Veterinary and public health aspects in tilapia (Oreochromis niloticus niloticus) aquaculture in Kenya, Uganda and Ethiopia

Daniela Florio
University of Bologna

Andrea Gustinelli
University of Bologna

Peter Akoll
Makerere University

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*Aspetti veterinari e di sanità pubblica nell'allevamento della tilapia (*Oreochromis niloticus niloticus*) in Kenya, Uganda ed Etiopia*

Daniela Florio [1], Andrea Gustinelli [1], Monica Caffara [1], Francesco Turci [1], Francesco Quaglio [2], Robert Konecny [3], Tanja Nikowitz [4], Euty M. Wathuta [5], Adiel Magana [5], Elick O. Otachi [5], Geraldine K. Matolla [6], Hellen W. Warugu [6], David Liti [6], Raphael Mbaluka [7], Benson Thiga [7], Jonathan Munguti [8], Peter Akoll [9], Wilson Mwanja [10], Kassahun Asaminew [11], Zenebe Tadesse [11], Maria Letizia Fioravanti [1*]

[1] Dept. Veterinary Public Health and Animal Pathology, Bologna University, Italy; [2] Dept. Public Health, Comparative Pathology & Veterinary Hygiene, University of Padua, Italy; [3] Environment Agency, Vienna, Austria; [4] Dept. Freshwater Ecology, University of Vienna, Austria ; [5] Egerton University, Kenya; [6] Moi University, Kenya; [7] Sagana Fish Culture Farm, Kenyan Ministry of Fisheries Development, Kenya; [8] Kenya Marine & Fisheries Research Institute, Kenya ;[9] Dept. Zoology, Makerere University, Kampala, Uganda; [10] Dept. of Aquaculture, Ministry of Agriculture, Animal Industry and Fisheries, Entebbe, Uganda; [11] Ethiopian Institute of Aquaculture Research, Addis Ababa, Ethiopia.

SUMMARY - The European INCO-DEV BOMOSA Project (Integrating BOMOSA cage fish farming systems in reservoirs, ponds and temporary water bodies in Eastern Africa) intends to establish small scale fish farming in marginal water bodies in Kenya, Uganda and Ethiopia, creating rural aquaculture networks with the aim to economically integrate aquaculture with agriculture. In order to study veterinary and public health aspects in Bomosa fish farming systems, a parasitological survey on wild and cultured tilapias (*Oreochromis niloticus niloticus*) has been carried out from March 2007 to October 2008. A total of 982 tilapias - 685 from Kenya, 222 from Uganda and 75 from Ethiopia - were sampled and subjected to complete parasitological examination. During the survey, fish from different environments (258 from open water reservoirs, 445 from ponds and 279 from BOMOSA cages) were sampled in order to evaluate the factors which could influence the parasite diffusion and outbreak of diseases. Quantitative data, such as prevalence, intensity and abundance, and qualitative data are presented together with the main abiotic and biotic risk factors identified as relevant for veterinary and public health aspects in tilapia aquaculture.

*RIASSUNTO - Nell'ambito del progetto europeo INCO-DEV "Integrating BOMOSA cage fish farming systems in reservoirs, ponds and temporary water bodied in Eastern Africa" si è condotto uno studio sui fattori sanitari di carattere strettamente veterinario e di potenziale interesse in sanità pubblica che possono interessare l'allevamento della tilapia (*Oreochromis niloticus niloticus*) in Kenya, Uganda ed Etiopia. Sono state sottoposte ad esami necroscopici e parassitologici 982 tilapie - 685 dal Kenya, 222 dall'Uganda e 75 dall'Etiopia. Dei soggetti esaminati, 258 provenivano da acque libere, 445 erano allevati in bacini in terra e 279 in gabbie galleggianti approntate secondo il sistema "BOMOSA", sperimentato nell'ambito del progetto al fine di garantire un migliore sfruttamento di risorse idriche marginali e temporanee e una gestione semplificata da parte delle comunità locali. Vengono presentati i risultati quantitativi (prevalenza, intensità, abbondanza) e qualitativi delle indagini parassitologiche, unitamente alla descrizione dei fattori di rischio abiotici e biotici individuati in relazione agli aspetti veterinari e di sanità pubblica che possono interessare le tilapie allevate nei sistemi acquatici presi in considerazione.*

Key words: *Oreochromis niloticus niloticus*; Aquaculture; BOMOSA project; Kenya; Uganda; Ethiopia.

