxobolus imami*, *M. labiae*, *M. naffari*, *M. niloticus*, *Thelohanellus niloticus*, Tr – *Nematobothrium labeonis*, Ne – *Amplicaecum* sp. (type I) [L], *Camallanus kirandensis*, *C. longicaudatus*, *Rhabdochona* sp. [L]

Labeo parvus Boulenger: Mx – *Myxobolus kouoptamoensis*, *M. nchoutnounensis*, *M. njoyai*, *M. nyongana*, *Thelohanellus ndjamenaensis*, Mo – *Dactylogyrus brevicirrus*, *D. falcilocus*, *D. jucundus*, *D. longiphallus*, *D. omega*, *Dogielius kabaensis*, *D. parvus*, *D. rosumplicatus*

Labeo rosae (Boulenger): Mo – *Dactylogyrus pienaari*, *Paradiplozoon krugerense*, Ne – *Philometroides khalili*, Cr – *Chonopeltis australis*, *C. meridionalis*, *Ergasilus mirabilis*, *Lernaea barnimiana*, *L. cyprinacea*

Labeo rouaneti Daget: Mo – *Dactylogyrus jucundus*, *D. omega*, *D. sematus*, *Dogielius rosumplicatus*

Labeo ruddi Boulenger: Mo – *Dogielius junorstrema*, Cr – *Chonopeltis meridionalis*, *Lernaea cyprinacea*

Labeo senegalensis (Steindachner): Mx – *Thelohanellus costeae*, Mo – *Dactylogyrus cyclocirrus*, *D. labeous*, *D. rastellus*, *D. senegalensis*, *D. tubarius*, *Dogielius flosculus*, *D. tropicus*, Tr – *Nematobothrium* sp.

Labeo umbratus (Smith): Mo – *Dactylogyrus iwani*, *D. larindae*, *Dogielius intorquens*, *Paradiplozoon vaalense*, Cr – *Argulus japonicus*, *Chonopeltis australis*, *Lernaea barnimiana*

Labeo victorianus Boulenger: Pr – *Babesiosoma mariae*, Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus brachydiscus*, *D. brevicirrus*, *D. cyclocirrus*, *D. helicophallus*, *D. longiphallus*, *Paradiplozoon aegyptense*, Cr – *Chonopeltis brevis*, *Lernaea barnimiana*

***Labeo* sp.**: Mx – *Myxobolus bilongi*, *Thelohanellus assambai*, *T. sanagaensis*, Mo – *Dactylogyrus nathaliae*, *Dogielius grandijugus*, Ne – *Labeonema intermedium*, Cr – *Argulus africanus*, *A. rhipidiophorus*

Labeobarbus acutirostris (Bini): Tr – *Apatemon barbusi* [L]

Labeobarbus aeneus (Burchell): Pr – *Ichthyophthirius multifiliis*, Mo – *Paradiplozoon ichthyoxanthon*, Ce – *Schyzocotyle acheilognathi*, Cr – *Argulus japonicus*, *Chonopeltis australis*, *Lernaea barnimiana*, *L. haplocephala*

Labeobarbus altianalis (Boulenger): Mo – *Dactylogyrus brevicirrus*, *D. longiphallus*, *D. spinicirrus*, Ce – *Schyzocotyle acheilognathi*, Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Falcaustra straeleni*, *F. verbekei*, *Rhabdochona gendrei*, *R.* (*Globochona*) *paski*, Cr – *Chonopeltis brevis*, *Lamproglena barbicola*, *Lernaea barnimiana*

Labeobarbus batesii (Boulenger): Mo – *Dactylogyrus afer*

Labeobarbus beso (Rüppell): Tr – *Apatemon barbusi* [L], *Clinostomum* sp. [L], *Diplostomum montanum* [L]

Labeobarbus bynni (Forsskål): Mx – *Myxobolus caudatus*, *M. egypticus*, *M. fahmii*, *M. imami*, *M. naffari*, Mo – *Dactylogyrus aferoides*, *D. pseudanchoratus*, *D. sahelensis*, *Dogielius djolibaensis*, Tr – *Allocreadium aswanensis*, *A. sudanensis*, *Aspidogaster africanus*, Ce – *Khawia armeniaca*, *Khawia* sp., *Schyzocotyle acheilognathi*, Ne – *Cucullanus barbi*, *C. djilorensis*, *Rhabdochona gendrei*, *R.* (*Globochona*) *paski*, *Rhabdochona* sp., Cr – *Lernaea barnimiana*

Labeobarbus dainellii (Bini): Tr – *Apatemon barbusi* [L]

Labeobarbus gorgorensis (Bini): Tr – *Diplostomum montanum* [L]

Labeobarbus gorguari (Rüppell): Tr – *Apatemon barbusi* [L]

Labeobarbus intermedius (Rüppell): Tr – *Apatemon barbusi* [L], Ne – *Falcaustra straeleni*, *F. verbekei*, *Rhabdochona gendrei*, Cr – *Lernaea barnimiana*

Labeobarbus johnstonii (Boulenger): Cr – *Lernaea lophiara*

Labeobarbus kimberleyensis (Gilchrist et Thompson): Pr – *Trichodina compacta*, Mo – *Dactylogyrus varicorhini*, Ce – *Paradilepis scolecina* [L], *Schyzocotyle acheilognathi*, Cr – *Argulus japonicus*, *Lernaea cyprinacea*, Hi – *Batracobdelloides tricarinata*

Labeobarbus macrolepis (Pfeffer): Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus papernai*, *D. pseudanchoratus*, *D. ruahae*, *D. rufijii*, *Dogielius grandiphallus*

Labeobarbus marequensis (Smith): Pr – *Apiosoma mothlapitsis*, *A. phiala*, *Trichodina compacta*, *T. heterodentata*, Mo – *Afrodiplozoon polycotyleus*, *Dactylogyrus spinicirrus*, Tr – *Allocreadium mazoensis*, Ce – *Ligula intestinalis* [L], *Paradilepis delachauxi* [L], *Schyzocotyle acheilognathi*, Ne – *Contracaecum* sp. [L], *Rhabdochona esseniae*, *R. gendrei*, Cr – *Argulus japonicus*, *Chonopeltis meridionalis*, *Dolops ranarum*, *Lamproglena hoi*, *Lernaea barnimiana*, *L. cyprinacea*

Labeobarbus microbarbis (David et Poll): Ce – *Ligula intestinalis* [L]

Labeobarbus nedgia Rüppell: Tr – *Apatemon barbusi* [L], Ce – *Schyzocotyle acheilognathi*

Labeobarbus parawaldroni (Lévêque, Thys van den Audenaerde et Traoré): Mo – *Dactylogyrus aferoides*, *D. archaeopenis*, *D. parawaldronii*, *D. pseudanchoratus*, *D. ruahae*, *Dogielius pedaloe*

Labeobarbus petitjeani Daget: Mo – *Dactylogyrus aferoides*, *D. archaeopenis*, *D. clani*, *D. petitjeani*, *D. pseudanchoratus*, *D. sahelensis*, *Dogielius djolibaensis*

Labeobarbus polylepis (Wu): Cr – *Lamproglena hoi*

Labeobarbus reinii (Günther): Mo – *Dactylogyrus kulindrii*, *D. marocanus*, *D. oumiensis*, *D. reinii*

Labeobarbus sacratus (Daget): Mo – *Dactylogyrus archaeopenis*, *D. pseudanchoratus*, *D. ruahae*, *D. sacrati*, *Dogielius phrygieus*

Labeobarbus somereni (Boulenger): Mo – *Dactylogyrus spinicirrus*

Labeobarbus tropidolepis (Boulenger): Ce – *Khawia armeniaca*

Labeobarbus tsanensis (Nagelkerke et Sibbing): Ne – *Eustrongylides* sp. [L]

Labeobarbus wurtzi (Pellegrin): Mo – *Dactylogyrus falcilocus*, *D. pseudanchoratus*, *D. ruahae*, *D. wurtzii*, *Dogielius pedaloe*, *D. vexillus*

***Labeobarbus* sp.**: Pr – *Ichthyobodo* sp.

Leptocypris niloticus (Joannis): Mo – *Dactylogyrus brevicirrus*

Luciobarbus callensis (Valenciennes): Mo – *Dactylogyrus fimbriphallus*, *D. heteromorphus*, *D. ksibii*, *D. marocanus*, *D. tunisiensis*, Ce – *Khawia armeniaca*, *Schyzocotyle acheilognathi*

Luciobarbus maghrebensis (Doadrio, Perea et Yahyaoui): Mo – *Dactylogyrus falsiphallus*, *D. varius*

Luciobarbus magniatlantis (Pellegrin): *Dactylogyrus ksibii*

Luciobarbus moulouyensis (Pellegrin): Mo – *Dactylogyrus benhoussai*

Luciobarbus nasus Boulenger: Mo – *Dactylogyrus borjensis*, *D. marocanus*

Luciobarbus pallaryi (Pellegrin): Mo – *Dactylogyrus atlasensis*, *D. draaensis*, *D. fimbriphallus*, *D. guirensis*

Luciobarbus rifensis Doadrio, Casal-Lopez et Yahyaoui: Mo – *Dactylogyrus scorpius*

Luciobarbus setivimensis (Valenciennes): Mo – *Dactylogyrus ksibii*, *D. ksibioides*, *D. marocanus*, Ce – *Khawia armeniaca*, Ac – *Acanthogyrus maroccanus* [L]

Mesobola brevianalis (Boulenger): Pr – *Ambiphrya neobolae*, *Apiosoma caulata*, *A. phiala*, *Trichodina compacta*, *T. heterodentata*, *T. magna*, *Trichodinella epizootica*, *Tripartiella clavodonta*, *T. lechridens*

Opsaridium microlepis (Günther): Cr – *Lernaea barilius*

Pseudobarbus burgi (Boulenger): Cr – *Chonopeltis minutus*

Pseudobarbus calidus (Barnard): Cr – *Chonopeltis minutus*

Pseudobarbus erubescens (Skelton): Cr – *Chonopeltis minutus*

Raiamas christyi (Boulenger): Ne – *Rhabdochona* (*Globochona*) *tricuspidata*

Raiamas moorii (Boulenger): Ne – *Rhabdochona* (*Globochona*) *gambiana*

Raiamas senegalensis (Steindachner): Mo – *Ancyrocephalus barilii*, *Paradiplozoon aegyptense*, Cr – *Ergasilus cunningtoni*

Raiamas steindachneri (Steindachner): Cr – *Lernaea barilius*

Rastrineobola argentea (Pellegrin): Tr – *Allocreadium engraulicypridis*, Ce – *Ligula intestinalis* [L]

ORDER CYPRINODONTIFORMES

FAMILY NOTHOBRANCHIIDAE

Aphyosemion batesii (Boulenger): Mo – *Dactylogyrus batesii*

Aphyosemion bivittatum (Lönnberg): Mx – *Myxidium birgii*

Aphyosemion cameronense (Boulenger): Mo – *Cichlidogyrus amieti*, Ne – *Prosungulonema campanae*, *Rhabdochona* (*Rhabdochona*) *moraveci*

Aphyosemion exiguum (Boulenger): Mo – *Cichlidogyrus amieti*

Epiplatys multifasciatus (Boulenger): Ne – *Rhabdochona* (*Globochona*) sp.

Epiplatys sexfasciatus Gill: Tr – *Posthodiplostomoides leonensis* [L], *P. nanum* [L]

Epiplatys spilargyreius (Duméril): Tr – *Posthodiplostomoides leonensis* [L], *P. nanum* [L]

***Epiplatys* sp.**: Mo – *Gyrodactylus cyprinodonti*, Tr – *Clinostomum* sp. [L]

Nothobranchius furzeri Jubb: Tr – *Apatemon* sp. [L], *Emoleptalea* sp., Ne – Camalanidae gen. sp. [L], Gnathostomatidae gen. sp. [L]

Nothobranchius kadleci Reichard: Tr – *Emoleptalea* sp., Ne – Camalanidae gen. sp. [L], Gnathostomatidae gen. sp. [L]

Nothobranchius orthonotus (Peters): Ne – Camalanidae gen. sp. [L], Gnathostomatidae gen. sp. [L]

Nothobranchius pienaari Shidlovskiy, Watters et Wildekamp: Ne – Camalanidae gen. sp. [L], Gnathostomatidae gen. sp. [L]

FAMILY POECILIIDAE

***Aplocheilichthys* sp.**: Cr – *Paraergasilus lagoonaris*

Gambusia affinis (Baird et Girard): Tr – *Centrocestus cuspidatus* [L]

Lamprichthys tanganicanus (Boulenger): Cr – *Argulus exiguus*, *Ergasilus sarsi*

Micropanchax johnstoni (Günther): Pr – *Trichodina compacta*, *T. heterodentata*, *T. magna*

Poecilia reticulata Peters: Pr – *Ichthyophthirius multifiliis*, Cr – *Sebekia minor* [L]

Poropanchax luxophthalmus (Brüning): Mo – *Ancyrocephalus claveaui*

Poropanchax normani (Ahl): Mo – *Gyrodactylus cytophagus*

ORDER ELOPIFORMES

FAMILY ELOPIDAE

Elops machnata (Forsskål): Cr – *Argulus kosus*

ORDER LEPIDOSIRENIFORMES

FAMILY PROTOPTERIDAE

Protopterus aethiopicus Heckel: Tr – *Heterorchis crumenifer*, Ne – *Eustrongylides* sp. [L], Cr – *Argulus africanus*, *Dolops ranarum*

Protopterus annectens (Owen): Pr – *Trichodina* sp., Tr – *Heterorchis crumenifer*, *H. protopteri*, *H. senegalensis*

***Protopterus* sp.**: Tr – *Heterorchis crumenifer*

ORDER MUGILIFORMES

FAMILY MUGILIDAE

Chelon aurata (Risso): Mx – *Myxobolus exiguous*, Cr – *Caligus apodus*, *C. pageti*, *Colobomatus mugilis*, *Ergasilus sieboldi*, *Lernanthropsis mugilis*, *Parabrachiella mugilis*

Chelon dumerili (Steindachner): Pr – *Babesiosoma hannesi*

Chelon labrosus (Risso): Cr – *Caligus apodus*, *C. mugilis*, *Colobomatus mugilis*

Chelon ramada (Risso): Tr – *Saccocoelium obesum*, Ac – *Neoechinorhynchus ichthyobori*, Cr – *Caligus apodus*, *C. pageti*, *Colobomatus mugilis*, *Ergasilus lizae*, *E. sieboldi*, *Lernaeenicus neglectus*

Chelon richardsonii (Smith): Pr – *Babesiosoma hannesi*, Cr – *Argulus multipocula*, *Dermoergasilus mugilis*

Chelon saliens (Risso): Cr – *Caligus apodus*, *C. pageti*, *Colobomatus mugilis*, *Ergasilus lizae*, *E. sieboldi*, *Lernaeenicus neglectus*, *Parabrachiella mugilis*

Chelon tricuspidens (Smith): Cr – *Caligus engraulidis*

Crenimugil seheli (Forsskål): Cr – *Mugilicola smithae*

Liza luciae (Penrith et Penrith): Cr – *Argulus kosus*

Mugil cephalus Linnaeus: Pr – *Babesiosoma hannesi*, *Desseria* sp., *Trypanosoma* cf. *mugilicola*, Mx – *Myxobolus bizerti*, *M. exiguous*, Tr – *Saccocoelium obesum*, Cr – *Argulus kosus*, *Caligus apodus*, *C. engraulidis*, *C. pageti*, *Dermoergasilus mugilis*, *Ergasilus ilani*, *E. lizae*, *E. sieboldi*

Mugil curema Valenciennes: Ne – *Cucullanus djilorensis*

Planiliza alata (Steindachner): Cr – *Mugilicola smithae*

Planiliza macrolepis (Smith): Cr – *Mugilicola smithae*

Pseudomyxus capensis (Valenciennes): Cr – *Dermoergasilus mugilis*, *Mugilicola smithae*

ORDER OSTEOGLOSSIFORMES

FAMILY ARAPAIMIDAE

Heterotis niloticus (Cuvier): Mx – *Henneguya dini*, *Myxobolus heterotisi*, *M. sourouensis*, Mo – *Heterotesia voltae*, Tr – *Lecithochirium musculus*, *Nephrocephalus bagriincapsulatus* [L], Ce – *Sandonella sandoni*, Ac – *Tenuisentis niloticus*, Ne – *Multicaecum heterotis*, Cr – *Dysphorus torquatus*, *Lernaeogiraffa heterotidicola*

FAMILY GYMNARCHIDAE

Gymnarchus niloticus Cuvier: Tr – *Acanthostomum gymnarchi*, *Phyllodistomum linguale*, *Thaparotrema piscicola*, Ce – *Nesolecithus africanus*, Ne – *Nilonema gymnarchi*, *Raphidascaroides bishaii*

FAMILY MORMYRIDAE

Brevimyrus niger (Günther): Mx – *Henneguya odzai*

Brienomyrus brachyistius (Gill): Mx – *Henneguya ntemensis*, *Myxidium brienomyri*, *Sphaerospora sangmelimaensis*, Mo – *Bouixella pusilla*

Campylomormyrus elephas (Boulenger): Cr – *Ergasilus cunningtoni*, *E. odosus*

Campylomormyrus tamandua (Günther): Ne – *Contracaecum* sp. [L], *Procamallanus* (*P.*) *laeviconchus*

Cyphomyrus discorhynchus (Peters): Cr – *Afrolernaea longicollis*, *Chonopeltis flaccifrons*, *C. schoutedeni*, *Ergasilus mirabilis*

Gnathonemus petersii (Günther): Mx – *Hoferellus gnathonemi*, Mo – *Archidiplectanum archidiplectanum*, Ac – *Megistacantha horridum*, Ne – *Contracaecum* sp. [L], Capillariidae gen. sp.

