Metazoan Parasites of Fishes: Synoptic Information and Portal to the Literature for Aquarists

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Introduction
Although wild fish are commonly infected by metazoan parasites, it is only under unusual circumstances that they destabilize host populations in nature. However, when fishes are confined or clustered in high densities some metazoans can cause morbidity and mortality. This has obvious implications regarding the aquarium industry. Because methods used to inspect and treat fish for parasitic infections are time consuming, expensive, and they can negatively impact fish health, it is important to understand the risk to fish health that various parasitic infections represent. When risk is high, it is important to understand the conditions that facilitate disease, as well as treatment alternatives. Identification of parasites and an understanding of their biology are key steps in this process. With this in mind, a day-long workshop on metazoan parasites of fishes was conducted at the 2001 Eastern Regional Meeting of the American Zoo and Aquarium Association, with the authors of this report serving as the instructors. Because of time constraints, only the following groups of parasites were discussed: monogeneans, cestoidarians, digeneans, acanthocephalans, nematodes, leeches, branchiruans, copepods, and isopods. Each lecture stressed parasite morphology and anatomy, identification of parasites and diagnosis of infections, parasite life cycles, host-parasite interactions (including disease caused by parasites), and special considerations linked to captive environments, including quarantine, husbandry practices, prophylaxis, and treatment.

This report is a distillation of information presented at the aforementioned workshop. Because it is impossible to fully address this topic in the space provided, we urge those interested in more information to consult the references cited herein as a portal to the literature. However, readers should be aware that many important references pertaining to specific matters regarding the diverse fauna dealt with here have appeared both before and since the often more encompassing references we have chosen to cite.

Monogenea
Systematic summary and distribution in nature: Class Monogenoidea (phylum Platyhelminthes), 3 subclasses, Polyonchoinea, Polystomatoinea, and Oligonchoinea (Boeger and Kritsky, 1993) comprised of about 50 families and 2,200 nominal species. See Whittington (1998) for estimates of monogenean diversity. Monogeneans infect marine and freshwater fishes, amphibians, freshwater turtles, parasitic isopods and copepods, and hippopotami, and as a group they are considered ectoparasites. See Yamaguti (1963a), Schell (1985), Hendrix (1994), and Hoffman (1999) for taxonomic information regarding many species of monogeneans.

Biology summary: For descriptions of general anatomical features of monogeneans see Schell (1985), Hendrix (1994), Kearn (1998), and Hoffman (1999). Body with distinct anterior and posterior ends; anterior with sucker or set of suckers that facilitate feeding or temporary attachment during locomotion; posterior with primary attachment organ, i.e., haptor, consisting of lappet, sucker, or set of suckers. Most species possess a haptor with hooks. All known species are obligate parasites, hermaphroditic, and are probably capable of self-fertilization. Some monogeneans feed on mucous, gill epithelium, or blood. For additional information on monogenean biology see Bychowsky (1961) and Kearn (1998).

Life-cycle summary: Monogeneans exhibit direct life cycles. Eggs are produced singly, but reproductive output can be high because egg production can be continuous. Eggs are usually ejected from the uterus and sink; however, the eggs of some species hatch within the uterus. *Gyrodactylus* spp. are viviparous (see Cone 1995; Kearn, 1998). The infective stage of monogeneans, i.e., the oncomiracidium, may be a ciliated form that swims or a non-ciliated form that crawls. After colonizing the host, oncomiracidia begin larval development. Larvae may migrate from the colonization site to elsewhere on the host before sexual maturation occurs. Some monogeneans may use phoretic hosts (see Whittington, 1990; Bullard et al., 2000a).

Host and attachment site specificity: Monogeneans typically exhibit a high degree of host specificity. One very notable exception is *Neobenedenia melleni*, a pathogenic species that appears capable of infecting almost any marine teleost under the right circumstances (see Bullard et al., 2000b). Most monogeneans are ectoparasites and they have been collected from the buccal cavity, urinary bladder, cloaca, eyes, skin, fins, gills, and olfactory sacs of fishes. Adult worms typically exhibit a relatively high degree of site specificity while larvae and juveniles may exhibit more widespread attachment distributions. Adult site specificity may appear reduced when infections are heavy. See Yamaguti (1963a), Hendrix (1994), and Hoffman (1999) for host and infection site information for many species of monogeneans.

Mobility: Adults are capable of rapid locomotion across fish skin or from gill filament to gill filament. The monogenean body is extendible and possesses anterior and posterior attachment organs, facilitating inchworm-like movements that enable worms to remain securely attached to the host during all phases of locomotion. Some species are semi-stationary, and some of these cement themselves to the host (see Roubal and Whittington 1990; Bullard et al., 2000c).

