https://doi.org/10.21608/sjsci.2024.275695.1187

Manipulative Strategies of Aquatic Arthropods for Fish Parasitism: A Review

Fatma Ahmed[1,*], Ahmed Mahrous[2], Ahmed Yasser[2], Dina Abdullah[2], Fam Yousef[2], Hossam El-Deen Mohamed[2], Ismail Hassan[2], Manar M. Ali[2], Mohamed Ashraf[2], Asmaa N. Mustafa[1], Safa M. El-masry[1], Nasra M. H. Zohry[1], Ebtesam A. Yousef[1], Tarek G. Ismail[1], Azza M. Khedre[1], and Somaia A. Ramadan[1]

[1] *Faculty Member, Department of Zoology, Faculty of Science, Sohag University, 82524 Sohag, Egypt*
[2] *B.Sc. Student, Zoology and Chemistry Program, Department of Zoology, Faculty of Science, Sohag University, 82524 Sohag, Egypt*
[*] *Email: Fatmaelzahraa_ahmed@science.sohag.edu.eg*

Received: 11th March 2024, **Revised:** 28th April 2024, **Accepted:** 22nd May 2024
Published online: 31st May 2024

Abstract: Several aquatic arthropodan classes, including Arachnida, Crustacea, and Ostracoda can parasitize on fish and might cause discomfort or serious harm to their tissues. These organisms can parasitize fish through either obligatory, facultative, or opportunistic relationships according to their behavior or the surrounding conditions. Free-living arthropods may be induced to parasitize fish by various physiological, environmental, or biological stimuli. Some other non-fish parasites can switch their behavior towards fish parasitism. On one hand, members of both Classes Arachnida and Ostracoda are free-living and may exhibit opportunistic parasitism of fish through nocturnal predation or attacking weakened or captivated fish. On the other hand, Class: Crustacea is wide and comprises true fish parasites, non-fish parasites, and non-parasitic forms included in three Orders: Isopoda, Branchiura, and Copepoda. Isopods are efficient attackers targeting fish body surfaces, buccal cavities, and muscles through hydrodynamic impairment, sneaking as couples, and flesh-burrowing strategies. Branchiurans are true ectoparasites having well-developed senses for host localization and then follow a physiological strategy of injecting fish with toxic digestive fluids. When it comes to fish parasitic copepods, they are either females with robust structural features for infesting fish bodies or tiny-sized individuals capable of fish endo-parasitism by sneaking inside. Notably, the free-living and the non-fish parasitic crustaceans might be forced to parasitize fish facultatively, transitory, or occasionally. Parasitic and predatory arthropods possess structural facilities besides employing behavioral tactics for handling fish bodies successfully. The current manuscript surveyed the different strategies and unusual environmental conditions that facilitate fish parasitism by the aquatic Arthropods.
Keywords: Aquatic Mites, Crustacea, Fish, Ostracoda, Parasites.

1. Introduction

Aquatic arthropods are small-sized invertebrates sharing fish in their habitat under the water in complementary non-harmful relationships. However, these organisms might initiate fish parasitosis relations either obligatory or opportunistically [1 - 3]. The aquatic Arthropodan Classes: Crustacea, Ostracoda, and Arachnida are known to attack fish for parasitism or predation [4, 5]. The Class: Crustacea comprises obligatory parasitic species for fish that their existence leads to fish infestation and may cause severe harmful effects to the host [6, 7]. Meanwhile, other crustacean species are free-living or parasitic to other aquatic species, including Crustacea, and might infect fish occasionally [8]. The other two classes, Ostracoda, and Arachnida are mainly non-parasitic, free-living organisms but occasionally cause opportunistic parasitosis for fish in the form of predation during abnormal conditions [2, 9]. Therefore, obligatory parasitism of fish by arthropods is elicited by some distinct members of the Class: Crustacea, and the rest are opportunistic parasites.

Aquatic crustaceans are usually free-living metazoan invertebrates that live together and with other animals in balanced social relations. These relationships are bilateral and are either beneficial for both partners in what is known as “Mutualism”, or beneficial to the commensal but harmless for the host in what is known as “Commensalism” or “Cleaning symbiosis” [10]. Some distinct crustacean orders including Amphipoda, Ascothoracida, Branchiura, Cirripedia, Copepoda, Isopoda, Pentastomida, and Tantulocarida are parasitic and cause serious harm to the hosting crustacean or fish partner [10]. Branchiura, Copepoda, and Isopoda are the three main aquatic crustacean orders known to parasitize fish obligatory [3]. In some cases, crustacean parasitism is achieved opportunistically by the free-living and non-fish parasitic forms during unusual conditions [11, 12]. Nocturnal attacks on fish by crustacean zooplankton are frequent and may reveal serious harm [13]. Anyway, the existence of small numbers of crustacean parasites is of minor problem while their presence in large numbers is of major problem leading to severe distress and localized damage to different tissues of the infested fish [14 - 16]. Several pathological changes were reported by the crustacean parasitism of fish that lower the host physiology, metabolic activity, and fitness for swimming, which lowers its economic value [17, 18].

Furthermore, crustacean infestation is usually associated with secondary infections by several infectious diseases since it lowers the resistance of the hosting fish, which might be fatal for the infested fish [16]. Noteworthy, crustacean infestation induces several changes in the hosting fish behavior that appear as short-term individual responses to the attack, which act as indirect extrinsic stimulation attracting more parasites towards the infested fish [19, 20].

Obligate parasitism of fish by the three previously mentioned crustacean orders is achieved by several manipulative strategies that differ between them following their size, gender, behavior, or structural facilities. Isopods are optimal crustacean attackers for fish body surface, buccal cavity, and muscles. Hydrodynamic impairment, sneaking as couples, and flesh burrowing are the common strategies for such attacks [18-22]. Branchiurans are true ecto-parasites having well-developed senses employed for host-searching. For complete infestation and feed obtaining, they employ a physiological strategy of toxifying the hosting fish via their injection with digestive fluids [16, 20]. As for the fish parasitic copepods, they are either females possessing strong structural facilities for fish body infestation, or tiny-sized individuals capable of fish endo-parasitism via sneaking inside strategy [23, 24].

Opportunistic parasitism is a kind of facultative or occasional relationship with a host. The most important target for predatory organisms is the host body infestation for feeding [15, 25]. Therefore, fish predation by arthropods starts with direct mechanical infestation of their bodies then the large-sized predatory organisms leave the fish after blood or mucus sucking and the small-sized individuals may go for deeper mechanical infestation for endo-parasitosis. Nocturnal attacks and attacking the captivated fish are the strategies followed by both Arachnids and Ostracods for fish parasitosis [2, 9]. The non-parasitic forms of the crustacean Orders: Isopoda and Branchiura can parasitize fish through two different strategies, transient parasitism, and facultative stimulated parasitism [8, 11, 26]. The non-fish parasitic crustacean individuals, such as crustacea-crustacea parasites, might infest a fish host occasionally through the bodies of infected crustacean parasites [12, 26]. Once an infected crustacean parasitizes a fish, its crustacean parasite might infest the host fish in a secondary infestation strategy. A barnacle attached to several species of fish parasitic crustacea was observed attached to an isopod that was parasitic on an orange filefish (*Aluterus schoepfii*) [26].