Hippopotamyrus harringtoni (Boulenger): Ce – *Lytocestus marcuseni*

Hippopotamyrus pictus (Marcusen): Ac – *Megistacantha horridum*

Hippopotamyrus psittacus (Boulenger): Cr – *Ergasilus cunningtoni*, *E. nodosus*

Hippopotamyrus wilverthi (Boulenger): Cr – *Chonopeltis flaccifrons*

Hyperopisus bebe (Lacepède): Mx – *Henneguya odzai*, Ac – *Megistacantha horridum*, Ne – *Contracaecum* sp. [L]

Marcusenius cyprinoides (Linnaeus): Ac – *Megistacantha horridum*, Ne – *Cucullanus mormyr*, *Procamallanus* (*P.*) *laeviconchus*

Marcusenius greshoffii (Schilthuis): Ne – *Rhabdochona* (*R.*) *marcusenii*, Cr – *Ergasilus cunningtoni*

Marcusenius macrolepidotus (Peters): Pr – *Apiosoma constricta*, *A. dermatum*, *A. piscicola*, *Hemitrichodina robusta*, *Trichodina compacta*, *T. heterodentata*, *T. magna*, Mo – *Mormyrogyrodactylus gemini*, Tr – *Clinostomum vandehorsti* [L], Cr – *Afrolernaea edi*, *A. longicollis*, *A. mormyroides*, *Chonopeltis schoutedeni*, *Ergasilus mirabilis*, *E. sarsi*, *Sebekia minor* [L]

Marcusenius monteiri (Günther): Cr – *Chonopeltis congicus*, *C. schoutedeni*

Marcusenius moorii (Günther): Mx – *Henneguya nyongensis*, *H. odzai*, Cr – *Ergasilus cunningtoni*

Marcusenius senegalensis (Steindachner): Mx – *Henneguya odzai*, Mo – *Bouixella furcillata*

Marcusenius stanleyanus (Boulenger): Ne – *Contracaecum* sp. [L]

***Marcusenius* sp.**: Cr – *Chonopeltis flaccifrons*

Mormyrops anguilloides (Linnaeus): Mo – *Bouixella deliciosa*, *B. torta*, Ac – *Megistacantha sanghaensis*, Ne – *Contracaecum* sp. [L], *Dujardinascaris mormyropsis*, *Procamallanus* (*P.*) *laeviconchus*, *Spinitectus mucronatus*, Cr – *Afrolernaea edi*, *A. longicollis*, *Argulus africanus*, *Dolops ranarum*, *Ergasilus cunningtoni*

Mormyrops boulengeri Pellegrin: Ne – *Spinitectus monstrosus*, *S. mucronatus*

Mormyrops longirostris Peters: Cr – *Afrolernaea longicollis*, *Argulus africanus*, *Dolops ranarum*

Mormyrops macrophthalmus Günther: Cr – *Ergasilus cunningtoni*

Mormyrops nigricans Boulenger: Cr – *Ergasilus cunningtoni*

Mormyrops rume Valenciennes: Ne – *Procamallanus* (*P.*) *laeviconchus*

Mormyrops zanclirostris (Günther): Ne – *Spinitectus mucronatus*

***Mormyrops* sp.**: Cr – *Afrolernaea longicollis*

Mormyrus caschive Linnaeus: Mx – *Henneguya mailaoensis*, *H. mormyri*, Ce – *Lytocestus filiformis*, Ne – *Cucullanus mormyr*, *Eustrongylides* sp. [L], *Procamallanus* (*P.*) *laeviconchus*, *Rhabdochona* sp. [L], *Spinitectus allaeri*, *S. mormyri*

Mormyrus kannume Forsskål: Pr – *Trichodinella epizootica*, *Tripartiella dactylodentata*, *Trypanosoma mukasai*, Tr – *Basidiodiscus ectorchis*

Mormyrus lacerda Castelnau: Cr – *Chonopeltis liversedgi*

Mormyrus longirostris Peters: Cr – *Chonopeltis schoutedeni*

Mormyrus niloticus (Bloch et Schneider): Mo – *Bouixella mormyris*

Mormyrus rume Valenciennes: Mx – *Henneguya odzai*, Mo – *Bouixella beninensis*, *B. gorei*, *B. koutouani*, *B. mormyrume*, *B. yaoi*, Ne – *Spinitectus mormyri*, Cr – *Afrolernaea nigeriensis*

***Mormyrus* sp.**: Ce – *Lytocestus filiformis*, Ne – *Cucullanus mormyr*, *Procamallanus* sp., *Pseudoproleptus africanus*, *Spinitectus thurstonae*, Cr – *Chonopeltis schoutedeni*

Petrocephalus catostoma (Günther): Pr – *Trichodina compacta*, *T. heterodentata*, *T. magna*, Cr – *Afrolernaea edi*, *Ergasilus mirabilis*

Petrocephalus grandoculis Boulenger: Cr – *Ergasilus cunningtoni*, *E. nodosus*

Petrocephalus simus Sauvage: Mx – *Myxidium petrocephali*, *Sphaerospora sangmelimaensis*

Pollimyrus isidori (Valenciennes): Cr – *Ergasilus cunningtoni*

Stomatorhinus corneti Boulenger: Cr – *Afrolernaea brevicollis*

FAMILY NOTOPTERIDAE

Papyrocranus afer (Günther): Mo – *Quadriacanthus euzeti*

FAMILY PANTODONTIDAE

Pantodon buchholzi Peters: Ne – *Gendria* sp., *Spinitectus allaeri*

ORDER PERCIFORMES

FAMILY ANABANTIDAE

Ctenopoma kingsleyae Günther: Mx – *Henneguya somahiensis*, Mo – *Heteronchocleidus adjanohouni*, *H. ctenopomae*, *H. ouemensis*, *H. tuzetae*, Tr – *Callodistomum diaphanum*, *Clinostomum* sp. [L], *Halipegus ctenopomi*, *Phyllodistomum ghanense*, Ne – *Camallanus* (*Zeylanema*) *ctenopomae*, *Quimperia lanceolata*, *Rhabdochona* (*Globochona*) *paski*

Ctenopoma multispine Peters: Pr – *Trichodina anabantidarum*, *T. labyrinthipicis*, *T. microspina*, *Tripartiella ctenopomae*

Ctenopoma muriei (Boulenger): Mo – *Gyrodactylus anabanti*, *G. ctenopomi*, *Heteronchocleidus niloticus*, *Macrogyrodactylus anabanti*, *M. ctenopomi*

Ctenopoma petherici Günther: Mx – *Henneguya pethericii*, *Myxidium petrocephali*, *Myxobolus pethericii*, Mo – *Heteronchocleidus adjanohouni*, *H. ctenopomae*, *H. ouemensis*, *H. tuzetae*

Microctenopoma intermedium (Pellegrin): Pr – *Trichodina anabantidarum*, *T. labyrinthipicis*, *T. microspina*, *Tripartiella microctenopomae*

Microctenopoma nanum (Günther): Mx – *Henneguya ctenopomae*, *Myxobolus amieti*

Sandelia capensis (Cuvier): Ne – *Contracaecum* sp. [L], Cr – *Argulus capensis*

FAMILY CARANGIDAE

Decapterus russelli (Rüppell): Ne – *Contracaecum* sp. [L]

Trachinotus blochii (Lacepède): Cr – *Argulus trachynoti*

FAMILY CENTRARCHIDAE

Micropterus dolomieu Lacepède: Pr – *Apiosoma epibranchialis*, *A. piscicola*, Cr – *Dolops ranarum*

Micropterus salmoides (Lacepède): Pr – *Trichodina heterodentata*, *T. magna*, Tr – *Tylodelphys* sp. [L], Ac – *Polyacanthorhynchus kenyensis* [L], Ne – *Contracaecum* sp. [L], *Porrocaecum* sp. [L], Cr – *Achtheres micropteri*, *Dolops ranarum*

FAMILY CHANNIDAE

Parachanna insignis (Sauvage): Ne – *Procamallanus* (*Spirocamallanus*) *parachannae*

Parachanna obscura (Günther): Pr – *Cyrilia nili*, Microsporidia gen. sp., Mx – *Myxidium distichodi*, *M. parachannae*, *M. sangei*, Mo – *Eutrianchoratus chibami*, *E. imbachi*, *E. magnus*, *E. malleus*, *E. minutus*, Cr – *Argulus ambloplites*, *Dolops ranarum*

FAMILY CICHLIDAE

Aristochromis christyi Trewavas: Ac – *Acanthogyrus tilapiae*

Astatoreochromis alluaudi Pellegrin: Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*, Mo – *Cichlidogyrus longipenis*, Cr – *Dolops ranarum*, *Ergasilus lamellifer*

Astatotilapia burtoni (Günther): Mo – *Cichlidogyrus gillardinae*

Astatotilapia desfontainii (Lacepède): Ne – *Procamallanus* (*P.*) *laeviconchus*

Astatotilapia flaviijosephi (Lortet): Mo – *Cichlidogyrus bifurcatus*

Aulonocranus dewindti (Boulenger): Mo – *Cichlidogyrus discophonum*, *C. pseudoaspiralis*

Bathybates fasciatus Boulenger: Mo – *Cichlidogyrus casuarinus*, Cr – *Argulus personatus*, *Ergasilus megacheir*

Bathybates ferox Boulenger: Cr – *Argulus personatus*, *Leiperia cincinnalis* [L]

Bathybates graueri (Steindachner): Ce – *Neogryporhynchus lasiopeius* [L]

Bathybates hornii Steindachner: Mo – *Cichlidogyrus casuarinus*

Bathybates leo Poll: Mo – *Cichlidogyrus casuarinus*

Bathybates minor Boulenger: Mo – *Cichlidogyrus casuarinus*, Cr – *Ergasilus megacheir*

Bathybates vittatus Boulenger: Mo – *Cichlidogyrus casuarinus*

Benitochromis batesii (Boulenger): Mo – *Urogyrus cichlidarum*

Benthochromis horii Takahashi: Mo – *Cichlidogyrus attenboroughi*, Ce – *Paradilepis maleki* [L]

Boulengerochromis microlepis (Boulenger): Mo – *Cichlidogyrus nshomboi*, Cr – *Lernaea bistricornis*

Callochromis pleurospilus (Boulenger): Cr – *Lernaea bistricornis*

Cardiopharynx schoutedeni Poll: Cr – *Lernaea bistricornis*

Chetia flaviventris Trewavas: Pr – *Apiosoma viridis*, *Trichodina compacta*, *T. heterodentata*, Ce – *Neogryporhynchus lasiopeius* [L], *Paradilepis delachauxi* [L], *Paradilepis* sp. [L], *Valipora minuta* [L], Ne – *Contracaecum* sp. [L], Cr – *Dolops ranarum*

Chromidotilapia guntheri (Sauvage): Mo – *Cichlidogyrus dionchus*, *C. longicirrus*, *C. tilapiae*, *Onchobdella krachii*, Tr – *Clinostomum tilapiae* [L]

Chromidotilapia kingsleyae Boulenger: Tr – *Euclinostomum heterostomum* [L]

Copadichromis chrysonotus (Boulenger): Cr – *Lernaea palati*

Copadichromis quadrimaculatus (Regan): Cr – *Lernaea lophiara*

Coptodon bakossiorum (Stiassny, Schliewen et Dominey): Mo – *Cichlidogyrus berminensis*, *C. tiberianus*

Coptodon bemini (Thys van den Audenaerde): Mo – *Cichlidogyrus berminensis*

Coptodon cameronensis (Holly): Mx – *Myxobolus israelensis*

Coptodon camerunensis (Lönnberg): Mo – *Cichlidogyrus aegypticus*, *C. anthemocolpos*, *C. arthracanthus*, *C. berradae*, *C. bilongi*, *C. cubitus*, *C. digitatus*, *C. dossoui*, *C. ergensi*, *C. microscutus*, *C. ornatus*, *C. tilapiae*, *C. yanni*

Coptodon coffea (Thys van den Audenaerde): Mo – *Cichlidogyrus arthracanthus*, *C. levequei*, *C. ouedraogoi*, *C. tiberianus*

Coptodon dageti (Thys van den Audenaerde): Mo – *Cichlidogyrus aegypticus*, *C. arthracanthus*, *C. cubitus*, *C. digitatus*, *C. flexicolpos*, *C. microscutus*, *C. ornatus*, *C. tiberianus*, *C. yanni*, *Enterogyrus coronatus*

Coptodon deckerti (Thys van den Audenaerde): Mo – *Cichlidogyrus arthracanthus*

Coptodon discolor (Günther): Mo – *Cichlidogyrus digitatus*, *C. dionchus*

Coptodon guineensis (Günther): Pr – *Neonosemoides tilapiae*, Mx – *Myxobolus agolus*, *M. brachysporus*, *M. djoudjensis*, *M. equatorialis*, *M. galilaeus*, *M. gandiolensis*, *M. israelensis*, *M. sarigi*, Mo – *Cichlidogyrus aegypticus*, *C. agnesi*, *C. amphoratus*, *C. anthemocolpos*, *C. arthracanthus*, *C. berradae*, *C. bilongi*, *C. cubitus*, *C. digitatus*, *C. dossoui*, *C. ergensi*, *C. flexicolpos*, *C. gallus*, *C. gillesi*, *C. halinus*, *C. kouassii*, *C. lagoonaris*, *C. louipaysani*, *C. microscutus*, *C. nageus*, *C. ouedraogoi*, *C. tiberianus*, *C. tilapiae*, *C. vexus*, *C. yanni*, *Enterogyrus coronatus*, *Gyrodactylus cichlidarum*, Ac – *Acanthogyrus tilapiae*, Cr – *Ergasilus latus*, *Paraergasilus lagoonaris*