Problems caused by monogeneans: Intense monogenean infections can be associated with morbidity and mortality of fishes in aquariums and aquaculture systems. Confinement probably facilitates autoinfection and infection of new hosts. In nature, most oncomiracidia fail to acquire a host. Heavily infected fishes may flash (indicative of infections on skin or gills) or pipe (indicative of infections on gills) and they may exhibit poor overall body condition. Severe infections may promote lesions that facilitate the onset of secondary bacterial, fungal, or viral infections (Stoskopf, 1993; Cone 1995). Not all species of monogeneans will multiply rapidly on hosts maintained in captive environments; however, those that do are usually serious pathogens. Populations of viviparous species can rapidly increase to problematic levels. For more information regarding problems caused by monogeneans see Cone (1995).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes), mechanical removal using forceps is sometimes useful regarding large species that infect the external body surface, osmotic shock (freshwater or saltwater dips) is sometimes useful, formalin baths are sometimes useful for small species, water treatments using organophosphates or praziquantel are sometimes warranted. When considering treatments for monogenean infections it is important to remember that the eggs of egg-laying species can be relatively resistant to water treatments, e.g., see Thoney (1990). Thus treatments must sometimes be repeated to kill newly hatched worms before they can mature and lay eggs. It is also important to realize that eggs and oncomiracidia can remain viable away from the host for some time. Thus, treating infected fish without treating their environment may only provide temporary relief. It is also important to recognize the potential for secondary infections associated with severe monogenean infections (Cone, 1995). For more particulars on treatment see Stoskopf (1993) and Noga (1996).

Proper fixation for taxonomic study: Monogeneans should be removed from the host, placed in a petri dish with water (of a specific gravity equal to that of the water surrounding the parasite in nature). Next, a large volume of near-boiling fresh water is added to the dish immediately before adding 10% neutral buffered formalin. The end concentration of formalin should be 5%. Monogeneans are soft-bodied worms that are easily damaged with metal dissection tools, and it is sometimes best to fix them in situ by cutting the tissue surrounding the attachment site and fixing it with the worms attached. Use only glass pipettes or fine (4x0) brushes to transfer specimens. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Cestoidaria

Systematic summary and distribution in nature: Subclass Eucestoda (phylum Platyhelminthes), at least 12,000 species in 13 orders, with 10 orders restricted to aquatic hosts. Six orders include most species infecting fishes, Caryophyllidea (represented mainly in freshwater siluriforms and cypriniforms), Spathebothridea (represented in freshwater teleosts), Pseudophyllidea (represented in a variety of marine and freshwater teleosts), Proteocephalidea (commonly represented in salmonids), Tetraphyllidea (represented as adults in elasmobranchs), and Trypanorhyncha (represented as adults in elasmobranchs). Most important features used for identification are the shape and structures of the scolex, i.e., the attachment organ, and arrangement of the gonads within the proglottids, i.e., the segments. For detailed information on cestode taxonomy see Yamaguti (1959), Khalil et al. (1994), and Hoffman (1999).

Biology summary: For descriptions of cestode morphology see Khalil et al. (1994) and Yamaguti (1959). Body typically composed of scolex, and a series of segments, i.e., the strobila, each containing a set of gonads. There is no mouth or gut, and feeding is by direct absorption across the syncitial tegument. Most tapeworms are hermaphrodites and all tapeworms are obligate endoparasites. They may occur as adults in the gut, or as larvae in the gut or in other tissues. Most problems caused by cestode infections in definitive hosts stem from lesions caused by attachment or blockage of the gut. Basic information about the biology of cestodes may be found in Wardle and Mcleod (1952).

Life-cycle summary: Cestodes exhibit indirect life cycles. Copulation takes place in the definitve host and gravid proglottids release eggs that are voided in the feces. Occasionally proglottids break free from the strobila and are passed into the environment where they rupture. Eggs designed to hatch liberate a ciliated larva, i.e., the coracidium, whereas those designed not to hatch contain a non-ciliated larva, i.e., the oncosphere. Copepods are frequently the first intermediate host and they become infected by eating eggs or coracidia. Development in intermediate hosts typically takes place outside the gut. Life-cycle details regarding the majority of fish-infecting cestodes are lacking. Hoffman (1999) provides some information regarding species infecting North American freshwater fishes.

Host and attachment site specificity: Host specificity varies according to life-cycle stage, but is poorly understood. In definitive hosts, specificity is probably mediated by functional aspects of the scolex, a structure that frequently shows species-specific modifications. Adults are typically found in the intestine of teleosts or the spiral valve of elasmobranchs. Larvae may be found in the gut, body cavity, musculature, or other organs of fishes, molluscs, crustaceans, or any number of other invertebrate taxa. For information regarding host associations of cestodes that infect fishes see Yamaguti (1959), Khalil et al. (1994), and Hoffman (1999).

Mobility: Tapeworms are largely sedentary but they may undertake migrations within the gut in response to feeding status of the host or competition with other parasites.

Problems caused by cestodes: With the exception of several species that are known pathogens, e.g., the widespread species *Bothriocephalus acheilognathii* ; see Dove and Fletcher (2000), cestodes rarely cause morbidity and mortality in captive fish populations because their life cycles are too complex to be easily completed in captivity and the presence of a few worms typically does not promote disease. Sometimes, however, fishes can enter quarantine with heavy cestode infections that they acquired in the wild or at aquaculture facilities and these may or may not be problematic. The attachment activities of cestodes can cause tissue damage (including blood loss), they can block the gut, and their sequestering of host food and nutrients can cause nutritional deficiencies. Dick and Choudhury (1995a) provided information on some pathogenic cestodes.