In the current article, the common adaptive behavioral tactics, and structural manipulative strategies of Arthropoda for the obligatory as well as opportunistic parasitism of fish were surveyed.

2. Fish Body Manipulation Strategies by Crustacea

For the host invasion, crustacean parasites can manipulate fish bodies by primary strategies of behavioral and tactical planning for the direct mechanical infestation of fish to establish permanent or transient attachment sites on their bodies. Following, they might exhibit secondary infestation strategies that elevate the impairment of the hosting fish activity and biology, which magnifies the harm to the fish's health status mechanically as well as physiologically. These infestation strategies are facilitated by several structural facilities that differ between groups. Owing to their generous size, crustacea are mainly ectoparasitic organisms that parasitize the external body of the fish, which inhibits their growth and causes malnutrition and mortality in young fish [3, 15]. Buccal cavity infestation by the parasitic Crustacea impairs mouth breeding in *Oreochromis* sp., which is particularly lethal to early juveniles [27, 28]. The tiny-sized or early stages of Crustacea can anchor with their cephalothorax or pereopods into the fish flesh and infest their internal organs, which is commonly fatal [29]. Crustacean endo-parasitism delayed the gonad development and sexual maturity besides the negative effects on the food conversion efficiency of the infested fish [23]. Moreover, crustacean infestation of fish influences the rate of further infestations since it increases the fish's susceptibility to infection, which shows behavioral host-parasite interplay [30]. Awareness of the behavioral adaptation of crustacean parasites and the host-pathogen interaction is critical for understanding their pathogenicity, which helps develop effective prevention strategies [31]. In addition, awareness about the immune evasion strategies reported by the fish' parasitic crustacea including, the avoidance of fish immune system surveillance, immune suppression and/or modulation, escaping, indirect life cycles, and molecular simulation should be considered and interpreted with the behavioral strategies for deeper understanding of the crustacean parasitosis of fish [31, 32]. Currently, we surveyed the behavioral manipulative strategies by the different parasitic crustacean orders for fish.

2.1. Order: Isopoda

Isopoda is the largest-sized crustacean parasitic group for fish, reported 20 - 50 mm in length [16]. Structurally, Order: Isopoda is characterized by three regions of their bodies head, peraeon, and pleaon. The unsegmented head holds two pairs of antennae, one pair of eyes variable in size, and a mouth. The peraeon is segmented and consists of seven segments each possessing a pair of walking legs "peraeopods". The pleaon is also segmented and consists of six segments each possessing a pair of swimming legs ”pleopods”, except for the last segment, which forms a swimming tail “pleotelson” by the fusion with biramous “uropods” [6].

Parasitic Isopods for Fish

Fish isopod parasites are known as obligate parasites that inhabit mainly marine fish species, consuming their blood or hemolymph as well as potentially their muscles and mucus [22] [16]. A few are opportunistic parasitic isopods on farmed and freshwater fish species [6, 15]. Isopod parasitism on fish may cause mortality upon severe tissue harm [33].

Out of the 95 families of the Order: Isopoda, seven families, all from the suborder Cymothoida, are known to be parasitic on or predating fish or other crustaceans [22, 34]. Of them, six families under the superfamily Cymothooidea are partly or wholly parasitic on fish either temporarily or permanently. The fish parasitic Cymothoids are classified into two major families, Cymothoidae and Gnathiidae; and four other minor families Tridentellidae, Barybrotidae, Aegidae, and Corallanidae. The family Cymothoidae includes blood-feeding isopods having several species with immature or adult forms that are parasitic on fish [16]. They are found to settle on the body surface and caudal peduncle, mainly towards the buccal cavity of hosting fish, or live inside the gill chamber, which obstructs the opercular respiratory movements [16, 21, 35].

Cymothoids have a unique Holoxenic life cycle, involving only one permanent fish host. Early in life, the isopod attaches itself permanently to a fixed site on the hosting fish and goes through a male stage before changing into a female [21, 36]. The female's ventrum serves as a special brood pouch, inside which the eggs and larvae develop, on the hosting fish [16, 21]. Only the larval stages of the family Gnathiidae are parasites on marine fish species, while the adults are free-living, non-feeding aquatic isopods; therefore, Gnathiidians are larval isopod parasites of marine fish [8, 16]. Structurally, they are unique isopods that have five functional pairs of legs instead of seven in other isopods [8, 34]. The other four families, Tridentellidae, Barybrotidae, Aegidae, and Corallanidae, are mainly free-living and are transitory parasites or micro-predators that usually leave their hosting fish after drawing blood [8, 26]

Sites of Isopod Infestation

Site selection for the attachment of fish-parasitic isopods is species-specific and sometimes genus-specific. Different genera were reported to select a specific attachment site on their fish host bodies such as the anterior trunk, lateral line, gills chamber, or buccal cavity [22]. The site specificity for isopod parasitism is limited by the parasite's needs as well as the host morphology and habits; therefore, it can leave either a negative or no impact depending on the hosting fish [4, 22].

Mainly, the generous-sized isopod Cymothoids are surface hackers, which attack and attach to the fish's body surface ecto-parasitizing their scales, skin, fins, and eyes. Behaviourally, some ectoparasitic isopods perform different tricks to escape inside the fish body, either the buccal dwelling or the flesh burrowing [22]. The buccal dwellers assault the fish's gill cavities to escape into the fish's buccal cavity; this includes the isopods belonging to the genus *Cymothoa* sp. [37]. Notably, these cymothoid isopods may act as vectors for the transmission of some viral infections such as “lymphocystis” to the fish's internal organs during feeding on gill tissue [38]. Flesh burrowers sneak through ecto-parasitizing the hosting fish's body surface and then burrowing their muscles reaching the internal organs in some cases [29].

Structural Facilities and Behavioral Strategies of Isopods for Fish Parasitism

I. Structural Facilities

Despite having homologous bodies, different families and/or genders of fish's parasitic isopod Cymothoids have demonstrated diverse structural and behavioral adaptations for fish's body manipulation from different sites of the body. Structurally, the general shape of adults’ parasitic isopods looks like free-living forms but possesses hook-like legs. Their mouths are shaped like cones, having tiny, pointed mandibles that pierce the host tissue to get inside the blood vessels or sinuses, and maxillipeds that rip at its flesh [8]. However, some characteristic structural features are different between male and female paratypes. An example of the fish parasitic cymothoid isopod, *Ceratothoa africanae* sp., is displayed in Figure 1 [15]. The stomach, especially the hindgut, fills up quickly, causing the body to swell and exert pressure on the host tissues, which may lead to immediate death if present in the gills chamber [21]. The feed moves gradually to the midgut glands for digestion; as a result, the parasites are non-feeding and typically fed only sometimes [4]. Interestingly, Cymothoids are known by a protandrous hermaphrodite, as the firstly reaching male parasitizes a fish changes his sex into a female, and secrets pheromone or neurohormone to prevent any following male from further hermaphroditic action [8].