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* Corresponding Author: c/o Dept. Veterinary Public Health and Animal Pathology, Via Tolara di sopra, 50 – 40064 Ozzano Emilia (BO), Italy. Tel.: +39-051-2097068; Fax: +39-051-2097039; E-mail: marialeti.fioravanti@unibo.it.

INTRODUCTION

Aquaculture activities can be actually encountered among the zootechnical segments more involved in the production of animal proteins in developing countries, furnishing a precious contribute to the global food security, poverty alleviation, rural livelihoods, employment and income generation (www.fao.org/fishery/publications/sofia).

Concerning the African continent, although productive and technological advances have been recently achieved in the tilapia farming in Egypt, the aquaculture activities undertook in East African countries are still under-developed for several reasons, among which the lack of know-how both in productive and commercial areas is of great importance (Coche & Pedini, 1997).

The BOMOSA project is based on the application of cage-based fish farming systems, especially prototyped for East African conditions, as a small-scale productive model integrating rural resources, technological optimization, socio-economic aspects and capacity building in order to furnish a know-how indispensable for the sustainable and permanent developing of Eastern Africa aquaculture.

Diseases could represent an important constraint to aquaculture production, causing both production losses and public health problems. Therefore, the knowledge of the diseases of major concern in a fish farming system is necessary in order to assess the risk factors influencing their introduction/spreading and define the measures useful to their prophylaxis and control.

Although several scientific papers and books have been published in the past on pathology of wild and farmed fish from Africa (Robert & Sommerville, 1982; Paperna *et al*., 1984; Van As & Basson, 1984; Michel, 1989; Paperna, 1996), the collection of new data was necessary to identify the diseases of major concern in the BOMOSA fish cage farming, mainly in reference to the risk factors influencing diseases outbreaks in the cage farmed fish, their influence on productive parameters and their possible public health implications.

This paper deals specifically with the results of parasitological and histological analyses carried out during the BOMOSA project, while the results of bacteriological examinations will be presented in a successive paper.

MATERIALS AND METHODS

From March 2007 to October 2008 a total of 982 tilapias (*Oreochromis niloticus niloticus*) were examined, 685 from Kenya, 222 from Uganda and 75 from Ethiopia (Table 1).

Oreochromis niloticus niloticus	N. examined	Wild	Caged	Pond farmed
Kenya	685	56	184	445
Uganda	222	137	85	0
Ethiopia	75	65	10	0
Total	982	258	279	445

Table 1 - Number of tilapias examined from different countries and aquatic systems.
Tabella 1 - Numero di tilapie esaminate suddivise in base alla provenienza e al sistema di allevamento.

In Kenya the tilapias were sampled from 6 BOMOSA sites (Figure 1): Sagana Fish Farm (430 pond farmed and 27 caged fish); Sangoro Farm (15 pond farmed); Ruthagati Dam (6 wild and 35 caged); Ngeki Dam (50 wild and 97 caged); Ngei Dam (8 caged) and Lukenya Dam (17 caged).

In Uganda from 3 BOMOSA sites: Kasolwe Dam (65 wild and 27 caged); Ndolwe Dam (67 wild and 41 caged) and Busoga Dam (5 wild and 17 caged).

In Ethiopia from 6 sites: Babogaya Lake (12 wild); Hora Lake (12 wild); Alagae Dam (10 wild); Wonji Dam (28 - 18 wild and 10 caged); Awassa and Chamo Lakes (13 wild).