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes) and praziquantel and niclosamide administered orally are the drugs of choice. When considering treatments for cestode infections it is important to remember that some species may occur outside the gut and killing them can cause a massive immune challenge to the host that causes acute disease. Thus, only species known to cause problems should be treated. For more particulars on the treatment of cestodes see Stoskopf (1993) and Noga (1996).

Proper fixation for taxonomic study: Relax specimens in cold tap water for 2-5 minutes, then pipette into near-boiling saline and allow to stand for a minute or so before transferring into 10% neutral buffered formalin. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Digenea

Systematic summary and distribution in nature: Subclass Digenea (phylum Platyhelminthes), 10 orders, and about 71 families. For information on digenean taxonomy see Yamaguti (1958). Digeneans are the most common parasitic platyhelminths, and they infect a broad range of fresh water, brackish water, marine, and terrestrial vertebrates and invertebrates. See Yamaguti (1958) and Yamaguti (1975) for information regarding the hosts and habitats of digeneans.

Biology summary: For descriptions of morphological features of digeneans see Yamaguti (1958), Schell (1985), and Kearn (1998). Adult body with distinct anterior and posterior ends. Anterior end with or without a well-developed oral sucker. Medial region of body with or without muscular sucker, i.e., acetabulum. Body spines present or absent. Digeneans are obligate parasites and most species are endoparasites. Most taxa are hermaphroditic. For more information regarding the biology of digeneans see Kearn (1998).

Life-cycle summary: Digeneans exhibit indirect life cycles. Worms copulate while they are in the definitive host. Eggs are released into the external environment and upon hatching they liberate a free-swimming, ciliated larva, i.e., the miracidium. The miracidium typically penetrates a mollusc (the typical first intermediate host) and asexual reproduction commences: a sporocyst (germinal sac) and/or redia (functionally similar to sporocyst, but with a pharynx and rudimentary gut) may occur. Second generations of sporocysts and rediae may develop, i.e., daughter sporocysts and daughter rediae. The second free-swimming stage of the parasite, i.e., the cercaria, is shed from the first intermediate host. Cercariae have eyespots and flagellum-like tails that they use for swimming. The cercaria penetrates the second intermediate host (sometimes a fish) and develops into an encysted or un-encysted larva, i.e., the metacercaria. Typically, the second intermediate host is eaten, at which time the metacercaria invades the digestive tract of the definitive host (sometimes a fish) and matures. Metacercariae may persist in alternate hosts (sometimes a fish) before being eaten by the definitive host. Not all life cycles depend on trophic interactions between hosts; some exclude an encysted metacercaria and a second intermediate host and thus they depend on the proximity of definitive hosts to intermediate hosts. For concise summaries of life cycles for particular digeneans see Yamaguti (1975).

Host and attachment site specificity: Relative to other platyhelminths, digeneans exhibit a low degree of host specificity regarding their definitive hosts; however, many species exhibit greater specificity for their intermediate hosts. As a group, digeneans infect nearly every imaginable site in the definitive host, including the body cavity, gut, skin, gill, kidney, liver, heart, pericardium, eye, gall bladder, fins, muscle, etc. As species, digeneans exhibit relatively high levels of attachment site specificity. For information regarding host associations and attachment locations of digeneans that infect fishes see Yamaguti (1958, 1975) and Hoffman (1999).

Mobility: Digeneans generally move in a somewhat awkward inchworm-like fashion by stretching and contracting while alternating attachment of the oral and ventral suckers.

Problems caused by digeneans: Digeneans are often unproblematic regarding the health of aquarium fishes because their life cycles are too complex to be completed in captivity and the presence of a few worms typically does not cause disease. Sometimes, however, fishes that enter quarantine can be infected with heavy worm burdens that they acquired in the wild or especially at aquaculture facilities. At aquaculture facilities where life cycles can often be easily completed, miracidia, cercariae, and metacercariae of some species are known to cause problems. For additional information on diseases caused by digeneans see Stoskopf (1993), Paperna (1995), Noga (1996), and Hoffman (1999).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes) and praziquantel administered orally is the drug of choice to treat digeneans. When considering treatments for digenean infections it is important to remember that some species may occur outside the gut, e.g., metacercariae of many species, and the killing them can create a massive immune challenge to the host that causes acute disease. Thus, only species known to cause problems should be treated. In some instances, larval worms (metacercariae) that are encysted in superficial tissues can be surgically removed. For more particulars on the treatment of digeneans see Stoskopf (1993), Paperna (1995), and Noga (1996).

Proper fixation for taxonomic study: For digeneans attached within the digestive tract, remove the tract from host and place it in a bag. With a pair of sharp scissors, make a longitudinal incision through the wall of the tract, pour nearly boiling water into the bag, and then add an equivalent volume of 10% neutral buffered formalin into the bag and gently shake. The hot water relaxes the worms before fixation and the resulting concentration of fixative is roughly 5%. For digeneans elsewhere in the body, dissect out the worms and relax and fix as described above. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Acanthocephala

Systematic summary and distribution in nature: Phylum Acanthocephala, 3 orders, Archaeacanthocephala, Palaeacanthocephala, and Eoacanthocephala, the latter two orders occurring in fish hosts. For information on acanthocephalan taxonomy see Yamaguti (1963b), Nickol (1995), and Hoffman (1999).