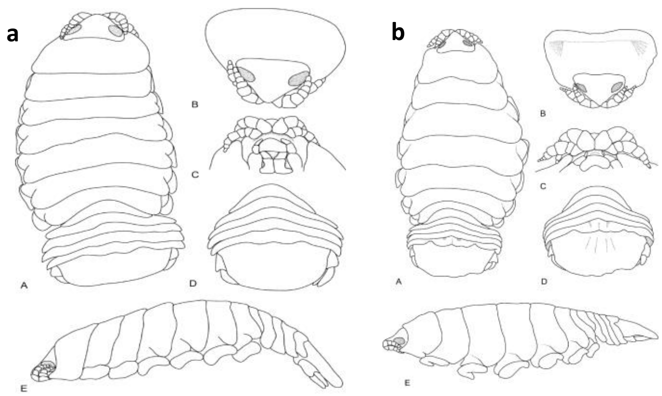

Figure 1. Structural features of male **(a)** and female **(b)** paratypes (SAM A45938; 30 mm) of the fish parasitic cymothoid isopod *C. africanae* sp. A dorsal view; B, antero-dorsal view of pereonite 1 and cephalon; C, ventral view of cephalon; D, dorsal view of pleotelson; E, lateral view [15].

Using their pereopods and mouth parts, flesh-burrowing Cymothoids can anchor the skin of hosting fish forming small holes as their habitat that are opened to the outside; therefore, vulnerable to secondary infections of several microbial diseases [16, 21] In addition, gill parasitism reveals the occurrence of cavities within the fish gill chambers and the displacement of gill filaments because of pressure from the lodged parasite [39-41].

II. Behavioral Strategies

Various behavioral strategies supported by structural facilities were reported to be followed by the different ectoparasitic and micro-predator Cymothoid families for fish host infestation, manipulation, and burnout.

(1) Hydrodynamic Impairment Strategy

It is a strategy followed by the surface hackers’ isopods during their ectoparasitism on fish. The Cymothoid isopod (*Anilocra nemipteri*) was reported ecto-parasitizing the eyes of coral reef-inhabiting fish, bridled monocle bream (*Scolopsis bilineatus*) [18]. Due to its firm attachment above the host's eyes, and since it can grow to be more than 15% of the host's total length this isopod impairs the swimming ability of the parasitized fish and reduces its speed either physiologically at slow swimming speed or hydrodynamically at high swimming speeds. In addition, the destabilizing effect of the asymmetrically attached parasite makes this parasitism strategy of a high energetic cost on the parasitized fish because it increases the friction drag along its body surface; therefore, raises even the fish's resting metabolism by the continuous O_2 consumption due to its demand for the hydrostatic balance [18].

(2) Sneaking as Couples

It is the common ecto-parasitism strategy followed by almost all the surface attackers as well as buccal dwellers of the parasitic Cymothoid isopods. In surface hackers, either the small males attach permanently to the hosting fish alongside their females, or they leave after fertilization and complete their free-swimming life. In the gill parasitic forms, both males and females inhabit the same fish even at different gill arches. The male buccal dwellers stay out on the gills with juveniles, while the adult female sneaks inside into the fish buccal cavity. In flesh dweller forms, the small-sized males stay in the same pouch as the females [8].

Working as couples was reported for fish surface parasitism in a study conducted by Aneesh et al. [42]. The ectoparasites were settled invariably in pairs at specific sites on the fish surface facing the floor of the buccal cavity. Three different couples were seen on the fish under investigation (belonid fish *Strongylura strongylura*) during the parasite infestation arranged as; male-female (18 + 18 = 36) > juvenile–juvenile (4 + 4 = 8) > male–transitional stage (3 + 3 = 6) according to their numbers.

The isopod Cymothoid *Cymothoa frontalis* exhibited another model of the coupled sneaking strategy during his parasitism on a fish buccal cavity that was reported through an early study conducted by Mladineo and Valic [43] for artificial crustacean infection. The infective stage of the isopod parasite *Ceratothoa oestroides* pulli II was aggressive free-swimming larvae and could infect sea bream after 7 days of staying on its surface. Within two hours of leaving females, the infective larvae swim quickly on the water's surface, attach themselves to a host's body surface, and then migrate quickly in pairs under the fish operculum in the buccal cavity to have their first meal after long starvation [8]. Interestingly, strong competitive mechanisms were seen when there were three or more larvae parasitizing a single fish host that forced the unpaired larvae either to stay out on the body surface without migration to the buccal cavity or detach from the buccal cavity. In another interesting study, different behavioral actions were reported by Smit and coworkers [22] followed by the different genders of isopod buccal dwellers. as there is an organized synergistic behavioral contribution between males and females concerning their role in manipulating their hosting fish. Males remain outside attaching to the gill arches just behind females, supporting them while dwelling inside the fish's buccal cavity, so they are named "gill attaching parasites". Female dwellers live inside the fish's mouth, suck out the blood of its tongue until atrophy, and then replace it, so that is named as "tongue biters or tongue replacing parasites".

(3) Flesh Burrowing Strategy

It is followed by some tinny-sized isopod cymothoids, which show a kind of meso-parasitism relationship with the hosting fish. Using their pereopods and mouthparts, they can burrow inside the hosting fish and feed on its blood and muscles [21, 39, 41]. Some odd examples of parasitic isopods were reported endo-parasitizing the fish's internal organs possibly transmitting secondary infections as well. *Lymphocystisvirus* was reported to be occasionally transmitted to the visceral organs by a parasitic cymothoid isopod [38]. In the freshwater environment, a new isopod genus, and species from the family Ergasilidae, named *Urogasilus brasiliensis* n. g., n. sp. was isolated from the urinary bladder of some fish [29]. Figure 2 displays the different ecto- as well as meso-parasitic sites of fish's parasitic isopod Cymothoids [22].

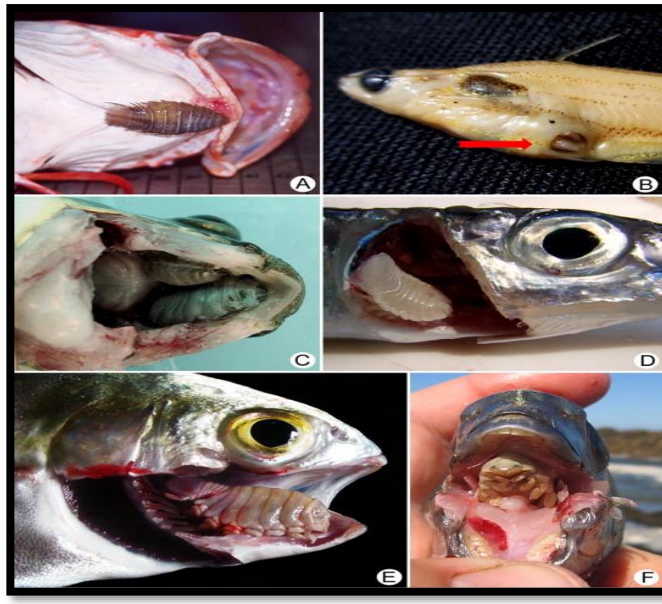

Figure 2. Several tactics of both genders from Cymothoids Isopoda for ecto-parasitism on various external attachment sites of fish. (**A**) An ecto-parasitoid that lives on the ventral surface; (**B**) a meso-parasitoid that burrows into muscles; (**C, E, F**) female parasitoids that live inside fish buccal cavities "tongue biter or tongue replacing parasites"; and (**D**) a male parasitoid that lives on fish's gill filaments supporting its female "gill attaching parasite" [22].