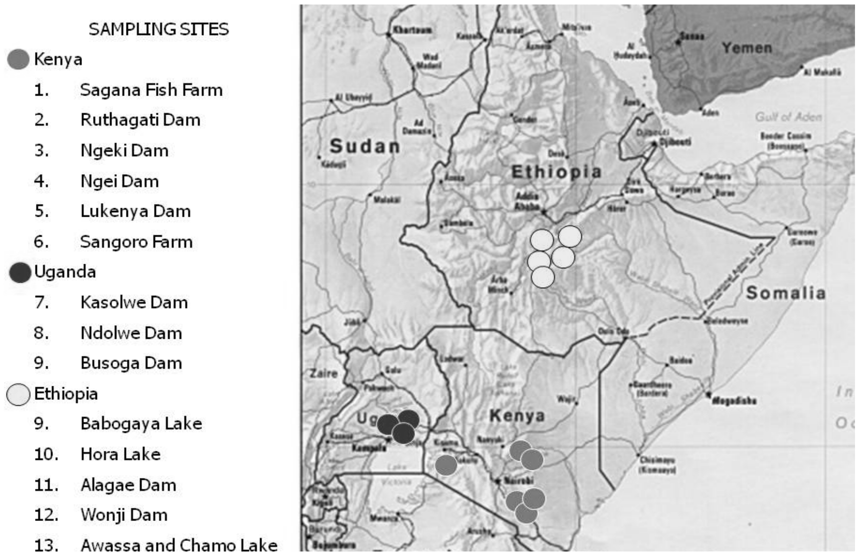


Figure 1 - Sampling sites.
Figura 1 - Siti di campionamento.

Necropsy and parasitological analyses were performed on all the sampled fish, following specific diagnostic protocols compiled on the basis of standard procedures.

For the identification of the parasite groups the taxonomic keys from the following manuals were used: Dawes (1956), Ukoli (1966), Kabata (1970), Lom & Dyková (1992), Hoffman (1999) and Scholz *et al.* (2004). Furthermore a number of scientific papers specific for the parasite groups recovered during the survey were collected and utilized for both identification and interpretation of the results. The manual "*Parasites, infections and diseases of fishes in Africa. An update*" (Paperna, 1996) was constantly consulted as a reference text during the survey.

The identification of protozoan and myxozoan parasites was mainly based on the morphological characters of fresh specimens, while metazoans were isolated and fixed in 70% ethanol and then identified after clarification and/or staining. Some parasites were observed also by scanning electron microscope (SEM).

Intensity of infection, abundance and prevalence were calculated for all the recovered parasites following the indications of Bush *et al.* (1997).

The identification of monogeneans was carried out with the aid of a phase-contrast microscope after partial digestion of the parasite and mounting in ammonium picrate and glycerin, following the method of Harris & Cable (2000).

For identification of clinostomids, in addition to the morphological analyses, the total DNA was extracted from 10 metacercariae (6 from skin and 4 from gills) and 5 adult of *Clinostomum* spp. The 18S, ITS, 28S rRNA gene were amplified by single or nested PCR, with different sets of primers (Hillis & Dixon, 1991; Cribb *et al.*, 1998; Mariaux, 1998; Lockyer *et al*., 2003). PCR products were then sequenced (PRIMM, Milano - Italy).

In some cases, infected tissues were fixed in 10% neutral buffered formalin and subjected to histological exams by standard techniques in order to study the histopathological effects induced by parasites and evaluate their pathogenic effects on the host.

RESULTS AND DISCUSSION

Quantitative data

A total of 805 (82%) out of 982 tilapias were positive for parasites, among which 589 (86%) out of 685 in Kenya, 178 (80.2%) out of 222 in Uganda and 38 (50.6%) out of 75 tilapia examined in Ethiopia (Figure 2).

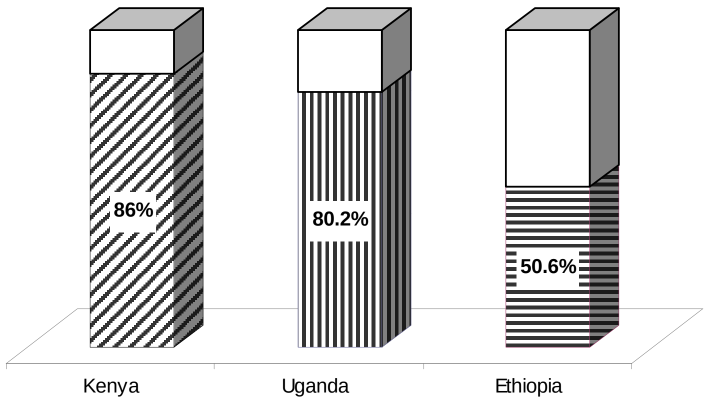


Figure 2 – Parasite prevalence in tilapia from different countries.
Figura 2 – Prevalenza di parassiti in tilapia nei diversi paesi.