Biology summary: For descriptions of acanthocephalan morphology see Yamaguti (1963b) and Nickol (1995). Body composed of a trunk with an eversible spiny proboscis. There is no true body cavity, but a fluid-filled pseudocoelom is present. There is no gut, and feeding is by absorption across a metabolically active syncitial tegument. Sexes are separate and the reproductive system is complex and taxonomically useful. Acanthocephalans are obligate endoparasites. For more information on acanthocephalan biology see Nickol (1995).

Life-cycle summary: Acanthocephalans exhibit indirect life cycles that always involve an arthropod first intermediate host. Copulation takes place in the definitive host and gravid females lay characteristic bipolar eggs that are voided in the feces. Larvae, i.e., acanthellae, usually infect first intermediate hosts that consume eggs. Amphipods most often serve as aquatic intermediate hosts for acanthocephalans and the larval stage inside these hosts is called a cystacanth. Definitive hosts become infected when they consume infected intermediate hosts. Paratenic fish hosts are known and in some instances obligate second intermediate hosts exist. For more information on acanthocephalan life history see Nickol (1995).

Host and attachment site specificity: Levels of host specificity vary among species of acanthocephalans. Adult acanthocephalans are generally found in the intestine of their definitive hosts, sometimes with a preference for the rectum. Cystacanths in paratenic fish hosts are most often found in visceral organs or the body cavity. The level of host specificity of cystacanths is generally lower than that of corresponding adults. For more information on host affiliations and attachment locations of various acanthocephalans see Yamaguti (1963b) and Hoffman (1999).

Mobility: Acanthocephalans are among the most sedentary of the parasitic worms and many species have near-permanent or permanent attachment within the gut. The proboscis or trunk may anchor deeply in the mucosa or perforate the gut completely, eliciting a proliferative response that serves to permanently anchor the worm.

Problems caused by acanthocephalans: Acanthocephalans are usually unproblematic regarding the health of aquarium fishes because their life cycles are too complex to be easily completed in captivity, and the presence of a few worms typically does not cause disease. Sometimes, however, fishes can enter quarantine with heavy acanthocephalan infections that they acquired in the wild or at aquaculture facilities. For industries like the aquarium industry that require exhibit fishes to appear attractive, infections of some acanthocephalans, e.g., species whose bodies can dangle from the rectums of their hosts, can be problematic from a cosmetic perspective. In addition to tissue destruction associated with attachment, some acanthocephalans can block the lumen of the organ they inhabit and this may have a deleterious affect on the host. See Nickol (1995) for information regarding disease caused by acanthocephalans in wild-caught fishes.

Prevention and treatment of infections: Treatments for acanthocephalans have not been formulated. As always, quarantine is the best defense (with close observation of new fishes). Treatments used for nematodes may be effective, e.g., oral delivery of benzimidazole compounds. When considering treatments for acanthocephalan infections it is important to remember that some acanthocephalans may be encysted in the flesh and killing them may create a serious immune challenge for the host that causes acute disease.

Proper fixation for taxonomic study: Ideally, acanthocephalans should be fixed with the proboscis extended. Although ideal methods of fixation may vary for different species, small species can usually be relaxed for 2-5 minutes in tap water before pipetting into near-boiling saline (if the worms burst, try pipetting them directly into 10% neutral buffered formalin) and subsequently fixing them in 10% neutral buffered formalin. For larger acanthocephalans, pipette them from the tap water directly into 10% formalin. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Nematoda

Systematic summary and distribution in nature: Phylum Nematoda is an enormous group with at least 250 families, approximately half of which are animal parasites. Seventeen families have representatives that are parasites of fishes, and these taxa probably represent secondary invasions of aquatic habitats from the terrestrial environment. For information on nematode taxonomy see Yamaguti (1961), Moravec (1994), Hoffman (1999), and Anderson (2000).

Biology summary: For descriptions of nematode morphology see Yamaguti (1961), Hoffman (1999), and Anderson (2000). Body typically elongate and tubular, with tri-radiate symmetry. Characterized by a non-living proteinaceous cuticle that forms a hydrostatic skeleton that holds the body form while maintaining a high internal pressure within the pseudocoelom. A muscular pharynx is present at the anterior end to force food in against the aforementioned pressure, and sphincters are present on other orifices to prevent the loss of body contents. Sexes are separate, and molts occur between life-history stages. Nematodes that infect fishes are obligate endoparasites. For more information on nematode biology see Dick and Choudhury (1995b) and Anderson (2000).