(4) Transient Opportunistic Parasitism

Transient parasitism is the common strategy followed by some rare free-living micro-predator isopod families. Predation for transitory feeding is their behavior for opportunistic parasitism on fish. They commonly feed on the blood of the hosting fish and leave it as free swimmers. Therefore, this opportunistic parasitism is forced physiologically by hunger. Four isopodan families, Tridentellidae, Barybrotidae, Aegidae, and Corallanidae, are mainly free-living and are transitory parasites or micro-predators that usually leave their hosting fish after drawing blood [8, 26].

The micro-predatory family Tridentellidae, and Corallanidae are two monogeneric families having mouth parts that seem to be well suited to piercing and rasping the tissues of their fish hosts [36, 44]. Both families comprise a few species that behave as facultative fish parasites [12]. Barybrotidae is a family including a monotypic genus with only one species, *Barybrotes indus*, that has been found parasitizing the gills of the devil ray *Mobula mobular* [45]. Family: Aegidae has less modified pereopods and is found parasitizing fish of brackish and freshwater in warm countries with free-swimming capability as adults [8].

2.2. Order: Branchiura

Branchiura members are entirely parasitic organisms for freshwater, brackish water, and marine water fish from their second larval stage onwards [16, 46]. The members of this order are mostly obligate, and a few are opportunistic ectoparasites for fish; therefore, they usually go for the direct manifestation of their host body invasion. The common family of fish parasitic Branchiura is Argulidae, which includes the "fish louse" or "fish lice" members [47]. The most common genus in that family is *Argulus*, which includes more than 100 common obligate ectoparasitic species for various freshwater as well as marine water fish species worldwide and causes serious outbreaks during elevated water temperatures [46 - 49]. *Argulus* spp. grow in a direct life cycle through a complex series of molts that require one host (a fish) to complete their development from eggs to mature adults [47]. Usually, gravid females swim and locate hard inanimate objects (like stones, walls, equipment, etc.) to lay their eggs on after mature adults copulate on or off the host (Figure 3) [12]. Noteworthy, temperature-related variations in hatching times are observed to cause delays of weeks or months.

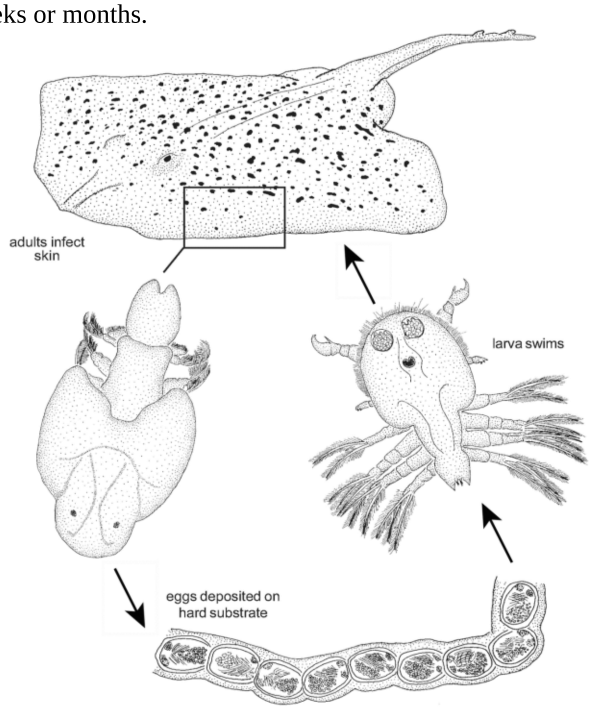

Figure 3. Representative scheme of the life cycle of *A. megalops* as drawn by Williams and Bunkley-Williams [12]. After hatching, infectious larvae will look for a suitable fish host for attachment, molting, and maturation; then detach to switch host.

Several species under the genus *Argulus* were isolated from various fish species worldwide and taxonomically identified [50, 51]. These ectoparasitic Branchiurans were reported to cause epidemiology in several kinds of water bodies worldwide [47, 52]. Grant and his coworkers [24] displayed several skin irritations and inflammations at the attachment sites of fish lice heavy infestation as shown in Figure 4. *Argulus* spp. bodies are oval to rounded, flattened dorso-ventrally, and wholly covered dorsally by a horseshoe- or rounded-shaped carapace. The head is segmented into five segments that possess two compound eyes and a thin, needle-like device called a "stylet" placed in front of the mouth tube that helps the parasite pierces the host tissues and absorb bodily fluids, and two suckers on both sides of the mouth help attachment to the host [47]. Head appendages positioned ventrally are developed for attachment [16]. The trunk is separated into a thoracic area that houses four pairs of powerful swimming legs, which are equipped with spines and hooks that facilitate movement [47]. A short abdomen that contains a single bilobed unit (the caudal fin).

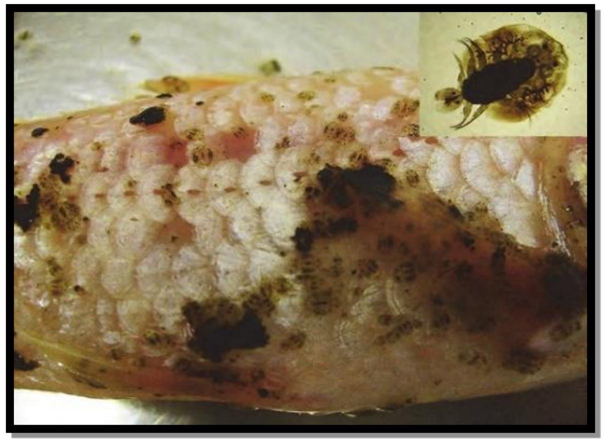

Figure 4. Heavily infested fish with a generously sized fish louse *Argulus* sp. having characteristic paired eyes, suckers, legs, and egg sacks; low magnification under the light microscope. Irritations and inflammations are appeared at the sites of the parasites' attachment [24].

The second common genus is *Dolops*, which has only one species reported in Africa, and it is not the same as *Argulus* in that it has a hook instead of a sucker on its second maxilla. Chonopeltis are typically the smallest; the mouth tube present in other Argulids is absent, the head appendages are weak and primitive, and the second maxilla's cup-shaped sucker is developed. The width and size of the carapace decreased. If isolated from its host, *Chonopelitis* cannot swim, in contrast to the Argulids, which can swim actively [16]. Both *Argulus* sp. and *Dolops ranarum* remain parasitic throughout their lives, but they also switch hosts when they shed their skin, lay eggs, and mate [6]. Males and females can live independently for up to 15 days [16].

Structural Facilities and Behavioral Strategies of Branchiurans for Fish Parasitism

Several studies were conducted to realize the tendency of Branchiura for fish parasitism. However, still little is known about the host-parasite interaction strategies and the hazardous effects of their parasitism on fish health and body integrity [53].

(1) Host Searching Strategy

It is a commonly known strategy of obligatory parasitism on fish by Branchiura that mainly relies on the parasite's sensation, the acute vision during light or dark conditions when conducting parasitism, or free-swimming activities [20]. This strategy is usually forced by environmental as well as physiological implementation according to the internal demands of the parasites' hunger. The Branchiuran parasites then go for the direct mechanical manifestation of fish bodies using several structural facilities of their bodies.

The fish are the primary target of *Argulus* spp. that show considerable behavioral adaptations from searching for mates and hosts until the complete host manipulation [20]. These ectoparasites frequently switch hosts causing skin damage and serve as vectors for fish pathogens. Mikheev and coworkers [30] demonstrated in a pioneering study that the obligatory ectoparasite *A. foliaceus* can search for a host fish in both light conditions initiated by external illumination and dark environments when internally stimulated by hunger. This forces the parasite to use all its senses - olfaction, vision, and/or mechano-reception - to locate its host.