Coptodon gutturosa (Stiassny, Schliewen et Dominey): Mo – *Cichlidogyrus aegypticus*, *C. arthracanthus*, *C. berminensis*, *C. tiberianus*, *C. tilapiae*

Coptodon kottae (Lönnberg): Mo – *Cichlidogyrus aegypticus*, *C. arthracanthus*, *C. ouedraogoi*, *C. tiberianus*, *C. tilapiae*

Coptodon louka (Thys van den Audenaerde): Mo – *Cichlidogyrus aegypticus*, *C. amphoratus*, *C. cubitus*, *C. digitatus*, *C. yanni*

Coptodon margaritacea (Boulenger): Mx – *Myxobolus sarigi*, *M. tilapiae*

Coptodon nyongana (Thys van den Audenaerde): Mx – *Myxobolus kainjiae*, Mo – *Enterogyrus cichlidarum*, *E. crassus*

Coptodon rendalli (Boulenger): Pr – *Apiosoma constricta*, *A. dermatum*, *A. piscicola*, *A. viridis*, *Chilodonella hexasticha*, *C. piscicola*, *Cryptobia* sp., *Hexamita* sp., *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *T. magna*, *Trichodinella epizootica*, *Tripartiella orthodens*, Mx – *Myxobolus tilapiae*, Mo – *Cichlidogyrus dossoui*, *C. papernastrema*, *C. quaestio*, *C. tiberianus*, *Gyrodactylus chitandiri*, *G. nyanzae*, Ce – *Neogryporhynchus lasiopeius* [L], *Paradilepis delachauxi* [L], *P. scolecina* [L], *Paradilepis* sp. [L], *Parvitaenia macropeos* [L], Ac – *Acanthogyrus tilapiae*, Ne – *Contracaecum* sp. [L], *Rhabdochona* sp., Cr – *Dolops ranarum*, *Lamproglena monodi*, *Leiperia cincinnalis* [L], *Lernaea cyprinacea*, *Sebekia minor* [L], *Subtriquetra rileyi* [L]

Coptodon thysi (Stiassny, Schliewen et Dominey): Mo – *Cichlidogyrus berminensis*

Coptodon walteri (Thys van den Audenaerde): Mo – *Cichlidogyrus aegypticus*, *C. arthracanthus*, *C. cubitus*, *C. digitatus*, *C. gallus*, *C. tiberianus*, *C. yanni*

Coptodon zillii (Gervais): Pr – *Apiosoma conica*, *A. piscicola*, *Chilodonella hexasticha*, *Goussia cichlidarum*, *Tetrahymena pyriformis*, *Trichodina magna*, *Trichodina* sp., *Trichodinella epizootica*, *Trichodinella* sp., *Trypanosoma cyanophilum*, *T. mansouri*, *Trypanosoma* sp., Mx – *Henneguya branchialis*, *Myxobolus dahomeyensis*, *M. dossoui*, *M. heterosporus*, *M. homeosporus*, *M. polycentropsi*, *M. tilapiae*, *M. zillii*, Mo – *Cichlidogyrus aegypticus*, *C. anthemocolpos*, *C. arthracanthus*, *C. cirratus*, *C. cubitus*, *C. digitatus*, *C. dionchus*, *C. ergensi*, *C. gallus*, *C. ornatus*, *C. sclerosus*, *C. tiberianus*, *C. tilapiae*, *C. vexus*, *C. yanni*, *Enterogyrus cichlidarum*, *Gyrodactylus malalai*, *G. cichlidarum*, *Scutogyrus longicornis*, Tr – *Clinostomum complanatum* [L], *C. tilapiae* [L], *Clinostomum* sp. [L], *Euclinostomum heterostomum* [L], *Posthodiplostomum nanum* [L], *Tylodelphys* sp. [L], Ce – *Amirthalingamia macracantha* [L], *Cyclustera magna* [L], Ac – *Acanthogyrus tilapiae*, *Polyacanthorhynchus kenyensis* [L], Ne – *Amplicaecum* sp. (type I) [L], *Amplicaecum* sp. (type II) [L], *Camallanus* sp., *Rhabdochona* (*Globochona*) *paski*, Cr – *Argulus fryeri*, *A. monodi*, *Dolops ranarum*, *Ergasilus latus*, *Lamproglena monodi*, *Lernaea barnimiana*, *L. hardingi*, *L. lophiara*

Ctenochromis horei (Günther): Mo – *Cichlidogyrus gistelincki*, *Gyrodactylus zimbae*

Ctenopharynx pictus (Trewavas): Ac – *Acanthogyrus tilapiae*

Cyathopharynx furcifer (Boulenger): Cr – *Lernaea bistricornis*

Cyphotilapia frontosa (Boulenger): Cr – *Ergasilus megacheir*

Cyprichromis microlepidotus (Poll): Mo – *Cichlidogyrus milangelnari*

Diplotaxodon argenteus (Trewavas): Cr – *Lernaea lophiara*

Eretmodus marksmithi Burgess: Mo – *Cichlidogyrus jeanloujustinei*

Genyochromis mento Trewavas: Ac – *Acanthogyrus tilapiae*

Gnathochromis pfefferi (Boulenger): Mo – *Cichlidogyrus irenae*

Haplochromis aeneocolor Greenwood: Mo – *Cichlidogyrus bifurcatus*, *C. haplochromii*

Haplochromis angustifrons Boulenger: Pr – *Pleistophora* sp., Mx – *Myxobolus kainjiae*, Mo – *Cichlidogyrus haplochromii*, Ne – *Eustrongylides* sp. [L]

Haplochromis argenteus Regan: Ne – *Eustrongylides* sp. [L]

Haplochromis astatodon Regan: Ne – *Contracaecum* sp. [L]

Haplochromis bicolor Boulenger: Mo – *Cichlidogyrus haplochromii*, Cr – *Ergasilus lamellifer*, *Lamproglena monodi*

Haplochromis cinereus (Boulenger): Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*

Haplochromis degeni (Boulenger): Mo – *Cichlidogyrus haplochromii*, Cr – *Ergasilus lamellifer*, *Lamproglena monodi*

Haplochromis eduardii Regan: Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena monodi*

Haplochromis elegans Trewavas: Pr – *Pleistophora* sp., Mx – *Myxobolus kainjiae*, Mo – *Cichlidogyrus bifurcatus*, *C. haplochromii*, *Gyrodactylus haplochromi*, Ne – *Rhabdochona* (*Globochona*) *paski*

Haplochromis graueri Boulenger: Ne – *Eustrongylides* sp. [L], *Rhabdochona* (*Globochona*) *paski*

Haplochromis guiarti (Pellegrin): Mo – *Cichlidogyrus dionchus*, *C. haplochromii*, Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], Cr – *Argulus africanus*, *Ergasilus lamellifer*, *Lamproglena monodi*

Haplochromis humilior (Boulenger): Pr – *Trypanosoma mukasai*

Haplochromis ishmaeli Boulenger: Ne – *Contracaecum* sp. [L], *Rhabdochona* (*Globochona*) *paski*

Haplochromis labiatus Trewavas: Ne – *Eustrongylides* sp. [L]

Haplochromis limax Trewavas: Mo – *Cichlidogyrus bifurcatus*, *C. haplochromii*

Haplochromis longirostris (Hilgendorf): Mo – *Cichlidogyrus dionchus*, *C. haplochromii*, *C. hurstonae*, Cr – *Ergasilus lamellifer*

Haplochromis macrognathus Regan: Mo – *Cichlidogyrus haplochromii*, *C. tilapiae*

Haplochromis macrops (Boulenger): Mo – *Cichlidogyrus haplochromii*, Cr – *Lamproglena monodi*

Haplochromis mahagiensis David et Poll: Ne – *Contracaecum* sp. [L]

Haplochromis nigripinnis Regan: Mo – *Cichlidogyrus haplochromii*, Ne – *Rhabdochona* (*Globochona*) *paski*

Haplochromis nubilus (Boulenger): Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*, Mo – *Cichlidogyrus haplochromii*, Ne – *Eustrongylides* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena monodi*, *Lernaea barnimiana*

Haplochromis nuchisquamulatus (Hilgendorf): Cr – *Ergasilus lamellifer*

Haplochromis obesus (Boulenger): Mo – *Cichlidogyrus dionchus*, *C. haplochromii*, Cr – *Argulus africanus*, *Ergasilus lamellifer*

Haplochromis obliquidens (Hilgendorf): Mo – *Cichlidogyrus dionchus*, *C. haplochromii*, Tr – *Clinostomum* sp. [L], Cr – *Argulus africanus*, *Ergasilus lamellifer*

Haplochromis pappenheimi (Boulenger): Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], Cr – *Argulus rhipidiophorus*, *Lamproglena monodi*

Haplochromis paucidens Regan: Ne – *Contracaecum* sp. [L]

Haplochromis petronius Greenwood: Mo – *Cichlidogyrus haplochromii*

Haplochromis placodus Poll et Damas: Ne – *Contracaecum* sp. [L]

Haplochromis retrodens (Hilgendorf): Mo – *Cichlidogyrus dionchus*, *C. haplochromii*, Cr – *Argulus africanus*, *Ergasilus lamellifer*, *Lamproglena monodi*

Haplochromis schubotzi Boulenger: Mo – *Cichlidogyrus haplochromii*, Cr – *Lamproglena monodi*

Haplochromis serranus (Pfeffer): Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*

Haplochromis serridens Regan: Ne – *Contracaecum* sp. [L], Ne – *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena monodi*

Haplochromis squamipinnis Regan: Mo – *Cichlidogyrus bifurcatus*, *C. haplochromii*, Ac – *Acanthogyrus tilapiae*, Ne – *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena monodi*

Haplochromis teegelaari Greenwood et Barel: Tr – *Allocreadium mazoensis*

***Haplochromis* sp.**: Mo – *Cichlidogyrus sclerosus*, Ce – *Ligula intestinalis* [L], Ac – *Acanthogyrus tilapiae*, Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Argulus jollymani*, *A. rhipidiophorus*, *Ergasilus lamellifer*, *E. macrodactylus*, *Lamproglena monodi*

Haplotaxodon microlepis Boulenger: Cr – *Ergasilus megacheir*

Hemibates stenosoma (Boulenger): Mo – *Cichlidogyrus casuarinus*

Hemichromis bimaculatus Gill: Mo – *Cichlidogyrus bychowskii*, *Gyrodactylus cichlidarum*, *Onchobdella pterigyalis*, *O. spirocirra*, *O. voltensis*, Ac – *Acanthogyrus tilapiae*, Cr – *Lamproglena monodi*

Hemichromis elongatus (Guichenot): Pr – *Trichodina centrostrigeata*, *T. linyanta*, *T. minuta*, Cr – *Ergasilus mirabilis*

Hemichromis fasciatus Peters: Mx – *Myxobolus agolus*, *M. dossoui*, *M. heterosporus*, Mo – *Cichlidogyrus bychowskii*, *C. dageti*, *C. dionchus*, *C. euzeti*, *C. falcifer*, *C. longicirrus*, *C. sanseoi*, *C. teugelsi*, *C. tilapiae*, *Enterogyrus melenensis*, *Gyrodactylus cichlidarum*, *Onchobdella aframae*, *O. bopeleti*, *O. voltensis*, Tr – *Clinostomum tilapiae* [L], *Clinostomum* sp. [L], *Posthodiplostomum nanum* [L], Ce – *Anomotaenia* sp. [L], Ac – *Acanthogyrus tilapiae*, Cr – *Lamproglena monodi*

Hemichromis letourneuxi Sauvage: Mo – *Cichlidogyrus dracolemma*, Ce – *Parvitaenia macropeos* [L]

Heterotilapia buttikoferi (Hubrecht): Mo – *Cichlidogyrus bonhommei*, *C. nuniezi*, *C. slembroucki*

Heterotilapia cessiana (Thys van den Audenaerde): Mo – *Cichlidogyrus nuniezi*

Interochromis loocki (Poll): Mo – *Cichlidogyrus buescheri*, *C. schreyenbrichardorum*, *C. vealli*

Konia eisentrauti (Trewavas): Mo – *Enterogyrus barombiensis*

Labeotropheus fuelleborni Ahl: Ac – *Acanthogyrus tilapiae*

Labeotropheus trewavasae Fryer: Tr – *Astiotrema turneri*

Lamprologus lemairii Boulenger: Cr – *Ergasilus kandti*

Lepidiolamprologus attenuatus (Steindachner): Cr – *Ichthyoxenos africana*

Lepidiolamprologus cunningtoni (Boulenger): Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L]

Lepidiolamprologus elongatus (Boulenger): Cr – *Ichthyoxenos africana*

Lethrinops lethrinus (Günther): Cr – *Lernaea lophiara*

Lethrinops micrentodon (Regan): Cr – *Lernaea lophiara*

***Lethrinops* sp.**: Cr – *Ergasilus macrodactylus*

Lichnochromis acuticeps Trewavas: Ac – *Acanthogyrus tilapiae*

Limnotilapia dardennii (Boulenger): Mo – *Cichlidogyrus steenbergei*, Cr – *Ergasilus kandti*, *E. megacheir*

Maylandia emmiltos (Stauffer, Bowers, Kellogg et McKaye): Ac – *Acanthogyrus tilapiae*, Tr – *Astiotrema turneri*

Maylandia zebra (Boulenger): Tr – *Astiotrema turneri*, Ac – *Acanthogyrus tilapiae*

Mchenga thinos (Stauffer, LoVullo et McKaye): Ac – *Acanthogyrus tilapiae*

Melanochromis auratus (Boulenger): Ac – *Acanthogyrus tilapiae*

Melanochromis heterochromis Bowers et Stauffer: Ac – *Acanthogyrus tilapiae*

Melanochromis vermivorus Trewavas: Tr – *Astiotrema turneri*, Ac – *Acanthogyrus tilapiae*

Mylochromis incola (Trewavas): Cr – *Lernaea lophiara*

Nimbochromis polystigma (Regan): Ac – *Acanthogyrus tilapiae*

Nyassachromis breviceps (Regan): Cr – *Lernaea lophiara*

Nyassachromis nigritaeniatus (Trewavas): Cr – *Lernaea lophiara*

Nyassachromis prostoma (Trewavas): Cr – *Lernaea lophiara*

Nyassachromis serenus (Trewavas): Cr – *Lernaea hardingi*

Ophthalmotilapia boops (Boulenger): Mo – *Cichlidogyrus centesimus*, *C. makasai*, *C. vandekerkhovei*

Ophthalmotilapia nasuta (Poll et Matthes): Mo – *Cichlidogyrus aspiralis*, *C. centesimus*, *C. glacicremoratus*, *C. makasai*, *C. rectangulus*, *C. sturmbaueri*, *C. vandekerkhovei,* Ce – *Valipora minuta* [L]

Ophthalmotilapia ventralis (Boulenger): Mo – *Cichlidogyrus centesimus*, *C. makasai*, *C. sturmbaueri*, *C. vandekerkhovei*

Oreochromis andersonii (Castelnau): Pr – *Trichodina centrostrigeata*, *T. compacta*, *T. linyanta*, *T. magna*, *Trypanosoma mukasai*, Ac – *Acanthogyrus tilapiae*, Ne – *Contracaecum* sp. [L], Cr – *Dolops ranarum*, *Opistholernaea laterobrachialis*

Oreochromis aureus (Steindachner): Pr – *Goussia cichlidarum*, Mo – *Cichlidogyrus bifurcatus*, *C. tilapiae*, *Gyrodactylus cichlidarum*, *Scutogyrus longicornis*, Cr – *Lernaea cyprinacea*