Life-cycle summary: Nematodes exhibit indirect life cycles involving 4 larval (L1-L4) and 1 adult (L5) stages. Copulation takes place in the definitive host and gravid females lay eggs that are voided in the feces. Eggs may or may not hatch before infecting the first intermediate host and larvae may or may not molt within the egg such that the larva released may be an L1, L2, or L3. Larvae infect intermediate hosts (often copepods) either when eggs or larvae are eaten and development within the first intermediate host is usually parenteral. Several intermediate hosts may be involved in the life history of nematodes that are parasites of fishes. Transmission between hosts is affected by predation. For more information on nematode life history see Dick and Choudhury (1995b), Hoffman (1999), and Anderson (2000).

Host and attachment site specificity: Levels of host specificity exhibited by nematodes vary widely but it is low in several problematic species. As a group, adult nematodes can potentially be found anywhere in the host; however, they are often found in the gut. Some dracunculoids are found in the brain, eyes, visceral organs, or in the musculature, e.g., see Benz and Pohley (1980) and Adamson et al. (1987). Larval nematodes are typically coelozoic or histozoic. Larval ascarids (family Anisakidae) and philometrids (family Philometridae) are sometimes found in large numbers in the mesenteries and visceral and reproductive organs of marine fishes, e.g., see Benz et al. (1987). For more information on host affiliations of nematodes as well as attachment locations see Yamaguti (1961), Hoffman (1999), and Anderson (2000)

Mobility: Nematodes move by characteristic dorso-ventral undulations called thrashing and they can usually move well in viscous body fluids. Larval nematodes are often encapsulated by the host (representing a resting phase for the parasite) and as such their movement is restricted.

Problems caused by nematodes: A small number of nematode species are known to cause morbidity and mortality in captive fish populations. Nematode feeding and attachment can directly damage tissues (including blood loss) and can lead to loss of condition or wasting. For more information on diseases caused by nematodes that infect fishes see Dick and Choudhury (1995b).

Prevention and treatment of infections: Nematodes are usually unproblematic regarding the health of aquarium fishes because their life cycles are too complex to be easily completed in captivity and the presence of a few worms typically does not promote disease. Sometimes, however, fishes can enter quarantine with heavy nematode infections that they acquired in the wild or at aquaculture facilities. Quarantine is the best defense (with close observation of new fishes). Taxa whose presence may prompt treatment include *Camallanus cotti* and *Spriocamallanus* spp. in freshwater fishes, *Spirocamallanus* spp. in marine fishes, and *Proleptus* spp. in elasmobranchs. Treatment may kill encysted nematode larvae, and if larvae are numerous, this may create an immune challenge for the host that causes acute disease. Treatments are typically based on the oral use of benzimidazole compounds. For more particulars on the treatment of nematodes see Stoskopf (1993) and Noga (1996).

Proper fixation for taxonomic study: Fix nematodes by pipetting them into a very small quantity of 95% glacial acetic acid, and then immediately transfer them into 10% neutral buffered formalin. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Hirudinea

Systematic summary and distribution in nature: Class Hirudinea (phylum Annelida), 2 subclasses containing fish parasites, Acanthobdellidea with 2 species and Euchirudinea (orders Arhynchobdellida and Rhynchobdellida) with many species. Acanthobdellids are found in fresh water, euchirudineads parasitic on fishes are found in marine and fresh water. See Sawyer et al. (1975), Sawyer (1986), Burreson (1995), and Hoffman (1999) for taxonomic information regarding many species of leeches that infect fishes.

Biology summary: For descriptions of general anatomical features of leeches see Sawyer (1986) and Burreson (1995). Body segmented, clitellum present, acanthobdellids with setae on cephalic segments, arhynchobdellids, or "jawed leeches," with pharynx containing toothed jaw-like apparatus, rhynchobdellids with proboscis. Anterior sucker associated with mouth and posterior sucker present. Hermaphroditic. Leeches that infect fishes are generally considered to be obligate parasites. For more information on leech biology see Sawyer (1986) and Burreson (1995).

Life-cycle summary: Leeches exhibit direct life cycles. Copulation occurs on or off host. Gravid individuals typically leave host to produce cocoons. Parent may or may not brood juveniles and transport them to the next host. For more information on leech life history see Sawyer (1986) and Burreson (1995).

Host and attachment site specificity: Host specificity varies among leech species. As a group, leeches are considered ectoparasites and they can be found virtually anywhere on the external body surface of fishes, e.g., general body surface, in mouth, in branchial chamber, in cloaca, and in olfactory sacs. Under some circumstances they can be endoparasites, e.g., see Moser and Anderson (1977). Casual observations of infections at aquariums and aquaculture facilities suggest that under circumstances of heavy infections leeches can display more widespread distributions over their hosts while in instances of light infections species often seem to realize what could be considered typical attachment locations on their hosts. For records of leech infections on various hosts see Sawyer et al. (1975), Sawyer (1986), Burreson (1995), and Hoffman (1999).

Mobility: Leeches often move in a looping inchworm-like fashion. While they often remain stationary on the host for long periods using their posterior sucker, they are capable of relatively rapid movements. Some euhirudineads are capable of swimming and even those species that cannot swim are capable of switching hosts.