In a further report, Mikheev and coworkers [20] reviewed the different behavioral adaptations of two fish parasitic species of *Argulus* (*A. foliaceus* and *A. coregoni*) for searching their fish hosts during day and night (Table 1). These parasites switch between two different tactics for the host-searching strategy during the day and night, "hover-and-wait" or "Cruising". The "hover-and-wait" tactic is used during the day to intercept and attack the swimming fish by swimming slowly while bending their bodies to one side. Whereas the "Cruising" tactic involving swimming at a significantly faster speed while keeping the body horizontal is more usable at night to attach the slowly moving or stationary fish [30].

Table 1. The common day-and-night behavioral traits observed by Mikheev and his coworkers [20] on two fish parasitic species of *Argulus* sp., *A. foliaceus* and *A. coregoni*.

Behavior/Stimulation	**Period**	
	At Day	**At Night**
Sensory Modalities	Vision, Olfaction, Mechanoreception	Olfaction, Mechanoreception
Motor Activity	Low	High
Internal Modifiers of Activity	Hunger state	
Fish-induced Stimuli	Visual, Chemical, Mechanical	Chemical, Mechanical
Host Searching Tactics	Hover-and-Wait	Cruising speed
More Efficient at host searching	*A. coregoni*	*A. foliaceus*

Host searching behavior of the parasite is motivated by several intrinsic as well as extrinsic inductions fortified by mechanoreception, vision, and olfaction senses during the day, and by the mechanoreception and olfaction during the night. Parasite starvation for up to 2 days was an intrinsic stimulus that accelerated their swimming speed for host searching, while fish behavior was an extrinsic stimulus that directly affected their host choice [30]. Noteworthy, the swimming speed of *A. foliaceus* is greater at night than during the daylight; therefore, the more reflective fish at daytime, while the slower and intermittently moved fish at night are more susceptible to the attachment of *A. foliaceus* [54].

As for *A. coregoni*, the host recognition by vision and olfaction changes with the parasites' maturation [55]. The newly hatched larvae and early juveniles are attracted to every bright object; therefore, they selected the bright fish species with the highest body reflectivity for their parasitism. In contrast, they did not react with bright objects in the absence of chemical stimulation by salmonid fish odor [55]. Young *A. coregoni* parasites are primarily attracted to fish with higher body reflectivity and exhibit little host specificity. However, an intrinsic ontogenetic shift in host preference was observed, with the parasites favoring the fish species on which they had previously been cultured in a lab [56].

After the hosting fish body infestation, *Argulus* sp. parasite feed by puncturing the skin of the hosting fish using its mouth tube, which is supplied with a spine by which the parasite can conceivably push the epidermal tissue into the deeper layers of the dermis [48, 53]. In addition, the parasite destroys the uncovered mucoid layers and epidermal layers and by using its maxillules' terminal hooks it can link injuries in deeper dermal layers causing severe skin damage, stress, and even death [48] [53]. Similar lesions were reported by the infestation with the Brachiuran ectoparasite *Dolops ranarum*, which pierces the catfish skin to feed on its blood. This parasite uses hooks on powerful contractile maxillae to attach temporarily to the host's integument, and it can detach and change its attachment site on its host [53].

(2) Toxin Injection

It is a physiological strategy supporting the mechanical infestation of Branchiuran parasites into fish tissue. Therefore, the heavy Branchiuran infestations can cause serious damage and fish mortality. *Argulus foliaceus L*., a freshwater fish louse, was observed to puncture its host's skin, and suck blood-fed, injecting a toxin [46]. *Argulus* sp. feeds by secreting and injecting comparatively large amounts of digestive fluids into fish muscles, which are poisonous to fatal. A single fish lice sting can be fatal to a small fish [16].

(3) Opportunistic Induced Parasitism

The tendency of Branchiura for fish parasitism differs between the different species and still little is known about the host-parasite interaction strategies and the hazardous effects of their parasitism on fish health and body integrity [53]. However, it was reported that stimulation toward parasitism is a unique strategy by which some free-swimming Branchiura switch to the parasitic life. They might be forced by several environmental factors towards fish parasitism in an opportunistic or facultative relationship. For instance, some males Branchiura are free-swimming and don't tend to parasitize fish, except during objectionable or abnormal environmental circumstances that stimulate their parasitic behavior toward fish. In an interesting and attractive study, Bandilla and co-workers [11] tried to track the tendency of the free-swimming males of *A. coregoni* for fish parasitism like their females by using several physical, chemical, and biological stimuli. They reported that males A. coregoni were more strongly attracted to both the light and the fish odor stimulations than their females. Notably during the study, the light stimulus was more attractive for both genders than the fish-smell. Anyway, both genders parasitized the fish successfully. Notably, some responses, through the detection of and the attraction to the applied biological stimulus of females' fish odor, were observed by Bandilla et al. from the free-swimming males *A. coregoni*; the action that was not observed in the adult females. However, this response didn't affect their choice of the hosting fish, as they didn't infest fish infected with females more than infesting the parasite-free fish. In addition, the free-swimming *A. coregoni* were found to parasitize fish in response to direct signals, most predominantly visual signals, from the hosting fish itself [11].

2.3. Order: Copepoda

Structural Facilities and Behavioral Strategies of Copepoda for Fish Parasitism

I. Structural Adaptations

Members of the Order: Copepoda are mainly free-living organisms and their parasitism on fish is based on their size and/or gender. However, copepods comprise the most common parasitic crustacean organisms for fish, which have small to microscopic body sizes [16]. Members of this order can manipulate their hosting fish by direct infestation of the external body surface, and they can sneak inside the internal organs. For the direct infestation of a fish's host body, female copepods are provided with specific structural parasitism facilities to anchor the body of the hosting fish. In an early descriptive study, the pre-metamorphosed females of *Pollachius pollachius* (L.) were described by the Zoologist Sproston [57]. Parasitism of the copepodid and chalimus stages of females was reported to cause localized damage and ischemia to the gills of fish hosts promoting only minor tissue responses in the form of mild inflammations [57].

The most common fish parasitic family of copepods is Caligidae, the sea lice of marine and brackish water fish species. *Caligus* spp. can parasitize their hosting fish via three different structural facilities during the three stages of their life. Adults cling to the hosting fish surface using prehensile antennae while all the other stages attach their hosts via invasive frontal filaments. The preadult stages detach from the hosts and can move over and feed on the fish epidermis using special mouthparts. These females possess secondary antennae by which they can attach to hosting fish tissues by forming constrictive links around the blood vessel at the anterior angle of the gill arch [58].

Unlike males, the small-sized adult female copepod, *Lernaea* sp., holds root-like mouthparts by which it attaches firmly and permanently to the skin of a fish host, hence named commonly "Anchor worm" [24, 59] The pressure of these copepod parasites on the hosts' organs associated with the feeding behavior of the infective as well as the other attached stages can cause varying degrees of atrophy and localized damage. They can anchor deeply in the fish musculature, which may cause mass mortalities of the young fish (Figure 5) [24].