Oreochromis esculentus (Graham): Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*, Mx – *Myxobolus brachysporus*, *M. homeosporus*, Mo – *Cichlidogyrus cirratus*, *C. halli*, *C. thurstonae*, *Quadriacanthus tilapiae*, Ac – *Acanthogyrus tilapiae*, Cr – *Argulus africanus*, *Dolops ranarum*, *Lamproglena monodi*, *Lernaea barnimiana*

Oreochromis leucostictus (Trewavas): Pr – *Epistylis* sp., *Tetrahymena pyriformis*, *Trichodina* sp., *Trichodinella* sp., Mo – *Cichlidogyrus halli*, *C. haplochromii*, *C. sclerosus*, *C. tilapiae*, *Scutogyrus gravivaginus*, Tr – *Clinostomum* sp. [L], *Tylodelphys* sp. [L], Ac – *Acanthogyrus tilapiae*, *Polyacanthorhynchus kenyensis* [L], Ne – *Contracaecum* sp. [L], Cr – *Argulus rhipidiophorus*, *Lernaea barnimiana*

Oreochromis lidole (Trewavas): Ac – *Acanthogyrus tilapiae*, Cr – *Lernaea tilapiae*

Oreochromis macrochir (Boulenger): Ce – *Paradilepis delachauxi* [L], Ac – *Acanthogyrus tilapiae*, Ne – *Contracaecum* sp. [L], Cr – *Argulus africanus*, *Dolops ranarum*, *Lamproglena monodi*, *Lernaea barnimiana*, *Opistholernaea laterobrachialis*

Oreochromis mortimeri (Trewavas): Mo – *Cichlidogyrus dossoui*, *C. halli*, *C. karibae*, *C. sclerosus*, *C. tilapiae*, *C. zambezensis*, *Scutogyrus gravivaginus*, *S. longicornis*, Cr – *Dolops ranarum*

Oreochromis mossambicus (Peters): Pr – *Apiosoma constricta*, *A. curvinucleata*, *A. dermatum*, *A. epibranchialis*, *A. phiala*, *A. piscicola*, *A. viridis*, *Chilodonella hexasticha*, *C. piscicola*, *Chilodonella* sp., *Goussia vanasi*, *Ichthyobodo necator*, *Ichthyophthirius multifiliis*, *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *T. magna*, *T. minuta*, *T. nigra*, *Trichodinella epizootica*, *Tripartiella clavodonta*, *T. lechridens*, *T. leptospina*, *T. nana*, *Trypanosoma mukasai*, Mo – *Cichlidogyrus dossoui*, *C. sclerosus*, *C. tilapiae*, *Enterogyrus cichlidarum*, *Gyrodactylus ulinganisus*, *Scutogyrus chikhii*, *S. longicornis*, Tr – *Clinostomum tilapiae* [L], *Clinostomum* sp. [L], *Diplostomum* sp. [L], *Euclinostomum heterostomum* [L], Ce – *Neogryporhynchus lasiopeius* [L], *Paradilepis scolecina* [L], *Paradilepis* sp. [L], *Parvitaenia macropeos* [L], Ne – *Contracaecum* sp. [L], Cr – *Alofia* sp. [L], *Argulus japonicus*, *A. kosus*, *Dolops ranarum*, *Leiperia cincinnalis* [L], *Lernaea barnimiana*, *L. cyprinacea*, Cr – *Sebekia minor* [L], *Subtriquetra rileyi* [L], Hi – *Batracobdelloides tricarinata*

Oreochromis mweruensis Trewavas: Mo – *Cichlidogyrus cirratus*, *C. dossoui*, *C. halli*, *C. papernastrema*, *C. sclerosus*, *C. tiberianus*, *C. tilapiae*, *Gyrodactylus nyanzae*, *Scutogyrus gravivaginus*

Oreochromis niloticus (Linnaeus): Pr – *Babesiosoma mariae*, *Chilodonella hexasticha*, *Chilodonella* sp., *Cryptobia* sp., *Epistylis* sp., *Goussia cichlidarum*, *Hexamita* sp., *Ichthyobodo* sp., *Ichthyophthirius multifiliis*, *Ichthyophthirius* sp., *Loma camerouensis*, *Paratrichodina africana*, *Trichodina centrostrigeata*, *T. heterodentata*, *T. magna, T. mutabilis*, *T. rectuncinata*, *T. reticulata*, *Trichodina* sp., *Trypanosoma mukasai*, Microsporidia gen. sp., Mx – *Henneguya suprabranchiae*, *Myxobolus agolus*, *M. brachysporus*, *M. branchiophilus*, *M. camerounensis*, *M. dahomeyensis*, *M. equatorialis*, *M. fomenai*, *M. fotoi*, *M. galilaeus*, *M. heterosporus*, *M. homeosporus*, *M. israelensis*, *M. kainjiae*, *M. nilei*, *M. saintlouisiensis*, *M. sarigi*, *M. tilapiae*, *Ortholinea africanus*, *Sphaerospora melenensis*, *S. tilapiae*, *Thelohanellus valeti*, *Triangula egyptica*, *Zschokkella nilei*, Mo – *Cichlidogyrus aegypticus*, *C. arthracanthus*, *C. cirratus*, *C. halli*, *C. mbirizei*, *C. rognoni*, *C. sclerosus*, *C. thurstonae*, *C. tilapiae*, *Enterogyrus cichlidarum*, *E. malmbergi*, *Gyrodactylus ergensi*, *G. hildae*, *G. malalai*, *G. nyanzae*, *G. occupatus*, *G. parisellei*, *G. yacatli*, *Scutogyrus longicornis*, *S. minus*, Tr – *Apatemon tilapiae* [L], *Clinostomum complanatum* [L], *C. macrosomum* [L], *C. tilapiae* [L], *Clinostomum* sp. [L], *Diplostomum magnicaudum* [L], *D. tilapiae* [L], *Echinochasmus* *liliputans* [L], *Euclinostomum ardeolae* [L], *Haplorchis* sp. [L], *Heterophyes aequalis* [L], *H. heterophyes* [L], *Plagioporus niloticus*, *Prohemistomum vivax* [L], *Pygidiopsis genata* [L], *Stellantchasmus pseudocirratus* [L], *Stictodora sawakinensis* [L], Ce – *Amirthalingamia macracantha* [L], *Anomotaenia* sp. [L], *Cyclustera magna* [L], *Neogryporhynchus lasiopeius* [L], *Parvitaenia macropeos* [L], Ac – *Acanthocephalus lucii*, *Acanthogyrus tilapiae*, *Polyacanthorhynchus kenyensis* [L], Ne – *Amplicaecum* sp. (type I) [L], *Amplicaecum* sp. (type II) [L], *Aplectana chamaeleonis*, *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Argulus africanus*, *A. rhipidiophorus*, *Dolops ranarum*, *Ergasilus latus*, *Lamproglena monodi*, *Leiperia cincinnalis* [L], *Lernaea barnimiana*, *L. cyprinacea*, *L. hardingi*, *Opistholernaea laterobrachialis*

Oreochromis placidus (Trewavas): Cr – *Lernaea cyprinacea*

Oreochromis shiranus Boulenger: Mo – *Cichlidogyrus halli*

Oreochromis spilurus (Günther): Mo – *Cichlidogyrus halli*, *C. sclerosus*, *C. tilapiae*

Oreochromis squamipinnis (Günther): Cr – *Lernaea tilapiae*

Oreochromis tanganicae (Günther): Mo – *Cichlidogyrus halli*, *C. mbirizei*, *Scutogyrus gravivaginus*, Ac – *Acanthogyrus tilapiae*, Cr – *Argulus striatus*, *Ergasilus flaccidus*, *E. kandti*

Oreochromis urolepis (Norman): Mo – *Cichlidogyrus tilapiae*

Oreochromis variabilis (Boulenger): Pr – *Babesiosoma mariae*, *Trypanosoma mukasai*, Mx – *Myxobolus brachysporus*, *M. homeosporus*, Mo – *Cichlidogyrus cirratus*, *C. halli*, *C. thurstonae*, *C. tilapiae*, *Gyrodactylus nyanzae*, *Scutogyrus gravivaginus*, Cr – *Argulus africanus*, *Dolops ranarum*, *Lamproglena monodi*, *Lernaea barnimiana*

***Oreochromis* sp.**: Pr – *Goussia cichlidarum*, Tr – *Clinostomum* sp. [L], Ac – *Acanthogyrus tilapiae*, Cr – *Lernaea barnimiana*

Orthochromis katumbii: Mo – *Cichlidogyrus consobrini*

Orthochromis polyacanthus (Boulenger): Cr – *Lamproglena monodi*

Orthochromis stormsi (Boulenger): Cr – *Lamproglena monodi*

Otopharynx argyrosoma (Regan): Cr – *Lernaea lophiara*

Parananochromis caudifasciatus (Boulenger): Mo – *Urogyrus cichlidarum*

Paratilapia polleni Bleeker: Mo – *Insulacleidus paratilapiae*

***Parectodus* sp.**: Ce – *Lytocestoides* sp.

Paretroplus polyactis Bleeker: Mo – *Insulacleidus paretropli*

Pelmatochromis buettikoferi (Steindachner): Mo – *Cichlidogyrus arfii*, *Onchobdella melissa*, *O. sylverai*

Pelmatolapia cabrae (Boulenger): Mo – *Cichlidogyrus berradae*, *C. legendrei*, *C. lemoallei*, *C. reversati*, Cr – *Ergasilus latus*

Pelmatolapia mariae (Boulenger): Mo – *Cichlidogyrus digitatus*, *C. ergensi*, *C. flexicolpos*, *C. lemoallei*, *C. ouedraogoi*, *C. slembroucki*, *C. testificatus*, *C. tiberianus*, *C. tilapiae*, *Scutogyrus vanhovei*, Tr – *Clinostomum tilapiae* [L]

Petrotilapia genalutea Marsh: Ac – *Acanthogyrus tilapiae*

Petrotilapia tridentiger Trewavas: Cr – *Paraergasilus minutus*

Pharyngochromis acuticeps (Steindachner): Mo – *Gyrodactylus occupatus*, Ce – *Amirthalingamia macracantha* [L], *Paradilepis delachauxi* [L], *Paradilepis* sp. [L]

Pharyngochromis darlingi (Boulenger): Pr – *Trichodina compacta*, Mo – *Cichlidogyrus haplochromii*

Placidochromis johnstoni (Günther): Ac – *Acanthogyrus tilapiae*, Cr – *Lernaea lophiara*

Plecodus paradoxus Boulenger: Cr – *Ergasilus kandti*, *E. megacheir*

Protomelas annectens (Regan): Ac – *Acanthogyrus tilapiae*

Protomelas fenestratus (Trewavas): Cr – *Argulus jollymani*

Protomelas taeniolatus (Trewavas): Ac – *Acanthogyrus tilapiae*

Pseudocrenilabrus multicolor (Schöller): Mo – *Cichlidogyrus bifurcatus*

Pseudocrenilabrus philander (Weber): Pr – *Apiosoma constricta*, *A. dermatum*, *A. epibranchialis*, *A nasalis*, *A. iphiala*, *A. piscicola*, *A. viridis*, *Chilodonella hexasticha*, *C. piscicola*, *Epistylis transvaalensis*, *Goussia vanasi*, *Ichthyobodo necator*, *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *T. magna*, *T. minuta*, *T. nigra*, *Trichodinella epizootica*, *Tripartiella clavodonta*, Mo – *Cichlidogyrus philander*, *Enterogyrus cichlidarum*, *E. coronatus*, *Gyrodactylus chitandiri*, *G. occupatus*, *G. parisellei*, *G. sturmbaueri*, *G. thlapi*, *G. yacatli*, Tr – *Panamphistomum benoiti*, *Paralecithobotrys africanus*, *Trematobrien haplochromios*, Diplostomidae gen. sp. [L], Ce – *Amirthalingamia macracantha* [L], *Neogryporhynchus lasiopeius* [L], *Paradilepis maleki* [L], *P. scolecina* [L], *Paradilepis* sp. [L], *Parvitaenia samfyia* [L], *Parvitaenia* sp. 3 [L], *Valipora campylancristrota* [L], *V. minuta* [L], Ne – *Contracaecum* sp. [L], Cr – *Dolops ranarum*, *Lamproglena monodi*, *Lernaea cyprinacea*

Pseudosimochromis babaulti (Pellegrin): Mo – *Cichlidogyrus georgesmertensi*

Pseudosimochromis curvifrons (Poll): Mo – *Cichlidogyrus frankwillemsi*, *C. franswittei*, Cr – *Ergasilus kandti*

Pseudosimochromis marginatus (Poll): Mo – *Cichlidogyrus franswittei*

Pseudotropheus elongatus Fryer: Ac – *Acanthogyrus tilapiae*

***Pseudotropheus* sp.**: Cr – *Ergasilus macrodactylus*, *E. sarsi*

Pterochromis congicus (Boulenger): Cr – *Ergasilus cunningtoni*, *E. kandti*, *E. megacheir*, *E. nodosus*, *Lamproglena monodi*

Ptychochromis grandidieri Sauvage: Ce – *Valipora minuta* [L]

Ptychochromis oligacanthus (Bleeker): Mo – *Insulacleidus ptychochromidis*

Ptychochromoides betsileanus (Boulenger): Ne – *Falcaustra therezieni*

Pungu maclareni (Trewavas): Mo – *Enterogyrus barombiensis*, *Urogyrus cichlidarum*

Rhamphochromis lucius Ahl: Cr – *Lernaea hardingi*, *L. lophiara*

Sargochromis carlottae (Boulenger): Ne – *Contracaecum* sp. [L], Cr – *Dolops ranarum*

Sargochromis codringtonii (Boulenger): Mo – *Cichlidogyrus karibae*, *C. quaestio*, Ne – *Contracaecum* sp. [L], Cr – *Dolops ranarum*, *Lamproglena monodi*

Sargochromis giardi (Pellegrin): Cr – *Dolops ranarum*, *Leiperia cincinnalis* [L]

Sargochromis mellandi (Boulenger): Mo – *Cichlidogyrus consobrini*, *C. zambezensis*, Cr – *Lernaea hardingi*

Sarotherodon caudomarginatus (Boulenger): Mo – *Cichlidogyrus giostrai*

Sarotherodon galilaeus (Linnaeus): Pr – *Ambiphrya ameiuri*, *Amphileptus* sp., *Apiosoma epibranchialis*, *Chilodonella hexasticha*, *Cryptobia* sp., *Hexamita* sp., *Paratrichodina africana*, *Tetrahymena corlissi*, *Trichodina compacta*, *T. frenata*, *T. magna*, *Tripartiella orthodens*, *Vorticella* sp., Mx – *Henneguya branchialis*, *H. sarotherodoni*, *Myxobolus agolus*, *M. brachysporus*, *M. equatorialis*, *M. galilaeus*, *M. homeosporus*, *M. israelensis*, *M. kainjiae*, *M. nounensis*, *M. sarigi*, *M. tilapiae*, *M. tingrelaensi*, Mo – *Cichlidogyrus acerbus*, *C. aegypticus*, *C. arthracanthus*, *C. bychowskii*, *C. cirratus*, *C. dionchus*, *C. douellouae*, *C. halli*, *C. nageus*, *C. njinei*, *C. sclerosus*, *C. thurstonae*, *C. tilapiae*, *Enterogyrus amieti*, *E. cichlidarum*, *Gyrodactylus cichlidarum*, *G. ergensi*, *Scutogyrus bailloni*, *S. longicornis*, Tr – *Clinostomum complanatum* [L], *C. tilapiae* [L], *Clinostomum* sp. [L], Ce – *Cyclustera magna* [L], Ac – *Acanthogyrus tilapiae*, Ne – *Amplicaecum* sp. (type I) [L], *Amplicaecum* sp. (type II) [L], *Gendria tilapiae*, Cr – *Ergasilus latus*, *Lamproglena monodi*, *Lernaea hardingi*