Problems caused by leeches: Leeches can cause morbidity and mortality in captive fish populations. As direct agents of disease their feeding and attachment activities can cause tissue damage (including blood loss) and osmoregulatory problems, and they can vector pathogens, e.g., blood-born protozoa; see Burreson (1995). As indirect promoters of disease they cause lesions and stress that can predispose hosts to opportunistic pathogens, e.g., bacteria and fungi. For more information on problems caused by leeches see Burreson (1995).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes), mechanical removal using forceps is sometimes effective, osmotic shock (freshwater or saltwater dips) is sometimes helpful with small leeches, water treatments using organophosphates are often necessary. When considering treatments for leech infections it is important to remember that these parasites can detach from and live away from their hosts for significant periods and that they deposit cocoons on substrates. Thus, treating fishes infected with leeches without treating their environment may only provide temporary relief. It is also important to recognize the potential for secondary infections associated with severe leech infections. For more particulars on treatment see Stoskopf (1993) and Noga (1996).

Proper fixation for taxonomic study: Leeches, especially thick species, must be fixed properly or they can be useless for taxonomic study. Proper preparation for taxonomic study involves starving bloated leeches (if necessary), relaxation using menthol crystals, and fixation in 8-10% neutral buffered formalin or in alcohol-formalin-acetic acid (AFA). For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential. Leeches typically stay alive for long periods off the host and hence specimens can sometimes be sent alive to specialists for proper handling.

Branchiura
Systematic summary and distribution in nature: Subclass Branchiura (class Crustacea, phylum Arthropoda), 1 family, Argulidae, containing about 150 species in 4 genera: *Argulus* (cosmopolitan distribution in fresh and salt water), *Dolops* (South America, Africa and Tasmania in fresh water), *Chonopeltis* (Africa in fresh water), and *Dipteropeltis* (South America in fresh water). For information on branchiuran taxonomy see Wilson (1903), Meehean (1940), Yamaguti (1963c), Cressey (1976, 1978), and Kabata (1988).

Biology summary: For descriptions of branchiuran morphology see Martin (1932), Meehean (1940), Sutherland and Whittrock (1986), Overstreet et al. (1992), and Gresty et al. (1993). Body composed of a carapace and thorax (each ventrally bearing true appendages) and abdomen. Sexes are separate. Molts occur between life-history stages. Branchiurans are obligate ectoparasites. *Argulus* species possess an oral stylet that is used to inject enzymes into the host to begin the digestion process externally. Dentiferous mandibles are used by branchiurans to rasp host tissue into the mouth tube. For more information on branchiuran biology see Wilson (1903), Schram (1986), and Lester and Roubal (1995).

Life-cycle summary: Branchiurans exhibit direct life cycles. Copulation takes place on or off the host. Gravid females leave the host to lay eggs on hard substrates, e.g., stones, sticks, aquarium props, aquarium glass or acrylic. Larvae can infect hosts immediately upon hatching. For more information on branchiuran life history see Wilson (1903), Shimura et al. (1981), and Lester and Roubal (1995).

Host and attachment site specificity: Most branchiruans are parasites of fishes, but records from amphibians also exist (see Yamaguti, 1963c). Host specificity seems relatively low for branchiurans, e.g., see LaMarre and Cochran (1992). However, a portion of this phenomenon may stem from the fact that these parasites can rapidly detach from one host and attach to another, thus confounding efforts to assess host specificity using some capture methods. Branchiurans are generally found on the general body surface or in the mouth or branchial chambers. For records of branchiuran infections on various hosts see Wilson (1903), Meehean (1940), Yamaguti (1963c), Cressey (1976, 1978), Kabata (1988), and Hoffman (1999).

Mobility: Branchiurans are excellent swimmers and adults and larvae can easily transfer among hosts. They can also live away from the host for significant periods and can detach from the host when disturbed, e.g., when host is netted.

Problems caused by branchiurans: Branchiurans can cause morbidity and mortality in captive fish populations. As direct agents of disease their feeding and attachment activities can cause tissue damage (including blood loss) and osmoregulatory problems, and they can vector pathogens, i.e., viruses, bacteria, and nematodes; e.g., see Cusack and Cone (1986). As indirect promoters of disease they cause lesions and stress that can predispose hosts to opportunistic pathogens, e.g., bacteria and fungi. For more information on problems caused by branchiurans see Kabata (1970) and Lester and Roubal (1995).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes), mechanical removal using forceps is sometimes effective, osmotic shock (freshwater or saltwater dips) is sometimes effective, water treatments using organophosphates or diflubenzuron are sometimes warranted. When considering treatments for branchiuran infections it is important to remember that these parasites can detach and live away from their hosts for significant periods and that they deposit eggs on hard substrates. Thus, treating fishes infected with branchiurans without treating their environment may only provide temporary relief. It is also important to recognize the potential for secondary infections associated with severe branchiuran infections. For more particulars on treatment see Stoskopf (1993) and Noga (1996).