II. Behavioral Strategies

Sneaking inside strategy

By this strategy, copepods perform fish ecto-parasitism and might go for their endo-parasitism owing to their small body sizes. Adherence and penetration of the copepod parasitic pre-metamorphosed females might take place from the base of the gill filaments to reach the internal organs of the hosting fish. An early study had reported the ectoparasitism of the early stages of the copepod *Lernaeocera branchialis* to the Atlantic cod gills followed by their sneaking for the endo-parasitism reaching the fish gonads [23]. This manipulative plan revealed successive local hemorrhage along the parasite way from the hyoid arch of gills up to the gonads.

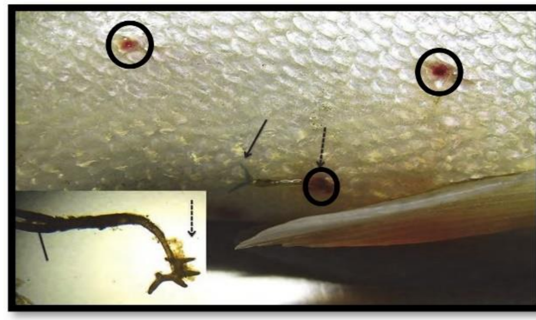

Figure 5: Anchor worm (*Lernaea* sp.) infestation to a fish's muscles (arrows) using a holdfast organ (dashed arrow); low magnification under a light microscope. Irritations and inflammations appear at the sites of the parasites' attachment (circled areas) [24].

3. Fish Opportunistic Parasitism Strategies by Predatory Arthropods

3.1. Class: Arachnida (Acarina) (Water mites) (Eight-legged arthropods)

Water mites are not true parasites for fish and few records listed water mites' infestations in fish as accidental or unusual parasitic organisms [60]. Unusually, they attack the mucous membranes, colonize, and proliferate on weakened or stressed fishes and may kill the infested juveniles [9, 60] Most likely, water mites' larvae are parasitic forms that are usually encapsulated with a layer of collagenous connective tissue. Fish's skin, gills, esophagus, intestine, and air bladder can become heavily infested and damaged by several genera of aquatic mites that might not have been classified [61]. In early surveys, some species of distinct genera of mites were reported in parasitism relationships with different fish species. The gills of stocked freshwater eel *Anguilla anguilla* were parasitized by all the life stages (larvae, protonymphs tritonymphs, and adults) of the mite species *Histiostoma* (*Ichtanoetus*) *anguillarum* subg. and sp. n. (Acari, Anoetidae) except for the deutonymphs stage [62]. Another species of the same genus, *H. piscium* sp. n., infested the farmed *Pangasius sutchi* and was embedded into a thick gelatinous sheath that filled its swim bladder [63]. Similarly, *H. papillate* was reported to attack farmed Murray cod and kill its juveniles [60].

Likewise, the genus *Schwiebea* (*Jacotietta*) *estradai* was reported to parasitize the skin, gills, and intestine of the wild as well as farmed trout *Salmo trutta fario* leaving high mortality [64]. Unexpectedly, the aquatic oribatid mite of the genus *Trhypochthoniellus* (Acari: Trhypochthoniidae), which is usually found on aquatic plants and organic matter was reported in unusual parasitism relationships with two fish species. It was observed to infest the gills of wild Atlantic salmon *Salmo salar* juveniles and the skin, fins, gills, and buccal cavity of farmed Nile tilapia *Oreochromis niloticus* [9]. Intensive growths of mites from this species were seen scattered dorsally on the caudal peduncle and anal fin as well as ventrally on the buccal cavity and pectoral girdle (Figure 6) [9].

All the developmental stages from eggs until the gravid females were observed on the fish. This indicates the suitable environment for their life and reproduction on the fish mucus membranes. The isolated adult females of the parasitic mites were identified from their dorsal and ventral morphology via Scanning Electron Microscopy (SEM) as oribatid mite *T. longisetus* (Figure 7) [9].

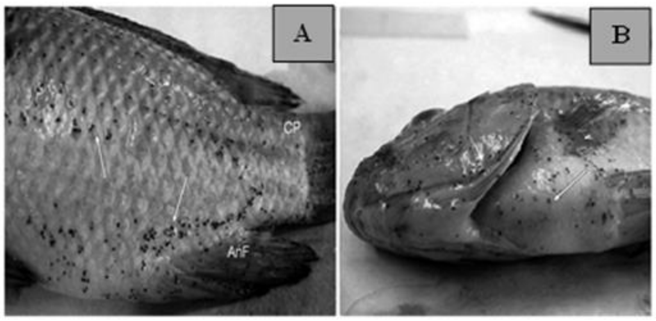

Figure 6. Infestation of the water mite *Trhypochthoniellus longisetus longisetus* to the skin of Nile tilapia *O. niloticus*. (**A**) Lateral view and (**B**) ventral view of the fish. Intensive growths of mites were seen on the dorsal side of the caudal peduncle (CP) and anal fin (AnF), and on the ventral side of the pectoral girdle (arrows) [9].

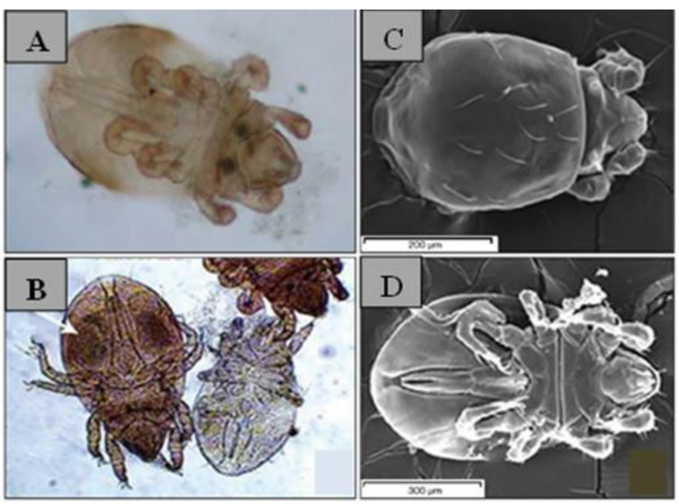

Figure 7. Photomicrographs of light and scanning electron microscopes show the morphology of the female water mite *Trhypochthoniellus longisetus longisetus* isolated from Nile tilapia skin. (**A** and **B**) Light micrographs: (**A**) ventral view of adult female; and (**B**) adult female possessing developing eggs (arrow) and associated with a nymph. (**C** and **D**) Scanning electron micrographs: (**C**) dorsal view (200 µm); and (**D**) ventral view of female (300 µm) [9]

.

It is worth mentioning that several species of mites and insects commonly infest salted, dried fish and their marketing byproducts [65, 66]. Successful experimental infestation of salted, sun-dried fish flesh with the mite *Suidasia medanensis* was reported after one week of exposure without a statistically significant correlation to the flesh salinity retention, meanwhile, the study suggests the presence of a critical salinity level for the successful infestation [67]. On the other hand, a statistically significant correlation was observed between the flesh moisture content and the number of infesting adults of *S. medanensis* [68].