Sarotherodon melanotheron Rüppell: Mx – *Myxobolus beninensis*, *M. dahomeyensis*, *M. diamaensis*, *M. heterosporus*, *M. nokoueensis*, *M. nyongana*, *M. sarotherodoni*, Mo – *Cichlidogyrus acerbus*, *C. halinus*, *C. halli*, *C. lagoonaris*, *Enterogyrus foratus*, *Gyrodactylus cichlidarum*, *Scutogyrus minus*, Tr – *Clinostomum tilapiae* [L], *Euclinostomum heterostomum* [L], Ac – *Acanthogyrus tilapiae*, Cr – *Ergasilus latus*, *Paraergasilus lagoonaris*

Sarotherodon mvogoi (Thys van den Audenaerde): Mx – *Myxobolus israelensis*, *M. tilapiae*, Mo – *Cichlidogyrus mvogoi*

Sarotherodon nigripinnis (Guichenot): Cr – *Ergasilus latus*

Sarotherodon occidentalis (Daget): Mo – *Cichlidogyrus bouvii*, *C. fontanai*, *C. guirali*, *C. halli*, *C. paganoi*, *C. sanjeani*, *Scutogyrus ecoutini*

Schwetzochromis neodon Poll: Cr – *Lamproglena monodi*

Serranochromis angusticeps (Boulenger): Pr – *Babesiosoma mariae*, *Trichodina centrostrigeata*, *T. compacta*, *T. magna*, *Trypanosoma mukasai*, Mo – *Cichlidogyrus zambezensis*, Ne – *Contracaecum* sp. [L], *Procamallanus* (*Spirocamallanus*) *serranochromis*, Physalopteridae gen. sp. 2

Serranochromis macrocephalus (Boulenger): Pr – *Trypanosoma mukasai*, Mo – *Cichlidogyrus dossoui*, *C. halli*, *C. quaestio*, *C. sclerosus*, *C. zambezensis*, Ne –

Contracaecum sp. [L], *Procamallanus* (*Spirocamallanus*) *serranochromis*, Cr – *Dolops ranarum*, *Lamproglena monodi*

Serranochromis meridianus Jubb: Ne – *Rhabdochona* sp., Philometridae gen. sp., Cr – *Leiperia cincinnalis* [L]

Serranochromis robustus (Günther): Pr – *Trypanosoma mukasai*, Mo – *Cichlidogyrus zambezensis*, Ne – *Contracaecum* sp. [L], *Procamallanus* (*Spirocamallanus*) *serranochromis*, Cr – *Argulus cunningtoni*, *Dolops ranarum*

Serranochromis stappersi Trewavas: Mo – *Cichlidogyrus zambezensis*

Serranochromis thumbergi (Castelnau): Mo – *Cichlidogyrus zambezensis*, Cr – *Lamproglena monodi*

***Serranochromis* sp.**: Cr – *Dolops ranarum*

Simochromis diagramma (Günther): Mo – *Cichlidogyrus banyankimbonai*, *C. muterezii*, *C. raeymaekersi*, *Gyrodactylus sturmbaueri*, *G. thysi*, *G. zimbae*, Cr – *Argulus exiguus*, *Ichthyoxenos tanganyikae*

***Simochromis* sp.**: Cr – *Ergasilus megacheir*

Stigmatochromis woodi (Regan): Ac – *Acanthogyrus tilapiae*

Stomatepia pindu Trewavas: Mo – *Enterogyrus barombiensis*, *Urogyrus cichlidarum*

Taeniolethrinops praeorbitalis (Regan): Cr – *Lernaea lophiara*

Tanganicodus irsacae Poll: Mo – *Cichlidogyrus evikae*

Thoracochromis callichromus (Poll): Cr – *Lamproglena monodi*

Thoracochromis moeruensis (Boulenger): Cr – *Ergasilus sarsi*, *Lamproglena monodi*

Thoracochromis schwetzi (Poll): Ne – *Prosungulonema africanum*, *P. campanae*, Cr – *Lamproglena monodi*

Thoracochromis wingatii (Boulenger): Mo – *Cichlidogyrus haplochromii*, Ne – *Contracaecum* sp. [L], *Galoiceps* sp. [L], *Hysterothylacium* sp. [L], *Rhabdochona* (*Globochona*) *paski*, Cr – *Lamproglena monodi*

Tilapia brevimanus Boulenger: Mo – *Cichlidogyrus albareti*, *C. digitatus*, *C. hemi*

Tilapia busumana (Günther): Mo – *Cichlidogyrus tilapiae*

Tilapia sparrmanii Smith: Pr – *Apiosoma constricta*, *A. dermatum*, *A. viridis*, *Chilodonella hexasticha*, *C. piscicola*, *Goussia vanasi*, *Ichthyobodo necator*, *Trichodina centrostrigeata*, *T. compacta*, *T. heterodentata*, *T. magna*, *T. minuta*, *T. nigra*, *Trypanosoma mukasai*, Mo – *Cichlidogyrus dossoui*, *C. papernastrema*, *C. quaestio*, *C. tiberianus*, Tr – Diplostomidae gen. sp. [L], Ce – *Amirthalingamia macracantha* [L], *Neogryporhynchus lasiopeius* [L], Ne – *Contracaecum* sp. [L], *Cucullanus djilorensis*, *Cucullanus* sp., Cr – *Argulus japonicus*

***Tilapia* sp.**: Pr – *Paratrichodina africana*, *Trichodina equatorialis*, Mx – *Myxobolus occularis*, *M. sheroidalis*, Mo – *Cichlidogyrus arthracanthus*, *Gyrodactylus occupatus*, *G. parisellei*, *Urogyrus cichlidarum*, Tr – *Clinostomum* sp. [L], *Euclinostomum heterostomum*

[L], Ce – *Bothriocephalus* sp., *Parvitaenia samfyia* [L], Ac – *Polyacanthorhynchus kenyensis* [L], Ne – *Porrocaecum* sp. [L], Cr – *Argulus africanus*, *A. rhipidiophorus*, *Dolops ranarum*, *Ergasilus kandti*, *E. macrodactylus*, *Lernaea cyprinacea*

Trematocara unimaculatum Boulenger: Mo – *Cichlidogyrus brunnensis*

Trematocranus placodon (Regan): Ac – *Acanthogyrus tilapiae*

Tropheops microstoma (Trewavas): Ac – *Acanthogyrus tilapiae*

Tropheops tropheops (Regan): Ac – *Acanthogyrus tilapiae*, Cr – *Lernaea lophiara*, *Paraergasilus minutus*

Tropheus moorii Boulenger: Ne – *Cystoopsis* sp.

Tylochromis bangwelensis Regan: Cr – *Ergasilus kandti*, *E. sarsi*

Tylochromis intermedius (Boulenger): Mo – *Cichlidogyrus pouyaudi*

Tylochromis jentinki (Steindachner): Mo – *Cichlidogyrus berrebii*, *C. kothiasi*, *C. pouyaudi*

Tylochromis labrodon Regan: Cr – *Lamproglena monodi*

Tylochromis lateralis (Boulenger): Cr – *Ergasilus cunningtoni*, *E. nodosus*

Tylochromis microdon Regan: Cr – *Ergasilus cunningtoni*, *E. nodosus*, *E. sarsi*

Tylochromis mylodon Regan: Cr – *Ergasilus kandti*, *E. sarsi*, *Lernaea barnimiana*

Tylochromis polylepis (Boulenger): Mo – *Cichlidogyrus mulimbwai*, *C. muzumanii*, Cr – *Ergasilus kandti*

Tylochromis sudanensis Daget: Mo – *Cichlidogyrus chrysopiformis*, *C. djietoi*, *C. sigmocirrus*

***Tylochromis* sp.**: Cr – *Lernaea barnimiana*

Tyrannochromis macrostoma (Regan): Ac – *Acanthogyrus tilapiae*

Oreochromis mossambicus* x *O. niloticus: Mx – *Myxobolus dahomeyensis*, *M. dossoui*, Mo – *Cichlidogyrus mbirizei*

Oreochromis niloticus* x *O. aureus: Mx – *Myxobolus israelenses*

Oreochromis niloticus x O. mweruensis*:* Mo – *Cichlidogyrus halli*, *C. mbirizei*, *Gyrodactylus nyanzae*

Oreochromis niloticus* x *Sarotherodon galilaeus: Mx – *Myxobolus brachysporus*, *M. galilaeus*, *M. sarigi*

'Carnivorous cichlid': Ce – *Proteocephalus bivitellatus*

Unknown cichlid: Ce – *Lytocestoides tanganyikae*, Tr – *Neocladocystis tanganyikae*

FAMILY ELEOTRIDAE

Kribia kribensis (Boulenger): Mx – *Kudoa eleotrici*

FAMILY GOBIIDAE

Awaous lateristriga (Duméril): Cr – *Paraergasilus lagoonaris*

Glossogobius giuris (Hamilton): Pr – *Trichodina heterodentata*, Mo – *Dactylogyrus jubbstrema*, Cr – *Ergasilus mirabilis*

Periophthalmus barbarus (Linnaeus): Ne – *Porrocaecum* sp. [L]

Sicyopterus fasciatus (Day): Ne – *Rhabdochona* (*R*.) *srivastavai*

FAMILY HAEMULIDAE

Pomadasys commersonnii (Lacepède): Ne – *Contracaecum* sp. [L], Cr – *Argulus kosus*

Pomadasys multimaculatus (Playfair): Cr – *Argulus kosus*

Pomadasys olivaceus (Day): Ne – *Contracaecum* sp. [L]

FAMILY LATIDAE

Lates angustifrons Boulenger: Cr – *Argulus rubropunctatus*, *Opistholernaea longa*

Lates microlepis Boulenger: Ne – *Dujardinascaris* sp., Cr – *Argulus africanus*, *A. rubropunctatus*, *Dolops ranarum*, *Leiperia cincinnalis* [L], *Opistholernaea longa*

Lates niloticus (Linnaeus): Pr – *Ambiphrya ameiuri*, *Ambiphrya* sp., *Amphileptus niloticus*, *Amphileptus* sp., *Apiosoma amoebae*, *A. doliaris*, *A. epibranchialis*, *A. poteriformis*, *Paratrichodina africana*, *Riboscyphidia doliaris*, *R. globularis*, *R. mansourensis*, *Riboscyphidia* sp., *Tetrahymena* sp., *Trichodina compacta*, *T. frenata*, *T. lepsii*, *T. magna*, *T. rectuncinata*, Mx – *Henneguya ghaffari*, *H. mandouri*, *H. maraensis*, *H. massii*, *H. mbakaouensis*, *Myxidium latesi*, *Myxobolus latesi*, *M. latis*, *M. zillii*, Mo – *Diplectanum lacustre*, *Macrogyrodactylus latesi*, Ac – *Paragorgorhynchus albertianus*, *P. aswanensis*, *P. chariensis*, *Tenuisentis niloticus*, Ne – *Contracaecum* sp. [L], *Dichelyne fossor*, *Dichelyne* sp., *Philometra lati*, *P. spiriformis*, *Spinitectus allaeri*, Cr – *Argulus cunningtoni*, *A. rhipidiophorus*, *Dolops ranarum*, *Ergasilus kandti*, *Lamproglena clariae*, *Leiperia cincinnalis* [L], *Lernaea barnimiana*, *Opistholernaea longa*

***Lates* sp.**: Cr – *Argulus cunningtoni*, *A. rhipidiophorus*

FAMILY LUTJANIDAE

Lutjanus goreensis (Valenciennes): Tr – *Siphodera ghanensis*

FAMILY NANDIDAE

Polycentropsis abbreviata Boulenger: Mx – *Myxobolus polycentropsi*, Mo – *Cichlidogyrus inconsultans*, *C. nandidae*

FAMILY POLYNEMIDAE

Polydactylus quadrifilis (Cuvier): Cr – *Argulus dartevellei*

FAMILY POMACENTRIDAE

***Chromis* sp.**: Cr – *Chonopeltis inermis*

FAMILY SCIAENIDAE

Otolithes ruber (Bloch et Schneider): Cr – *Argulus kosus*

Pseudotolithus typus Bleeker: Cr – *Argulus arcassonensis*

FAMILY SERRANIDAE

Epinephelus itajara (Lichtenstein): Cr – *Argulus dartevellei*

FAMILY SPARIDAE

Boops boops (Linnaeus): Cr – *Argulus vittatus*

Pagellus erythrinus (Linnaeus): Cr – *Argulus vittatus*

Pagrus pagrus (Linnaeus): Cr – *Argulus vittatus*

Rhabdosargus holubi (Steindachner): Cr – *Argulus kosus*

Rhabdosargus sarba (Forsskål): Ne – *Procamallanus* (*Spirocamallanus*) *olseni*

Sarpa salpa (Linnaeus): Cr – *Argulus kosus*

Sparus aurata Linnaeus: Cr – *Argulus vittatus*

FAMILY SPHYRAENIDAE

Sphyraena barracuda (Edwards): Cr – *Argulus belones*

ORDER PLEURONECTIFORMES

FAMILY SOLEIDAE

Solea solea (Linnaeus): Cr – *Ergasilus lizae*

ORDER POLYPTERIFORMES

FAMILY POLYPTERIDAE

Erpetoichthys calabaricus Smith: Ne – *Gendria polypteri*, *Procamallanus* (*P.*) *siluranae*

Polypterus bichir Lacepède: Tr – *Callodistomum diaphanum*, Ce – *Polyonchobothrium polypteri*, *Proteocephalus pentastomus*, Ne – *Camallanus polypteri*, Cr – *Lernaea haplocephala*

Polypterus congicus Boulenger: Cr – *Lernaea haplocephala*

Polypterus endlicheri Heckel: Tr – *Callodistomum diaphanum*, Ce – *Polyonchobothrium polypteri*, *Proteocephalus pentastomus*, *P. sulcatus* (?), Ne – *Amplicaecum* sp. (type I) [L], *Procamallanus* (*Spirocamallanus*) *spiralis*

Polypterus senegalus Cuvier: Mo – *Diplogyrodactylus martini*, *Macrogyrodactylus polypteri*, *M. simetiensis*, Ce – *Polyonchobothrium polypteri*, *Proteocephalus pentastomus*, Ne – *Amplicaecum* sp. (type I) [L], *Gendria polypteri*, *Rhabdochona* (*Globochona*) *paski*, Cr – *Lernaea haplocephala*, *L. senegali*

***Polypterus* sp.**: Cr – *Argulus africanus*

ORDER SALMONIFORMES

FAMILY SALMONIDAE

Oncorhynchus mykiss (Walbaum): Pr – *Apiosoma* sp., *Ichthyophthirius multifiliis*, *Trichodina acuta*, *Trichodina* sp., Cr – *Argulus japonicus*

Salmo trutta Linnaeus: Pr – *Ichthyophthirius multifiliis*, *Trichodina* sp.