Proper fixation for taxonomic study: Fix directly in 10% neutral buffered formalin or 70% ethanol. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

Copepoda

Systematic summary and distribution in nature: Subclass Copepoda (class Crustacea, phylum Arthropoda), 3 orders hold most species that are parasites of fishes, Siphonostomatoida with well over 2,000 nominal species that are parasites of fishes, Poecilostomatoida and Cyclopoida each with far fewer species. Siphonostomes that are parasites of fishes are mainly marine, but 2 families (Caligidae and Lernaeopodidae) have a few representatives that have invaded fresh water. Poecilostomes that are parasites of fishes, i.e., ergasilids, chondracanthids, bomolochids, and their close allies, are found in both marine and freshwater environments. Cyclopoids that are parasites of fishes, i.e., lernaeids, are found in fresh water. For information on copepod taxonomy see Yamaguti (1963c), Kabata (1979), and Huys and Boxshall (1991).

Biology summary: For descriptions of copepod morphology see Yamaguti (1963c), Kabata (1979), and Huys and Boxshall (1991). Tremendous variation in body form is presented by this group. Body typically composed of cephalothorax, thorax, genital complex, and abodomen. Cephalothorax may ventrally bear the following appendages: antennules, antennae, mandibles, maxillules, maxillae, maxillipeds, 1-3 pairs of thoracic legs. Sexes are separate and may or may not look generally similar. Molts occur between life-history stages. Copepod parasites of fishes are obligate parasites; however, some early life-history stages are free-living but do not feed, and males of some species, e.g., *Ergasilus* spp., may not be associated with a host. For more information on copepod biology see Kabata (1970, 1979, 1981), Schram (1986), Huys and Boxshall (1991), and Boxshall and Defaye (1993).

Life-cycle summary: Copepod species that are parasites of fishes exhibit direct life cycles except for pennillid species, e.g., see Perkins (1983). After copulation, females produce and retain 2 eggs sacs. Typically, the eggs hatch as a nauplius (a free-living dispersal stage). Nauplii molt into infective copepodids that colonize the host. Sexual maturity is achieved through a series of molts or in some species through a gradual metamorphosis. Many life-cycle variations exist, e.g., see Kabata (1979, 1981). In species with 2-host life cycles, the intermediate host is often a prey species or one that otherwise shares the definitive host's habitat.

Host and attachment site specificity: The host specificity of most copepod parasites of fishes appears to be high; however, some species that cause problems in aquariums or aquaculture facilities can attach to a wide range of hosts, e.g., *Lernaea cyprinacea*. Copepods that are parasites of fishes are mainly ectoparasites; however some species are mesoparasites, and a lesser number are endoparasites, e.g., see Nagasawa et al. (1998). Most copepods have what appear to be preferred attachment locations on their fish hosts, and as a group they have been collected from the general body surface, fins, cloaca, buccal cavity, eyes, gills and branchial chambers, olfactory sacs, flesh, heart, lateral-line system, and even the uterii of fishes. Adult copepods typically exhibit higher levels of attachment site specificity than their corresponding larvae, but in instances of heavy infections adult site specificity may appear reduced. See Yamaguti (1963c), Kabata (1979), and Benz and Sullivan (1994) for host and infection site information for many species of copepods.

Mobility: Copepods that are parasites of fishes range from species that are stationary on the host as adult females, e.g., *Lernaea* spp. and lernaeopodids, to others whose adult females are highly mobile and capable of transferring from host to host, e.g., caligids. The adult females of some species partially, e.g., some lernaeids, pennillids, sphyriids, kroyeriids, and eudactylinids, or entirely, e.g., *Sarcotaces* spp., embed themselves in the tissues of their hosts (see Kabata, 1970, 1979, 1984). Adult males of most species are more mobile than corresponding adult females. Larvae are capable of swimming or crawling, with the nauplius typically serving as the dispersal stage, the infective copepodid serving as the colonization stage, and the ensuing parasitic larval stages serving as the mate seeking and attachment site seeking stages. Some species can live away from the host as adults for significant periods and can detach from the host when it is disturbed, e.g., when host is netted.

Problems caused by copepods: Copepods can cause morbidity and mortality in captive fish populations although this has not been formally documented for many species. As direct agents of disease their feeding and attachment activities can cause organ and tissue damage (including blood loss) and osmoregulatory problems, and they can vector pathogens, e.g., bacteria. As indirect promoters of disease they cause lesions and stress that can predispose hosts to opportunistic pathogens, e.g., bacteria and fungi. For more information on problems caused by copepods see Kabata (1970, 1984), Boxshall and Defaye (1993), and Lester and Roubal (1995).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes), mechanical removal using forceps is sometimes possible, osmotic shock (freshwater or saltwater dips) is sometimes useful, water treatments using organophosphates or diflubenzuron are sometimes warranted. When considering treatments for copepod infections it is important to remember that some species can detach from and live away from their hosts for significant periods. Thus, treating fishes infected with copepods without treating their environment may only provide temporary relief. It is also important to recognize the potential for secondary infections associated with severe copepod infections. For more particulars on treatment see Roth et al. (1993), Stoskopf (1993), and Noga (1996).