3.2. Class: Ostracoda

Ostracods are a bioluminescent microcrustacean group well known as live zooplankton food source for fish worldwide, providing their useful ingredients to their predator fish [69]. The bioluminescent fish, *Parapriacanthus ransonneti*, gets its luciferin and luciferase enzyme from the bioluminescent ostracod, *Cypridina noctiluca* live feed [70]. However, due to their poor digestibility, Ostracoda must be avoided in fish larviculture feed several days after hatching [71]. In addition, the live feed of ostracod zooplankton is known as an intermediate host for several fish parasites [72, 73].
In addition, Ostracoda is a micro-predator group of arthropods and is well known to scuba divers for their painful bites at night, their bioluminescent glow, and their nocturnal attacks on injured fish [13, 34]. Some individual case studies reported the predation and parasitism interaction between ostracods and fish; however, this relation is infrequent. *Vargula tsujii* parasitized free-living wild fish in India during different seasons in 2019, and they were observed to attack the gills, buccal cavity, and intestinal tract of the weekly sampled fish. The infesting ostracods were found more predominantly on the fish gills (Figure 8A), and the parasites were isolated and reported ~ 0.5 mm in diameter (Figure 8B) [2]. Noteworthy, there were no real risks to human health from eating the infested fish [74].

Figure 8. The ostracodan *Vargula tsujii* infests the gills of a stressed fish, *Carangoides gymnostethus*. **A**: *V. tsuiii* attached to gill filaments; and **B**: close view of the isolated parasite [2].

Surprisingly, ostracods were reported to be micro predators threatening even the aggressive wild fish. This might be due to their strategy of attacking fish at night or under confinement. The ostracod, *Sheina orri*, parasitized the gills of the epaulette shark captivated in coral reefs in Australia [75]. The nocturnal predation of luminescent cypridinid ostracod *V. tsujii* was reported on confined nearshore fishes in southern California [13].

4. Conclusion

Arthropods comprise several facultative fish parasites besides comprising true obligatory parasites for fish. The obligatory parasites are included in the Class: Crustacea, mainly the three Orders: Isopoda, Branchiura, and Copepoda. Meanwhile, the opportunistic species are included in the Classes: Arachnida, Ostracoda, and rarely free-living or non-fish parasitic Crustacea. The parasitic arthropods exhibit differential strategies by which they can manipulate fish hosts. Hydrodynamic impairment, sneaking as couples, and flesh-burrowing are the common strategies followed by Order: Isopoda. Host searching and toxins injection are the common strategies followed by Order: Branchiura. Direct infestation and sneaking inside are the common strategies followed by Order: Copepoda. Nocturnal attack and attacking the weakened or captivated fish are the main strategies of Classes: Arachnida and Ostracoda for their opportunistic parasitism on fish. Secondary infestation is a known strategy followed by non-fish parasitic crustacean individuals, crustacea-crustacea parasites, as a hidden way for the occasional secondary parasitism of fish parasitized by infected crustacea. As for the free-living forms of Crustacea, two different strategies were reported by the two Orders: Isopoda and Branchiura. Fish predation for transient parasitism for feeding is the strategy of some free-living members of the crustacean Order: Isopoda. Meanwhile, parasitism stimulation is the only strategy of the free-living members of the crustacean Order: Branchiura forced their facultative parasitism of fish.

CRediT authorship contribution statement:

"Conceptualization, F.A.; software, F.A.; validation, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M.; investigation, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M.; resources, F.A., M.M.A., A.M., A.Y., I.H., D.A., F.Y., M. A., and H.El-D.M.; data curation, F.A.; writing—original draft preparation, F.A., M.M.A., A.M., A.Y., I.H., D.A., F.Y., M. A., and H.El-D.M.; writing—review and editing, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M.; visualization, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M.; supervision, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M.; project administration, F.A., S.A.R, A.M.K., T.G.I, N.M.H.Z., E.A.Y., S.M.El-m., and A.N.M. All authors have read and agreed to the published version of the manuscript."

Data availability statement

The data used to support the findings of this study are available from the corresponding author upon request.

Declaration of competing interest

The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

References

[1] F. O. Iyaji, J. E. Eyo, *Bio-Research*, 6 (2009) 328–338.
[2] S. Jayapraba, A. Gopalakrishnan, M. Yosuva, *International Journal of Environmental Climate Change*, 10 (2020) 233–246.
[3] Ahmed, F., Hend, A., Bakheet, Y., Ahmed, Y., *Sohag Journal of Sciences*, 7 (2020) 123–129.
[4] G. Rameshkumar, S. Ravichandran, Parasitic Diseases, 38 (2014) 138–141.
[5] A. R. Faruk, Fish Parasites: Infectious Diseases Associated with Fish Parasite, Seafood Safety and Quality. 1st Ed. eBook ISBN9781351007566, 2018.
[6] J. F. N. Abowei, O. F. Briyai, S. E. Bassey, *Pharmacology and Toxicology*, 2 (2011) 213–226.
[7] J. F. N. Abowei, E. N. Ezekiel, *International Journal of Animal and Veterinary Advances*, 3 (2011) 337–351.
[8] S. Ravichandran, G. Rameshkumar, K. Kumaravel, *World Journal of Fish and Marine Sciences*, 1 (2009) 137–140.
[9] A. S. Olmeda, M. M. Blanco, J. L. Pérez-Sánchez, M. Luzón, M. Villarroel, A. Gibello, *Diseases of Aquatic Organisms*, 94 (2011) 77–81.
[10] N. J. Smit, N. L. Bruce, K. A. Hadfield, Parasitic Crustacea: State of knowledge and future trends. Springer, Germany, 2019.
[11] M. Bandilla, T. Hakalahti-Sirén, E. T. Valtonen, *International Journal of Parasitology*, 37 (2007) 1343–1349.
[12] E. H. Williams, L. Bunkley-Williams, Life cycle and life history strategies of parasitic crustacea, Parasitic crustacea. Chapter 5. Springer Nature, Switzerland, 2019.
[13] C. Stepien, R. Brusca, *Marine Ecology Progress Series*, 25 (1985) 91–105.
[14] A. Alaş, A. Öktener, D. Çakir Türker, *Transylvanian Review of Systematical and Ecological Research*, 17 (2015) 39–62.
[15] K. A. Hadfield, N. L. Bruce, N. J. Smit, *Zookeys*, 400 (2014) 1–42.
[16] K. Misganaw, A. Getu, *Fisheries and Aquaculture*, 7 (2016) 13–17.
[17] T. Scholz, *Veterinary Parasitology*, 84 (1999) 317–335.
[18] S. A. Binning, D. G. Roche, C. Layton, *Biology Letters*, 9 (2013) 20120927.
[19] V. N. Mikheev, A. F. Pasternak, Doklady *Biological Sciences*, 432 (2010) 230–232.
[20] V. N. Mikheev, A. F. Pasternak, E. T. Valtonen, *Parasites and Vectors*, 8 (2015) 1–10.
[21] S. Ravichandran, G. Rameshkumar, T. Balasubramanian, *Journal of Parasitic Diseases*, 34 (2010) 97–98.
[22] N. J. Smit, N. L. Bruce, K. A. Hadfield, *International Journal for Parasitology: Parasites and Wildlife*, 3 (2014) 188–197.
[23] J. L. Smith, R. Wootten, C. Sommerville, *Journal of Fish Diseases*, 30 (2007) 1–11.
[24] Grant, B., D. Huchzermeyer, B. Hohls, Manual for fish kill investigations in South Africa. Report for the Water Research Commission. South Africa, 2014.
[25] K. A. Hadfield, History of discovery of parasitic crustacea, Parasitic Crustacea. Springer Nature, Switzerland, 2019.
[26] N. L. Bruce, *National Institute of Water and Atmospheric Research (NIWA) Biodiversity Memoirs*, 122 (2009) 1–252.
[27] T. Hecht, F. Endemann, *Journal of Applied Ichthyology*, 14 (1998) 213–221.
[28] B. S. Dezfuli, L. Giari, R. Konecny, P. Jaeger, M. Manera, *Diseases of Aquatic Organ*isms, 53 (2003) 257–262.
[29] D. F. Rosim, G. A. Boxshall, P. S. Ceccarelli, *Parasitology International*, 62 (2013) 347–354.
[30] V. N. Mikheev, A. V. Mikheev, A. F. Pasternak, E. T. Valtonen, *Parasitology*, 120 (2000) 409–416.
[31] F. Ahmed, G. Kumar, F.M. Soliman, M.A. Adly, H.A.M. Soliman, M. El-Matbouli, M. Saleh, *Comparative Biochemistry and Physiology - Part D: Genomics and Proteomics*, 32 (2019) 100625.
[32] A. Sudhagar, G. Kumar, M. El-Matbouli, *International Journal Molecular Sciences*, 19 (2018) 245.
[33] M. Tavares-Dias, M. B. F. Dias-Júnior, A. C. Florentino, L. M. A. Silva, A. C. da Cunha, *Revista Brasileira de Parasitologia Veterinária*, 24 (2015) 136–147.
[34] A.M. Quattrini, A.W.J. Demopoulos, *International Journal for Parasitology: Parasites and Wildlife*, 5 (2019) 217-228.
[35] M. C. Oguz, A. Oktener, *Turkiye Parazitology Derg*., 31 (2007) 79–83.
[36] L. Bunkley-Williams, E. H. Williams, *Journal of Parasitology*, 84 (1998) 893–896.
[37] A. T. Yuniar, H. W. Palm, T. Walter, *Parasitology Research*, 100 (2007) 1193–1204.
[38] R. M. Overstreet, J. Jovonovich, H. Ma, Integr. *Comparative Biology*, 49 (2009) 127–141.