Salvelinus fontinalis (Mitchill): Mx – *Myxobolus ovoidalis*

ORDER SCORPAENIFORMES

FAMILY DACTYLOPTERIDAE

Dactylopterus volitans (Linnaeus): Cr – *Argulus dactylopteri*

ORDER SILURIFORMES

FAMILY AMPHILIIDAE

Amphilius atesuensis Boulenger: Mo – *Gyrodactylus amphiliusi*

Amphilius grandis Boulenger: Cr – *Chonopeltis brevis*

Amphilius longirostris (Boulenger): Mx – *Chloromyxum birgii*

Amphilius uranoscopus (Pfeffer): Tr – *Clinostomum* sp. [L]

***Amphilius* sp.**: Cr – *Chonopeltis brevis*

FAMILY ARIIDAE

Arius latiscutatus Günther: Cr – *Lepeophtheirus monacanthus*

Arius madagascariensis Vaillant: Ne – *Falcaustra therezieni*

***Arius* sp.**: Cr – *Dartevellia bilobata*

Carlarius heudelotii (Valenciennes): Cr – *Lepeophtheirus monacanthus*

FAMILY BAGRIDAE

Bagrus bajad (Forsskål): Mx – *Chloromyxum vanasi*, *Myxobolus bagri*, Mo – *Quadriacanthus bagrae*, Tr – *Acanthostomum absconditum*, *A. spiniceps*, *Emoleptalea exilis*, *Glossidium pedatum*, *Haplorchoides cahirinus*, *Heterophyes* sp. [L], *Phyllodistomum spatula*, *P. symmetrorchis*, Ac – *Paragorgorhynchus albertianus*, *P. aswanensis*, Ne – *Amplicaecum* sp. (type I) [L], *Capillostrongyloides fritschi*, *Contracaecum* sp. [L], *Dichelyne fossor*, *Philometra bagri*, *Procamallanus* (*P*.) *laeviconchus*, *Rhabdochona* (*Globochona*) *paski*, *Spinitectus allaeri*, Cr – *Argulus cunningtoni*, *A. rhipidiophorus*, *Dolops ranarum*, *Ergasilus kandti*, *E. nodosus*, *Lamproglena werneri*

Bagrus degeni Boulenger: Cr – *Argulus africanus*, *Dolops ranarum*

Bagrus docmak (Forsskål): Pr – *Trypanosoma mukasai*, Mo – *Quadriacanthus bagrae*, Tr – *Acanthostomum absconditum*, *A. spiniceps*, *Astiotrema reniferum*, *Glossidium pedatum*, *Haplorchoides cahirinus*, *Phyllodistomum spatula*, *Sandonia sudanensis*, Ac – *Paragorgorhynchus aswanensis*, Ne – *Amplicaecum* sp. (type I) [L], *Capillostrongyloides fritschi*, *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Procamallanus* (*P*.) *laeviconchus*, *Rhabdochona* (*Globochona*) *paski*, *Spinitectus allaeri*, Cr – *Argulus africanus*, *Dolops ranarum*, *Lernaea cyprinacea*

Bagrus filamentosus Pellegrin: Tr – *Acanthostomum spiniceps*

Bagrus meridionalis Günther: Tr – *Haplorchoides cahirinus*, *Malawitrema staufferi*, Ce – *Tetracampos martinae*, Ac – *Acanthogyrus tilapiae*, Cr – *Argulus africanus*, *Lernaea bagri*

Bagrus orientalis Boulenger: Mo – *Quadriacanthus bagrae*

***Bagrus* sp.**: Tr – *Nephrocephalus bagriincapsulatus* [L], Cr – *Argulus rhipidiophorus*, *Ergasilus nodosus*

FAMILY CLARIIDAE

Bathyclarias nyasensis (Worthington): Cr – *Chonopeltis inermis*

Clariallabes laticeps (Steindachner): Ne – *Paracamallanus cyathopharynx*

Clarias alluaudi Boulenger: Ne – *Procamallanus* (*P*.) *pseudolaeviconchus*

Clarias angolensis Steindachner: Pr – *Trypanosoma tobeyi*, *T. toddi*, Tr – *Euclinostomum clarias* [L]

Clarias anguillaris (Linnaeus): Pr – *Trypanosoma toddi*, Mx – *Henneguya branchialis*, *H. fusiformis*, *H. suprabranchiae*, *Myxobolus comoei*, Mo – *Gyrodactylus gelnari*, *G. rysavyi*, *G. tranvaalensis*, *Macrogyrodactylus congolensis*, *M. heterobranchii*, Tr – *Masenia proteropora*, *Nephrocephalus bagriincapsulatus* [L], *Orientocreadium batrachoides*, *Pseudoneodiplostomum thomasi* [L], Ce – *Kirstenella gordoni* (?), *Marsypocephalus rectangulus*, *Monobothrioides chalmersius*, *Proteocephalus glanduligerus*, *P. sulcatus* (?), *Tetracampos ciliotheca*, Ne – *Amplicaecum* sp. (type I) [L], *Camallanus polypteri*, *Eustrongylides africanus* [L], *Paracamallanus cyathopharynx*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*P*.) *pseudolaeviconchus*, *P*. (*Spirocamallanus*) *spiralis*, Cr – *Argulus africanus*, *A. dageti*, *Dolops ranarum*, *Ergasilus sarsi*, *Lamproglena clariae*

Clarias buettikoferi Steindachner: Ce – *Lytocestus puylaerti*

Clarias buthupogon Sauvage: Ne – *Porrocaecum* sp. [L]

Clarias camerunensis Lönnberg: Mo – *Birgiellus kellensis*, *Gyrodactylus camerunensis*, *G. nyongensis*, *Quadriacanthus voltaensis*, Ne – *Eustrongylides* sp. [L]

Clarias dumerilii Steindachner: Ne – *Porrocaecum* sp. [L]

Clarias ebriensis Pellegrin: Mo – *Quadriacanthus* eboreus, *Q. ivoiriensis*

Clarias gariepinus (Burchell): Pr – *Ambiphrya* sp., *Amphileptus* sp., *Balantidium* sp., *Capriniana* sp., *Chilodonella hexasticha*, *Chilodonella* sp., *Cryptobia iubilans*, *Cryptobia* sp., *Entamoeba* sp., *Epistylis* sp., *Goussia molnarica*, *Hexamita* sp., *Ichthyobodo* sp., *Ichthyophthirius multifiliis*, *Ichthyophthirius* sp., *Piscinoodinium* sp., *Protoopalina* sp., *Tetrahymena* sp., *Trichodina centrostrigeata*, *T. magna*, *T. maritinkae*, *T. matsu*, *T. rectuncinata*, *T. sangwala*, *Trichodina* sp., *Trypanosoma alhussaini*, *T. mukasai*, *Vorticella* sp., Microsporidia gen. sp., Mx – *Henneguya branchialis*, *H. clariae*, *H. fusiformis*, *H. laterocapsulata*, *H. samochimensis*, *H. suprabranchiae*, *Myxobolus gariepinus*, *M. lazera*, Mo – *Gyrodactylus alberti*, *G. alekosi*, *G. clarii*, *G. gelnari*, *G. groschafti*, *G. rysavyi*, *G. tranvaalensis*, *G. turkanaensis*, *Macrogyrodactylus clarii*, *M. congolensis*, *M. karibae*, *Paraquadriacanthus nasalis*, *Quadriacanthus aegypticus*, *Q. allobychowskiella*, *Q. ashuri*, *Q. bagrae*, *Q. clariadis*, *Q. fornicatus*, *Q. numidus*, *Q. papernai*, *Q. pravus*, *Q. voltaensis*, *Q. zuheiri*, Tr – *Allocreadium mazoensis*, *Astiotrema lazeri*, *A. reniferum*, *Clinostomoides brieni* [L], *Clinostomum macrosomum* [L], *Clinostomum* sp. [L], *Diplostomum heterobranchi* [L], *Diplostomum* sp. [L], *Dolichorchis tregenna* [L], *Euclinostomum dollfusi* [L], *E. heterostomum* [L], *Glossidium lazerae*, *G. pedatum*, *Gonocerca phycidis*, *Haplorchoides cahirinus*, *Malawitrema staufferi*, *Masenia bangweulensis*, *M. ghanensis*, *Neodiplostomum* sp. [L], *Orientocreadium batrachoides*, *O. indicum*, *Ornithodiplostomum* sp. [L], *Panamphistomum benoiti*, *Phyllodistomum bavuri*, *P*. *tana*, *P*. *vanderwaali*, *P.* cf. *symmetrorchis*, *Sanguinicola clarias*, *Thaparotrema botswanensis*, *Tylodelphys grandis* [L], *T. mashonensis* [L], *Tylodelphys* sp. [L], Cryptogonimidae gen. sp. [L], Diplostomidae gen. sp. [L], Ce – *Barsonella lafoni*, *Marsypocephalus aegyptiacus*, *M*. *rectangulus*, *M. tanganyikae*, *Proteocephalus glanduligerus*, *P. sulcatus* (?), *Stocksia pujehuni*, *Tetracampos ciliotheca*, Ac – *Neoechinorhynchus rutili*, *Neoechinorhynchus* sp., *Paragorgorhynchus aswanensis*, *Paragorgorhynchus* sp., Ne – *Amplicaecum* sp. (type I) [L], *Camallanus* sp., *Capillaria* sp., *Chordocephalus* sp., *Contracaecum* sp. [L], *Eustrongylides africanus* [L], *Eustrongylides* sp. [L], *Galeiceps* sp. [L], *Paracamallanus cyathopharynx*, *Paracamallanus* sp., *Procamallanus* (*P*.) *laeviconchus*, *P.* (*P*.) *pseudolaeviconchus*, *P.* (*Spirocamallanus*) *spiralis*, *Procamallanus* sp., *Rhabdochona* (*Globochona*) *paski*, *Rhabdochona* sp., *Spinitectus allaeri*, *S. petterae*, Atractidae gen. sp. [L], Physalopteridae gen. sp. 1, Cr – *Afrolernaea annemari*, *Argulus africanus*, *A. amblopIites*, *A. cunningtoni*, *A. japonicus*, *A. rhipidiophorus*, *A. striatus*, *Chonopeltis fryeri*, *C. inermis*, *Dolops ranarum*, *Ergasilus mirabilis*, *E. sarsi*, *Lamproglena clariae*, *L. cornuta*, *Lernaea composita*, *L. diceracephala*, *Sebekia okavangoensis* [L], Hi – *Batracobdelloides tricarinata*

Clarias jaensis Boulenger: Mo – *Birgiellus calaris*, *Quadriacanthus dageti*, *Q. nyongensis*, *Q. teugelsi*

Clarias laticeps (Steindachner): Ce – *Proteocephalus sulcatus* (?)

Clarias liocephalus Boulenger: Ce – *Tetracampos ciliotheca*, Ne – *Contracaecum* sp. [L]

Clarias ngamensis Castelnau: Tr – *Masenia bangweulensis*, *Orientocreadium batrachoides*, *Tylodelphys mashonensis* [L], Ce – *Monobothrioides woodlandi*, Ne – *Contracaecum* sp. [L], Cr – *Ergasilus mirabilis*, *E. sarsi*, *Lamproglena clariae*

Clarias pachynema Boulenger: Mx – *Myxidium nkamense*, Mo – *Birgiellus mutatus*, *Quadriacanthus levequei*, *Q. nyongensis*, *Q. teugelsi*, Ne – *Procamallanus* sp.

Clarias platycephalus Boulenger: Ne – *Contracaecum* sp. [L]

Clarias stappersii Boulenger: Pr – *Trichodina maritinkae*, Ne – *Contracaecum* sp. [L], *Paracamallanus cyathopharynx*, *Procamallanus* (*P*.) *pseudolaeviconchus*, *P*. (*Spirocamallanus*) *spiralis*, Cr – *Dolops ranarum*

Clarias submarginatus Peters: Mo – *Quadriacanthus macruncus*, *Q*. *ossaensis*, *Q*. *submarginati*

Clarias theodorae Weber: Pr – *Trypanosoma mukasai*, Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Paracamallanus cyathopharynx*, *Procamallanus* (*P*.) *pseudolaeviconchus*, *P*. (*Spirocamallanus*) *spiralis*, Cr – *Chonopeltis fryeri*, *C*. *inermis*

Clarias werneri Boulenger: Ce – *Tetracampos ciliotheca*, Ne – *Paracamallanus cyathopharynx*

***Clarias* sp.** Pr – *Eimeria* sp., Mx – *Myxobolus sheroidalis*, Mo – *Macrogyrodactylus clarii*, Tr – *Euclinostomum heterostomum* [L], Ne – *Contracaecum* sp. [L], *Eustrongylides* sp. [L], *Paracamallanus cyathopharynx*, *Procamallanus* sp., *Spinitectus allaeri*, Cr – *Argulus africanus*, *A. rhipidiophorus*, *Dolops ranarum*, *Lamproglena clariae*

Dinotopterus cunningtoni Boulenger: Ce – *Proteocephalus cunningtoni*, *P*. *dinotopteri*, Ne – *Eustrongylides* sp. [L], Cr – *Argulus striatus*

Heterobranchus bidorsalis Geoffroy Saint-Hilaire: Pr – *Chilodonella* sp., *Trichodina* sp., Mx – *Myxobilatus accessobranchialis*, Mo – *Quadriacanthus mandibulatus*, Ce – *Kirstenella gordoni*, *Marsypocephalus daveyi*, *M. heterobranchus*, *M. rectangulus*, *Marsypocephalus* sp., Ne – *Gendria chabaudi*, Cr – *Argulus africanus*, *A. dageti*, *Dolops ranarum*, *Ergasilus sarsi*, *Lamproglena cornuta*

Heterobranchus isopterus Bleeker: Mo – *Quadriacanthus agnebiensis*, *Q. ayameensis*, *Q. gourenei*, *Q. macrocirrus*, *Q. simplex*, Tr – *Masenia bangweulensis*, *M. ghanensis*, Ne – *Spinitectus allaeri*, *S. macilentus*

Heterobranchus longifilis Valenciennes: Pr – *Chilodonella* sp., *Cryptobia iubilans*, *Hexamita* sp., *Ichthyobodo* sp., *Ichthyophthirius multifiliis*, *Trichodina maritinkae*, *Trichodina* sp., Mo – *Macrogyrodactylus heterobranchii*, *Quadriacanthus longifilisi*, *Q. ndoubai*, *Q. thysi*, *Q. tricorniculai*, *Q. triunguisi*, Tr – *Masenia ghanensis*, *Orientocreadium indicum*, *Posthodiplostomum nanum* [L], Ne – *Paracamallanus cyathopharynx*, Cr – *Argulus africanus*, *Lernaea diceracephala*

***Heterobranchus* sp.**: Cr – *Argulus africanus*, *A*. *striatus*, *Dolops ranarum*

Xenoclarias eupogon (Norman): Ne – *Spinitectus allaeri*

Clarias gariepinus x ***Heterobranchus bidorsalis***: Mx – *Henneguya laterocapsulata*

FAMILY CLAROTEIDAE

Auchenoglanis biscutatus (Geoffroy Saint-Hilaire): Ce – *Monobothrioides tchadensis*, Ne – *Cithariniella khalili*, *Procamallanus* (*P*.) *laeviconchus*

Auchenoglanis occidentalis (Valenciennes): Mx – *Henneguya auchenoglanii*, Mo – *Bagrobdella anthopenis*, *B. auchenoglanii*, *B. fraudulenta*, Tr – *Nephrocephalus bagriincapsulatus* [L], *Phyllodistomum symmetrorchis*, *Sanguinicola chalmersi*, Ce – *Lytocestus* sp., *Monobothrioides cunningtoni*, *Proteocephalus synodontis*, Ne – *Cucullanus congolensis*, *Mexiconema africanum*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *spiralis*, *Rhabdochona* (*Globochona*) *paski* [L], Cr – *Argulus cunningtoni*, *A. gracilis*, *A. incisus*, *A. striatus*, *Dolops ranarum*, *Ergasilus latus*, *Lamproglena werneri*

***Auchenoglanis* sp.**: Ne – Capillariidae gen. sp.

Chrysichthys auratus (Geoffroy Saint-Hilaire): Pr – *Ichthyophthirius multifiliis*, *Neonosemoides* sp., *Riboscyphidia* sp., Mx – *Myxidium beninensis*, *Myxobolus chrysichthyi*, *M. clarias*, Mo – *Protoancylodiscoides auratum*, *P. chrysichthes*, *P. combesi*, *P. mansourensis*, Tr – *Clinostomum chrysichthys* [L], Ce – *Wenyonia minuta* (?)