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Isopoda

Systematic summary and distribution in nature: Order Isopoda (class Crustacea, phylum Arthropoda), 2 suborders within Isopoda contain species that are parasites or associates of fishes, Gnathiidea and Flabellifera. Gnathiids can be found in marine and estuarine habitats, while flabelliferans can infect fishes found in marine and freshwater habitats. Gnathiidea is only represented by the family Gnathiidae. Five families within Flabellifera, i.e., Cymothoidae, Aegiidae, Cirolanidae, Excorallanidae, and Corallanidae, are known parasites or associates of fishes (Bunkley-Williams and Williams, 1998). For information on isopod taxonomy see Schultz (1969), Kensley and Schotte (1989), Brusca (1980), and many contemporary references provided in Bunkley-Williams and Williams (1998).

Biology summary: For descriptions of isopod morphology see Schultz (1969), Brusca (1980), and Schram (1986). Adult body typically dorsoventrally flattened and composed of cephalon, pereon, and pleon. Cephalon bears antennules, antennae, mandibles, maxillules, maxillae, and maxillipeds. Pereon typically bears 7 pairs of pereopods, pleon bears 5 pairs of pleopods, 1 pair of uropods, and pleotelson. Sexes are separate in representatives of Gnathiidea, Aegiidae, Cirolanidae, Excorallanidae, and Corallanidae, but representatives of Cymothoidae are protandrous hermaphrodites. Isopod parasites of fishes range from taxa that are obligate parasites that remain permanently attached to the host as adults, e.g., cymothoids, to taxa that are considered facultative parasites or associates, e.g., corallanids (see Bunkley-Williams and Williams, 1998). Gnathiids are obligatory parasites at some point while they are larvae, adults are free-living and remain relatively hidden in the substrate (Lester and Roubal, 1995). For more information on isopod biology see Schultz (1969), Kabata (1970, 1984), Schram (1986), Lester and Roubal (1995), and Bunkley-Williams and Williams (1998).

Life-cycle summary: Isopods exhibit direct life cycles. Gnathiid adult females incubate eggs that hatch into praniza larvae. Larvae molt through a series of praniza stages, at least some of which are parasitic on fishes. The final praniza stage molts into the adult that takes up free-living residence in the benthos where several adult females may be guarded by an adult male. Cymothoids are protandrous hermaphrodites. An adult female mates with a functional male on the host and then incubates her embryos in a brood pouch. Eggs hatch in the pouch and they develop into juveniles before leaving to find a host. Once attached to the host, juveniles molt into adults, and at first they function as adult males. In the presence of an adult female, adult males will develop no further. When an adult female is not present, adult males will molt into an adult female. Aegiids, cirolanids, excorallanids, and corallanids develop into adults of separate sexes through a series of juvenile stages after hatching from the egg.

Host and attachment site specificity: The host specificity of isopods that associate with fishes varies considerably. Cymothoid species generally tend to be quite host specific while other taxa tend to hold species that are more catholic regarding host preference. Species that associate with a wide range of hosts often infect fishes that share a particular habitat. Isopods can be either ectoparasites or endoparasites, and they have been recorded from the general body surface, fins, gills and branchial chambers, olfactory sacs, buccal cavity, musculature, and internal organs of fishes. Cymothoid species tend to be relatively site specific, while other taxa often have more variable distributions on their hosts. See Schultz (1969), Moreira and Sadowsky (1978), Kensley and Schotte (1989), Brusca (1980), and many contemporary references provided in Bunkley-Williams and Williams (1998) for more information on host associations and attachment locations of isopods that associate with fishes.

Mobility: Isopod associates of fishes range from species whose adults are stationary on the host, i.e., cymothoids, to others that commonly detach from the host, sometimes swimming to another host or even living for periods as predators or scavengers, e.g., aegiids, corallanids, and cirolanids. Juvenile isopods are typically excellent swimmers. Some species detach from hosts when they are disturbed, e.g., when host is netted.

Problems caused by isopods: Isopods can cause morbidity and mortality in captive fish populations. As direct agents of disease their feeding and attachment activities can cause organ and tissue damage (including blood loss) and osmoregulatory problems, and they can vector pathogens, i.e., viruses, bacteria, blood-born protozoa, and nematodes; e.g., see Davies (1982). As indirect promoters of disease they cause lesions and stress that can predispose hosts to opportunistic pathogens, e.g., bacteria and fungi. For more information on problems caused by isopods see Kabata (1970, 1984), Honma et al. (1991), and Lester and Roubal (1995).

Prevention and treatment of infections: Quarantine is the first defense (with close observation of new fishes), mechanical removal using forceps is sometimes possible, osmotic shock (freshwater or saltwater dips) is sometimes effective, water treatments using organophosphates are sometimes warranted, diflubenzuron may also be effective. When considering treatments for isopod infections it is important to remember that some isopods can detach from and live away from their hosts for significant periods. When confronting gnathiid infections it should be remembered that only the larvae are parasitic and that the adults may be found in the substrate. Thus, treating fishes infected with isopods without treating their environment may only provide temporary relief. It is also important to recognize the potential for secondary infections associated with severe isopod infections. For more particulars on treatment see Stoskopf (1993; but see also remarks in Bunkley-Williams and Williams, 1998) and Noga (1996).

Proper fixation for taxonomic study: Fix directly in 10% neutral buffered formalin or 70% ethanol. For molecular studies, specimens should be fixed and stored in 95% ethanol. Proper labeling of specimens is essential.

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