[39] M. Rajkumar, P. Perumal, J. P. Trilles, *Diseases of Aquatic Organisms*, 66 (2005) 87–90.

[40] K. P. Jithendran, M. Natarajan, I. S. Azad, *Marine Finfish Aquaculture Network*, July-September (2008) 47–50.

[41] J. Praveenraj, K. Saravanan, P. K. Puneeth, S. Ravichandran, A. Baruah, D.S. Monalisha, S. R. Kiruba, D. S. Roy, *Journal of Parasitic Diseases*, 41 (2017) 593–595.

[42] P. T. Aneesh, K. Sudha, A. K. Helna, G. Anilkumar, J. P. Trilles, *Zookeys*, 353 (2014) 339–353.

[43] I. Mladineo, D. Valic, *Bulletin of the European Association of Fish Pathologists*, 22 (2002) 304–310.

[44] N. L. Bruce, *Zoological Journal of the Linnean Society*, 80 (1984) 447–455.

[45] P. S. Moreira, V. Sadowsky, *Boletim do Instituto Oceanográfico*, 27 (1978) 95–152.

[46] A. Öktener, A. H. Ali, A. Gustinelli, M. L. Fioravanti, *Ittiopatologia*, 3 (2006) 161–167.

[47] N. Steckler, R. P. Yanong, *Edis*, 2013 (2013) 1–5.

[48] A.J. Harrison, N.F.S. Gault, J.T.A. Dick, *Diseases of Aquatic Organs*, 68 (2006) 167–173.

[49] M. Koyun, *Notulae Scientia Biologicae*, 3 (2011) 16–19.

[50] L. A. M. Everts, A. Avenant-Oldewage, *Malaysian Applied Biology*, 38 (2009) 61–71.

[51] M. Saha, P. K. Bandyopadhyay, *Biolife*, 3 (2015) 813–819.

[52] N.G.H. Taylor, C. Sommerville, R. Wootten, *Journal of Fish Dis*eases, 29 (2006) 193–200.

[53] A. Avenant-Oldewage, *Journal of Fish Diseases*, 17 (1994) 641–647.

[54] V. N. Mikheev, E. T. Valtonen, P. Rintamäki-Kinnunen, *Parasitology*, 116 (1998) 425–430.

[55] V. N. Mikheev, A. F. Pasternak, E. T. Valtonen, *Parasitology Research*, 92 (2004) 220–224.

[56] V. N. Mikheev, A. F. Pasternak, E. T. Valtonen, *Parasitology*, 134 (2007) 1767–1774.

[57] N. G. Sproston, *Journal of Marine Biology Association*, 25 (1942) 441–466.

[58] I. Barber, R. Poulin, *Handbook of Fish Biology and Fisheries*, 1 (2008) 259–389.

[59] C. R. Berry, G. J. Babey, T. Shrader, *Journal of Wildlife Diseases*, 27 (1991) 206–213.

[60] R. B. Halliday, R. O. Collins, *Australian Journal of Entomology*, 41 (2002) 155–158.

[61] R. Heckmann, *Aquaculture Magazine*, 29 (2003) 20.

[62] A. Fain, C. Belpaire, *Bulletin et Annales de la Societe Royale Belge d'entomologie*, 121 (1985) 285–292.

[63] A. Fain, L. Lambrechts, *Bulletin et Annales de la Societe Royale Belge d'entomologie*, 121 (1985) 119–126.

[64] A. Fain, M. Ferrando, *Rev Iber Parasitology*, 50 (1990) 67–71.

[65] F. Eke, N. Ekechukwu, I. Onah, *Animal Research International*, 5 (2008) 900–903.

[66] S. M. Singh, S., R. Bharti, Abdul Aziz, S. Pradhan, B. Chhaba, N. Kaur, *Journal of Entomology and Zoology Studies*, 6 (2018) 2720–2725.

[67] A. K. Cicilykutty, N. K. Balasubramanian, P. A. John, *Acarologia*, 22 (1981) 291–293.

[68] A. K. Cicilykutty, N. K. Balasubramanian, P. A. John, *Acarologia*, 22 (1981) 295–297.

[69] R. Victor, Crustacea: Ostracoda. Freshwater Invertebrates of the Malaysian Region. 1st Ed. Academy of Sciences, Malaysia, 2004.

[70] M. Bessho-Uehara, N. Yamamoto, S. Shigenobu, H. Mori, K. Kuwata, Y. Oba, *Science Advances*, 6 (2020) eaax4942.

[71] L. U. Gonçalves, L. A. França, C. M. Epifânio, F.A.L. da Fonseca, A.M. de Alcântara, R.G. do Nascimento, E. N. S. da Conceição, Silva, L. E. Castanheira, *Aquaculture*, 505 (2019) 344–350.

[72] O.L. Lassiere, *Parasitology*, 97 (1988) 331–338.

[73] F. Moravec, K. Nagasawa, M. Miyakawa, *Diseases of Aquatic Organisms*, 66 (2005) 171–173.

[74] S. Jayapraba, A. Gopalakrishnan, *GSC Biological and Pharmaceutical Sciences*, 13 (2020) 273–283.

[75] M.B. Bennett, M.R. Heupel, S.M. Bennett, A. R. Parker, *International Journal of Parasitology*, 27 (1997) 275–281.