Chrysichthys brachynema Boulenger: Ce – *Proteocephalus beauchampi*, Cr – *Argulus africanus*, *A. rubescens*, *A. striatus*, *Leiperia cincinnalis* [L]

Chrysichthys longidorsalis Risch et Thys van den Audenaerde: Mo – *Protoancylodiscoides combesi*, *P. sanagaensis*, *P. valentini*

Chrysichthys mabusi Boulenger: Cr – *Leiperia cincinnalis* [L], *L. gracilis* [L], *Lernaea hardingi*

Chrysichthys nigrodigitatus (Lacepède): Pr – *Coccidia* sp., Mx – *Henneguya bopeleti*, *H. chrysichthyi*, *Myxidium beninensis*, *Myxobolus bouixi*, Mo – *Protoancylodiscoides chrysichthes*, *P. combesi*, *P. sanagaensis*, *P. spirovagina*, *P. valentini*, Tr – *Acanthostomum spiniceps*, *Aspidogaster africanus*, *Clinostomum complanatum* [L], *Siphodera ghanensis*, Ne – *Hysterothylacium* sp. [L], *Raphidascaroides bishaii*, *Rhabdochona* (*Globochona*) *paski*, Cr – *Chonopeltis brevis*, *Ergasilus latus*

***Chrysichthys* sp.**: Ce – *Proteocephalus beauchampi*, *P. sulcatus* (?), Ac – *Arhythmorhynchus siluricola*, Cr – *Dolops ranarum*

Clarotes laticeps (Rüppell): Ce – *Proteocephalus sulcatus* (?), Ne – *Camallanus* sp., *Capillaria* sp., *Contracaecum* sp. [L], *Cucullanus clarotis*, *Eustrongylides* sp. [L], *Paracamallanus cyathopharynx*, Cr – *Lamproglena hemprichii*

***Gephyroglanis* sp.**: Ac – *Arhythmorhynchus siluricola*

Notoglanidium macrostoma (Pellegrin): Mo – *Quadriacanthus anaspidoglanii*

Parauchenoglanis ballayi (Sauvage): Ce – *Monobothrioides* sp.

Parauchenoglanis monkei (Keilhack): Mo – *Bagrobdella parauchenoglanii*, Tr – *Neocladocystis congoensis*

Parauchenoglanis ngamensis (Boulenger): Pr – *Trypanosoma mukasai*

Parauchenoglanis punctatus (Boulenger): Ne – *Gendria thysi*

FAMILY MALAPTERURIDAE

Malapterurus electricus (Gmelin): Mx – *Henneguya malapteruri*, Mo – *Protoancylodiscoides katii*, *P. malapteruri*, Tr – *Brientrema malapteruri*, *Phyllodistomum spatulaeforme*, Ce – *Corallobothrium solidum*, *Electrotaenia malopteruri*, Ne – *Amplicaecum* sp. (type I) [L], *Capillaria* sp., *Capillostrongyloides fritschi*, *Contracaecum* sp. [L], *Dujardinascaris malapteruri*, *Procamallanus* (*P*.) *laeviconchus*, *Spinitectus allaeri*, Cr – *Lamproglena angusta*, *Lernaea barbicola*, *L. composita*

FAMILY MOCHOKIDAE

Chiloglanis pretoriae Van Der Horst: Pr – *Trichodina compacta*, Tr – *Clinostomum* sp. [L]

Synodontis acanthomias Boulenger: Ce – *Wenyonia acuminata*, Ne – *Cithariniella khalili*, *Falcaustra similis*, *Labeonema longispiculatum*, *Procamallanus* (*Spirocamallanus*) *dalenae*, *Synodontisia thelastomoides*

Synodontis afrofischeri Hilgendorf: Ne – *Procamallanus* (*Spirocamallanus*) *dalenae*, *Procamallanus* (*Spirocamallanus*) sp.

Synodontis ansorgii Boulenger: Mx – *Myxidium bouixi*, *Myxobilatus synodontis*, *Myxobolus dahomeyensis*

Synodontis batensoda Rüppell: Tr – *Allocreadium ghanensis*, *Basidiodiscus ectorchis*, *Pygidiopsis genata* [L], *Sandonia sudanensis*, Ce – *Proteocephalus synodontis*, *Wenyonia virilis*, *Wenyonia* sp., Ac – *Pararaosentis golvani*, Ne – *Cithariniella khalili*, *Contracaecum microcephalum* [L], *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *dalenae*, *Rhabdochona* (*Globochona*) *paski*

Synodontis batesii Boulenger: Mx – *Henneguya camerounensis*, *Myxobolus synodonti*

Synodontis budgetti Boulenger: Tr – *Sandonia sudanensis*, Ce – *Wenyonia virilis*

Synodontis caudovittatus Boulenger: Ce – *Proteocephalus synodontis*, *Wenyonia minuta*, *W. virilis*, *W. youdeoweii*

Synodontis clarias (Linnaeus): Mx – *Myxobolus stenosus*, Tr – *Basidiodiscus ectorchis*, *Sandonia sudanensis*, Ce – *Wenyonia virilis*, Ne – *Procamallanus* (*P*.) *laeviconchus*

Synodontis decorus Boulenger: Ne – *Spinitectus polli*, *Synodontisia thelastomoides*

Synodontis eupterus Boulenger: Tr – *Clinostomum* sp. [L], Ce – *Proteocephalus synodontis*, *Wenyonia virilis*, Ne – *Labeonema synodontisi*, *Procamallanus* (*Spirocamallanus*) *dalenae*, *P*. (*S*.) *spiralis*

Synodontis frontosus Vaillant: Ce – *Proteocephalus synodontis*, *Wenyonia minuta*, *W. virilis*, Ac – *Pararaosentis golvani*, Ne – *Cithariniella khalili*, *Falcaustra similis*, *Labeonema synodontisi*, *Procamallanus* (*Spirocamallanus*) *pseudospiralis*

Synodontis* cf. *geledensis Günther: Ce – *Wenyonia virilis*

Synodontis gobroni Daget: Ce – *Wenyonia youdeoweii*

Synodontis granulosus Boulenger: Cr – *Ergasilus megacheir*

Synodontis greshoffi Schilthuis: Ne – *Cithariniella khalili*, *Synodontisia thelastomoides*

Synodontis haugi Pellegrin: Ne – *Procamallanus* (*Spirocamallanus*) *dalenae*

Synodontis leopardinus Pellegrin: Pr – *Trichodina centrostrigeata*, *T. nkasa*, Cr – *Chonopeltis lisikili*, *Ergasilus mirabilis*

Synodontis longirostris Boulenger: Ne – *Cithariniella khalili*, Cr – *Chonopeltis elongatus*

Synodontis macrostigma Boulenger: Pr – *Trichodina nkasa*, Cr – *Chonopeltis lisikili*, *Ergasilus mirabilis*

Synodontis melanopterus Boulenger: Mo – *Synodontella melanoptera*

Synodontis membranaceus (Geoffroy Saint-Hilaire): Mo – *Synodontella davidi*, *S. synodontii*, Tr – *Sandonia sudanensis*, Ce – *Proteocephalus membranacei*, *Wenyonia acuminata*, Ac – *Hexaspiron nigericum*, *Neoechinorhynchus africanus*, *Pararaosentis golvani*, Ne – *Cithariniella khalili*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *dalenae*, *Procamallanus* (*Spirocamallanus*) sp., Cr – *Ergasilus kandti*

Synodontis multipunctatus Boulenger: Cr – *Ergasilus megacheir*

Synodontis nigrita Valenciennes: Mo – *Gyrodactylus nigritae*, *G*. *synodonti*, Tr – *Clinostomum* sp. [L], *Diplostomum sp*. [L], *Sandonia sudanensis*, Ce – *Proteocephalus synodontis*, *Wenyonia minuta*, *W. virilis*, Ne – *Cithariniella khalili*, *C*. *petterae*, *Falcaustra similis*, *Labeonema synodontisi*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *pseudospiralis*, *Synodontisia thelastomoides*

Synodontis nigriventris David: Ne – *Synodontisia thelastomoides*, Cr – *Ergasilus cunningtoni*, *E. nodosus*

Synodontis nigromaculatus Boulenger: Pr – *Trypanosoma mukasai*, Ne – *Contracaecum* sp. [L], *Falcaustra similis*, *Labeonema africanum*, *Procamallanus* (*P*.) *laeviconchus*, *Rhabdochona* (*Globochona*) *paski*, *Spinitectus polli*, *Spinitectus* spp., *Synodontisia okavangoensis*, Cr – *Chonopeltis lisikili*, *Dolops ranarum*, *Ergasilus mirabilis*, *E*. *sarsi*, *Lernaea hardingi*

Synodontis notatus Vaillant: Tr – *Emoleptalea synodontidos*, Ne – *Rhabdochona* (*Globochona*) *paski*

Synodontis obesus Boulanger: Mo – *Synodontella melanoptera*

Synodontis ocellifer Boulenger: Pr – Microsporidia gen. sp., Tr – *Sandonia sudanensis*, Ce – *Wenyonia virilis*, Ne – *Cithariniella khalili*, *Labeonema synodontisi*, *Procamallanus*

(*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *dalenae*, *Procamallanus* (*Spirocamallanus*) sp., *Synodontisia thelastomoides*

Synodontis pleurops Boulenger: Ne – *Synodontisia thelastomoides*

Synodontis rebeli Holly: Mo – *Synodontella apertipenis*, *S. melanoptera*, *S. sanagaensis*

Synodontis schall (Bloch et Schneider): Pr – *Apiosoma* sp., *Balantidium* sp., *Chilodonella* sp., *Cryptobia* sp., *Eimeria* sp., *Entamoeba synodontis*, *Hexamita africanus*, *Hexamita* sp., *Ichthyobodo* sp., *Ichthyophthirius* sp., *Trichodina* sp., Haemogregarine gen. sp., Microsporidia gen. sp., Mx – *Myxidium schalli*, *Myxobolus negmgoda*, *M. stenosus*, *Unicauda strongylura*, Tr – *Basidiodiscus ectorchis*, *Clinostomum* sp. [L], *Cholepotes ovofarctus*, *Emoleptalea rifaati*, *Sandonia sudanensis*, *Sanguinicola chalmersi*, Ce – *Monobothrioides* sp., *Proteocephalus beauchampi*, *P. synodontis*, *Wenyonia longicauda*, *W. minuta*, *W. synodontis*, *W. virilis*, *W. youdeoweii*, Ne – *Amplicaecum* sp. (type I) [L], *Camallanus polypteri*, *Cithariniella citharini*, *C. khalili*, *C. petterae*, *Cucullanus baylisi*, *C. clarotis*, *Falcaustra similis*, *Labeonema synodontisi*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *dalenae*, *P*. (*S*.) *pseudospiralis*, *Procamallanus* sp., *Procamallanus* (*Spirocamallanus*) sp., *Rhabdochona* (*Globochona*) *paski*, *Spinitectus allaeri*, *S. polli*, *Synodontisia thelastomoides*, Cr – *Argulus cunningtoni*, *A. rhipidiophorus*

Synodontis serratus Rüppell: Tr – *Emoleptalea rifaati*, Ce – *Proteocephalus synodontis*, *Wenyonia minuta*, *W. virilis*, *W. youdeoweii*, Ne – *Cithariniella citharini*, *C*. *khalili*, *Falcaustra similis*

Synodontis sorex Günther: Mo – *Synodontella arcopenis*, Tr – *Sandonia sudanensis*, Ce – *Wenyonia synodontis*, *W. virilis*, Ne – *Cithariniella khalili*, *Procamallanus* (*P*.) *laeviconchus*, *Synodontisia thelastomoides*

Synodontis tessmanni Pappenheim: Ne – *Procamallanus* (*Spirocamallanus*) *dalenae*, *P.* (*S*.) *spiralis*

Synodontis thamalakanensis Fowler: Ne – *Procamallanus* (*P*.) *laeviconchus*, Cr – *Chonopeltis lisikili*

Synodontis vanderwaali Skelton et White: Pr – *Trypanosoma mukasai*, Ne – *Falcaustra similis*, *Labeonema africanum*, *Procamallanus* (*P*.) *laeviconchus*, *P*. (*Spirocamallanus*) *dalenae*, *Synodontisia okavangoensis*, Cr – *Chonopeltis lisikili*

Synodontis vermiculatus Daget: Tr – *Sandonia sudanensis*, Ce – *Wenyonia synodontis*

Synodontis victoriae Boulenger: Mo – *Synodontella synodontii*, Tr – *Masenia synodontis*, Ne – *Procamallanus* (*Spirocamallanus*) *dalenae*

Synodontis zambezensis Peters: Pr – *Trichodina heterodentata*, Mo – *Synodontella synodontii*, *S*. *zambezensis*, Ne – *Capillaria* sp., *Labeonema synodontisi*, *Paracamallanus cyathopharynx*, *Procamallanus* (*Spirocamallanus*) *dalenae*, *Rhabdochona* sp., *Spinitectus polli*, *Synodontisia thelastomoides*, Philometridae gen. sp., Cr – *Dolops ranarum*, *Ergasilus mirabilis*

***Synodontis* sp.**: Tr – *Allocreadium ghanensis*, *Cholepotes ovofarctus*, *Sandonia sudanensis*, Ne – *Cucullanus baylisi*, *C*. *clarotis*

FAMILY PLOTOSIDAE

Plotosus lineatus (Thunberg): Cr – *Lepeophtheirus plotosi*

FAMILY SCHILBEIDAE

Parailia pellucida (Boulenger): Mo – *Schilbetrema bicornis*, Cr – *Ergasilus lamellifer*

Schilbe banguelensis (Boulenger): Cr – *Argulus africanus*

Schilbe grenfelli (Boulenger): Ne – *Gendria longispiculata*

Schilbe intermedius Rüppell: Pr – *Trypanosoma mukasai*, Mo – *Schilbetrema acornis*, *S. aegyptica*, *S. calamocleithrum*, *S. quadricornis*, *S. undinula*, *S. vacillans*, *Schilbetrematoides pseudodactylogyrus*, Tr – *Clinostomum* sp. [L], *Emoleptalea nwanedi*, Ce – *Dendrouterina herodiae* [L], Ne – *Cithariniella longicaudata*, *Contracaecum* sp. [L], *Falcaustra similis*, *Paracamallanus cyathopharynx*, *Procamallanus* (*P.*) *laeviconchus*, *Rhabdochona* sp., *Spinitectus* spp., *Synodontisia annulata*, Atractidae gen. sp., Philometridae gen. sp., Cr – *Dolops ranarum*, *Ergasilus mirabilis*

Schilbe laticeps (Boulenger): Cr – *Ergasilus cunningtoni*

Schilbe mandibularis (Günther): Mo – *Schilbetrema biclavula*, *S. dissimilis*, *Schilbetrematoides manizani*, Ne – *Labeonema bainae*

Schilbe marmoratus Boulenger: Ne – *Gendria sanghaensis*

Schilbe multitaeniatus (Pellegrin): Mx – *Henneguya camerounensis*

Schilbe mystus (Linnaeus): Pr – *Trichodina magna*, *T. sangwala*, Mx – *Chloromyxum alii*, *Henneguya ntondei*, *Myxidium schilba*, *Thelohanellus njinei*, Mo – *Schilbetrema eutropii*, *S. hexacornis*, *S. spirocirra*, *S. torula*, Tr – *Clinostomum vandehorsti* [L], *Prohemistomum vivax* [L], Ce – *Kirstenella gordoni* (?), Ac – *Paragorgorhynchus albertianus*, *Pararaosentis golvani*, Ne – *Amplicaecum* sp. (type I) [L], *Contracaecum* sp. [L], *Procamallanus* (*P.*) *laeviconchus*, *Rhabdochona* (*Globochona*) *paski*, *Spinitectus allaeri*, Cr – *Dolops ranarum*, *Ergasilus latus*, *E. mirabilis*

Schilbe uranoscopus Rüppell: Ne – *Amplicaecum* sp. (type I) [L]

***Schilbe* sp.**: Mo – *Schilbetrema tricera*, Cr – *Argulus africanus*, *Dolops ranarum*

ORDER SYNBRANCHIFORMES

FAMILY MASTACEMBELIDAE

Mastacembelus flavidus Matthes: Ne – *Spinitectus maleficus*

Mastacembelus frenatus Boulenger: Pr – *Trichodina frenata*

Mastacembelus micropectus Matthes: Ne – *Spinitectus micropectus*

Mastacembelus nigromarginatus Boulenger: Tr – *Phyllodistomum ghanense*

***Mastacembelus* sp.**: Cr – *Leiperia cincinnalis* [L]

ORDER TETRAODONTIFORMES

FAMILY MONACANTHIDAE

Aluterus monoceros (Linnaeus): Cr – *Argulus kosus*

FAMILY TETRAODONTIDAE

Tetraodon lineatus Linnaeus: Pr – *Trichodina fahaka*, Mo – *Heterobothrium fluviatilis*, Tr – *Astiotrema impletum*, Ac – *Acanthogyrus tilapiae*, *Paragorgorhynchus aswanensis*, *Pararaosentis golvani*, Ne – *Procamallanus* (*P.*) *laeviconchus*, Cr – *Argulus dageti*, *Dolops ranarum*

ORDER ZEIFORMES

FAMILY ZEIDAE

Zeus faber Linnaeus: Cr – *Argulus arcassonensis*

***Zeus* sp.**: Cr – *Argulus alexandrensis*

OTHER HOSTS

FISHES

Unidentified fish: Cr – *Argulus angusticeps*, *A. confusus*, *A. rijckmansii*, *Caligus pharaonis*, *Lamproglena wilsoni*

Unidentified shark: Cr – *Argulus melita*

FROGS

ORDER ANURA

UNIDENTIFIED FROG

Unidentified frog: Cr – *Dolops ranarum*

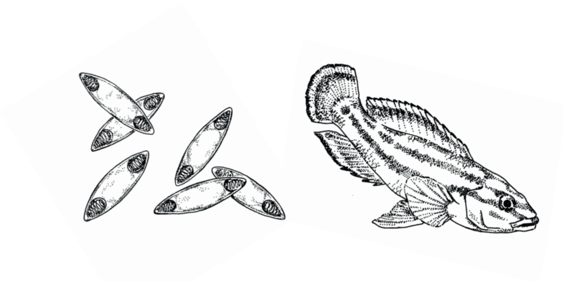
PART 6

EVOLUTIONARY PARASITOLOGY OF AFRICAN FRESHWATER FISHES

AND ITS IMPLICATIONS

FOR THE SUSTAINABLE MANAGEMENT

OF AQUATIC RESOURCES



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This book is intended as an aid in investigating the diversity and ecology of parasites of African freshwater fishes. However, given the species richness and degree of endemicity of African freshwater fishes, and the existence of several textbook cases in evolutionary biology, the evolution of their parasite fauna is also a promising subject for research.

When attempting to establish the historical relationships and diversification mechanisms of parasites through a phylogenetic approach, a recurrent question is to what extent the distribution of character states for typical parasitological traits such as host choice, host-specificity or infection site corresponds to the animals' classification. In this respect, the analysis of morphological or other phenotypic traits in combination with molecular data is critical to understanding parasite evolution. However, any morphology- or genetics-based phylogenetic approach requires coverage of as many representatives of the taxa under study as possible. Despite the progress in molecular techniques, achieving adequate taxon coverage (not to mention phenotypic characterisation) remains a challenge that hampers the development of comprehensive hypotheses about their evolutionary relationships.

Data on African fish parasites are relatively scarce and the rate of species discovery has not kept up with recent advances in phylogenetics and molecular biology. For example, regarding the evolution of cymothoid isopods, whose historical relationships have traditionally been regarded as correlated with their infection site, Smit *et al.* (2014) assert that the small numbers of representatives covered is a point of attention for recent molecular phylogenetic work. Phylogenetic studies on African lineages of fish parasites, or on fish parasite taxa that are well-represented in African freshwater bodies, are quite rare, with some notable exceptions, *e.g.* the morphology-based phylogeny of lernaeid copepods by Ho (1998). Therefore, there is no comprehensive view at present of the evolution of many of the taxa covered in this book. Another constraint is that the fossil record of parasites worldwide is often patchy or non-existent and rarely taken into consideration, despite its obvious potential, *e.g.* in developing a timeframe for parasite evolution (De Baets *et al.* 2015; Leung 2017).

A number of systematic studies on the tapeworms of African freshwater fishes have used molecular data to assess phylogenetic relationships and potential intraspecific variation (*e.g.*, Schaeffner *et al.* 2011; Kuchta *et al.* 2012). A pattern of low species richness, relatively narrow host-specificity and a wide geographical range, seems to emerge. Conversely, there has been little research into the molecular phylogeny and intraspecific genetic diversity of African fish acanthocephalans (Amin *et al.* 2016) and examination of these aspects could be worthwhile. For example, *Acanthogyrus tilapiae* has a broad host range among cichlids (Amin *et al.* 2008).

Indeed, for the study of host-specificity, in-depth understanding of parasite (molecular) taxonomy is a necessity, as several seemingly generalist species have proven to be incorrectly identified or to consist of complexes of closely related but more host-specific species (Pouyaud *et al.* 2006; Smit *et al.* 2014). Likewise, the current knowledge of digeneans infecting African freshwater fishes appears too fragmented to identify conclusive evolutionary patterns (see Scholz *et al.* 2016).

The scarce molecular work has mainly focused on diplostomids and clinostomids, with some recent advances in barcoding and classification (*e.g.*, Chibwana *et al.* 2013; Caffara *et al.* 2017). In the absence of reliable morphological characters for species-level identification in non-adult stages, such genetic work has also facilitated species delineation (Otachi *et al.* 2015) and life cycle reconstruction (Chibwana *et al.* 2015). When sequence data from a wider host and geographic range are included, patterns do emerge, *e.g.* on the link of infection site (eye lens or other tissues) with diplostomid evolution and host-specificity (Locke *et al.* 2015).

For monogeneans, with several relatively well-studied genera that are endemic or mostly constrained to Africa, some patterns in host use, host-specificity and speciation mechanisms can be discerned on an African scale. For example, congruence between the phylogeny of representatives of *Cichlidogyrus* and their cichlid hosts has been shown several times, although the speciation mechanisms underlying this pattern seem to differ between cichlid-monogenean systems (Mendlová *et al.* 2012; Vanhove *et al.* 2015). Relatively extensive sampling, especially of certain groups of cichlids, also indicated correlations between host genetic diversity and parasite species richness (Pariselle *et al.* 2003; Grégoir *et al.* 2015), and between host-specificity and host behaviour, phylogeny or ecology (Mendlová & Šimková 2014; Kmentová *et al.* 2016).

As with other parasite taxa, we are reminded that taxon sampling remains of the utmost importance. Whereas earlier work suggested that the morphology of the attachment organ in representatives of *Cichlidogyrus* was poorly influenced by host choice (Vignon *et al.* 2011), the addition in a phylogenetic reconstruction of a species that resulted from a distant host-switching event indicated an adaptative component to haptor morphology (Messu Mandeng *et al.* 2015). Unexpectedly distant host-switches are known from other African dactylogyridean monogeneans, such as *Quadriacanthus* (see Nack *et al.* 2016).

The monogeneans of other African freshwater fish families have recently also become the subject of molecular phylogenetic research, including those infecting catfishes (Francová *et al.* 2017) and cyprinids (Šimková *et al.* 2017). The above-mentioned work on the evolution of African monogeneans deals mainly with representatives of the Dactylogyridea. A different picture emerges for the gyrodactylids. The many endemic African lineages and genera are attractive subjects for evolutionary parasitology (*e.g.*, Přikrylová *et al.* 2017) and mechanisms deemed important in gyrodactylid speciation, such as hybridisation and host-switching, have been demonstrated for their African representatives (Barson *et al.* 2010; Přikrylová *et al.* 2013; Zahradníčková *et al.* 2016).

In addition to studying the patterns and processes underlying parasite biodiversity, evolutionary parasitology also considers the hypotheses that parasites may act as tags for the taxonomy and biogeography of African fishes (*e.g.*, Paugy *et al.* 1990; Barson *et al.* 2010; Pariselle *et al.* 2011; El Hafidi *et al.* 2013), or as drivers of the diversification of their hosts. The latter aspect has been explored for the African Great Lakes, comparing the parasite communities of different cichlid species or populations, and linking these to immunogenetics, trophic specialisation and sexual selection (Maan *et al.* 2006, 2008; Blais *et al.* 2007; Raeymaekers *et al.* 2013; Hablützel *et al.* 2014, 2016, 2017).

There are several practical applications of evolutionary and ecological fish parasitology. For instance, fish parasites may be used as indicators for anthropogenic stressors such as pollution (Sures *et al.* 2017). This approach has also been taken for African fishes (*e.g.*, Madanire-Moyo *et al.* 2012).

Greater knowledge of the diversity and speciation of African fish parasites will increase understanding of their host range and host-specificity. This is important in view of the co-introduction of parasites that potentially accompanies the translocation of fishes for aquaculture or fisheries (Vanhove *et al.* 2016). Alien parasites in Africa have already caused mass fish mortalities in hatcheries (Hecht & Endemann 1998). An overview for South Africa by Smit *et al.* (2017) lists 23 alien fish parasites, of which seven are considered invasive. The authors suggest that a lack of monitoring is the most likely explanation of this relatively modest number.

There have not been many reports of fish diseases in Africa and they have been given little attention. However, fish parasites are expected to gain importance with the further development of aquaculture (Hecht & Endemann 1998). This requires increased efforts to protect fish health but, as pointed out by Akoll *et al.* (2012a), African countries may lack the capacity to control fish health and implement biosecurity systems and hence, more awareness of fish parasites and their ecology is important to Africa. Although parasite infections do not always demonstrably harm their fish hosts (*e.g.*, Ndeda *et al.* 2013), Paperna (1996) lists numerous cases where fish parasites have detrimental effects, especially in aquaculture. It is therefore not surprising that Akoll *et al.* (2012b) emphasise the risks of fish parasites for the productivity and sustainability of African aquaculture. In addition, though seldom reported, there are potential dangers to fish populations in nature (*e.g.*, Marshall & Cowx 2003 discuss a tapeworm infecting an economically important cyprinid in Lake Victoria), to fisheries economics (consumer rejection of infected fish: Kabunda & Sommerville 1984) and to human health (fish-borne zoonoses: Florio *et al.* 2009). Building capacity for pathogen monitoring, identification and risk analysis in developing countries is vital for aquatic health management (Bondad-Reantaso *et al.* 2005) and for any integrated approach to health (Keune *et al.* 2017). It is hoped that this book can contribute to this endeavour.

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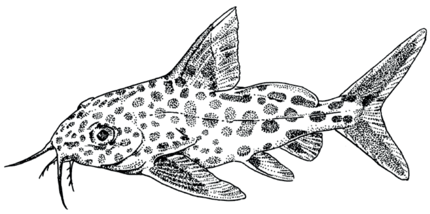
PART 7

PROSPECTS AND RECOMMENDATIONS



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The present book aims to summarise the current state of knowledge of the diversity of the parasites of freshwater fishes in Africa. It does not attempt to be an exhaustive monograph covering in detail every fish parasite group reported from freshwater ray-finned fishes (Actinopterygii) in Africa. The systematic chapter documents the progress achieved and reveals the existing gaps in our attempts to better define the parasite fauna of a continent that encompasses the whole Ethiopian (Afrotropical) zoogeographical region and the far southwestern part of the Palaearctic region. A simple comparison of the number of species and genera of helminth parasites listed in the current publication with those in two earlier checklists (Khalil 1971; Khalil & Polling 1997) shows a considerable increase over the last two decades. However, there is still much to discover.

The information summarised in the systematic chapter clearly demonstrates, as did the checklists of Khalil (1971) and Khalil and Polling (1997), how unbalanced the present knowledge of African fish parasites is in several aspects. First, attention paid to individual groups has been uneven, with the data available on protists and helminth larvae the most limited. Second, there are conspicuous differences in the number of studies on different fish hosts, with cichlids and catfishes the most intensively studied groups. Third, and probably most importantly, there are marked differences in the amount of data accumulated in the different regions and countries of Africa. The information for some regions such as the Nile Basin (Egypt and Sudan), African Great Lakes (especially Lake Victoria and Lake Tanganyika) and South Africa is reasonably representative but there are vast areas of Africa for which there is limited or no data.

Another important message that the editors would like to emphasise is in the methodological chapters. Though they may appear at first sight too succinct, the authors of individual chapters have used their long experience in parasite studies to present the most relevant information for processing fish parasites correctly and for the application of the best methods to ensure that valuable parasite material is available for subsequent morphological, taxonomic, histopathological, ecological, genetic and other studies. The book will serve as a guide to all principal steps including catching fish, their examination for different groups of unicellular and metazoan parasites and the appropriate processing of the parasites found.

The remaining chapters complement these two core parts of the book to provide a compendium of useful information for both advanced fish parasitologists and inexperienced beginners. However, the editors are aware that the present book represents only an initial attempt to support the advancement of fish parasitology in Africa. Therefore, any critical comments and suggestions for additional information are welcome and should be sent to the Editors.

The future progress of African fish parasitology is difficult to predict as it depends on many factors, such as the economic situation in individual countries, sufficient funding for basic and applied research, the availability of human resources including the development of a new generation of fish parasitologists trained in the best methods of modern biological and veterinary research, and also in assessing ecosystem health and richness of aquatic habitats.

Several recommendations important for the further development of studies on the fish parasites in Africa are suggested below:

1. Any study of fish parasites should be preceded by a comprehensive literature search to avoid repetition of work already done.
2. The exact purpose of a new study should be established before the start. The availability of facilities, expertise and funding should be assessed realistically. A study that is too ambitious may result in methodological mistakes.
3. Only fresh or recently dead fish should be examined, including those in outbreaks of mass mortality in aquaculture.
4. Suitable methods and proper equipment should always be used, especially good quality optics and chemicals (as fixatives, etc.).
5. There should be a focus on quality rather than quantity. It is better to examine fewer fish correctly than many fish incorrectly. Inadequate examination will result in unreliable data.
6. Faunal surveys should be the first step in exploring fish parasites in a given region and vouchers of parasites found should always be preserved, preferably in an internationally accessible collection.
7. International cooperation with experts on individual parasite groups and research areas is strongly recommended (and often inevitable) but only properly processed parasite material should be used. Avoid making requests for the identification of parasites from poor quality pictures of improperly processed material.

The present book is the outcome of a long term international cooperation of more than two generations of fish parasitologists and other specialists from South Africa and several European countries. We hope that our efforts will facilitate the further advancement of fish parasitology in the African continent.

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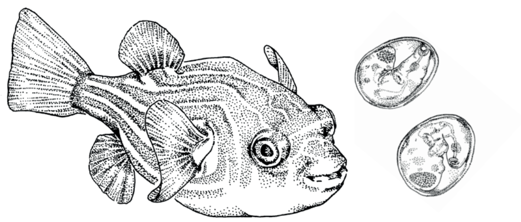

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