During the survey, fish from different environments (cages of BOMOSA plots, open ponds and water reservoirs) were taken into consideration in order to evaluate the factors which could influence the parasite diffusion and outbreak of diseases. In all the following Figures and tables the caged fish will be indicated as "caged", the pond farmed fish as "pond" and the fish from water reservoirs as "wild".

In Figure 3 the values of parasite prevalence in the different systems are shown.

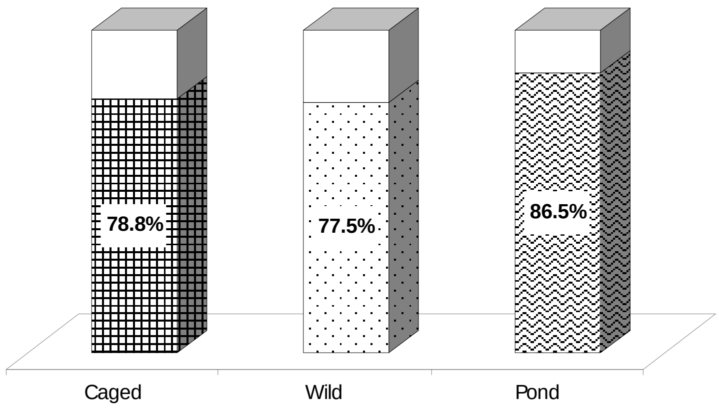


Figure 3 – Parasite prevalence in tilapia from different aquatic systems.
Figura 3 – Prevalenza di parassiti in tilapia nei diversi sistemi d'allevamento.

High prevalence values were observed in all the aquatic systems considered, without substantial differences.

The detailed results concerning quantitative parasitological data will be given in the following sections in relation to the different countries under study.

In addition to overall data, prevalence values will be given also in relation to parasite groups and aquatic systems. Furthermore, the values of mean intensity and abundance will be reported in relation to parasites and systems, and the prevalence of the main groups of parasites will be compared among the aquatic systems.

Kenya

The parasite prevalence values observed in pond farmed, caged and wild fish from sampling sites in Kenya are presented in Figure 4 and Table 2.

No remarkable differences were observed among the aquatic systems, with percentage of positivity slightly higher in farmed fish with respect to the wild ones.

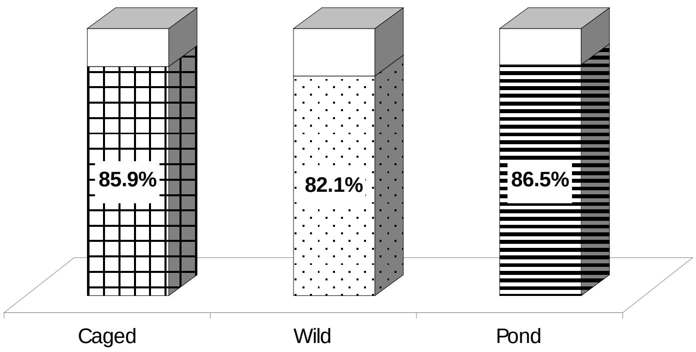


Figure 4 - Kenya: parasite prevalence in tilapia from different aquatic systems.
Figura 4 - Kenya: prevalenza di parassiti in tilapia nei diversi sistemi d'allevamento.

Kenya	Examined	Positive	Prevalence
Caged	184	158	85.9%
Wild	56	46	82.1%
Pond	445	385	86.5%
Total	685	589	82.0%

Table 2 – Kenya: prevalence of parasites in tilapia from different aquatic systems.
Tabella 2 – Kenya: prevalenza di parassiti in tilapia nei diversi sistemi d'allevamento.

As overall results, in Kenya 23 groups of parasites were isolated from the fish examined during the survey, as shown in Figure 5.

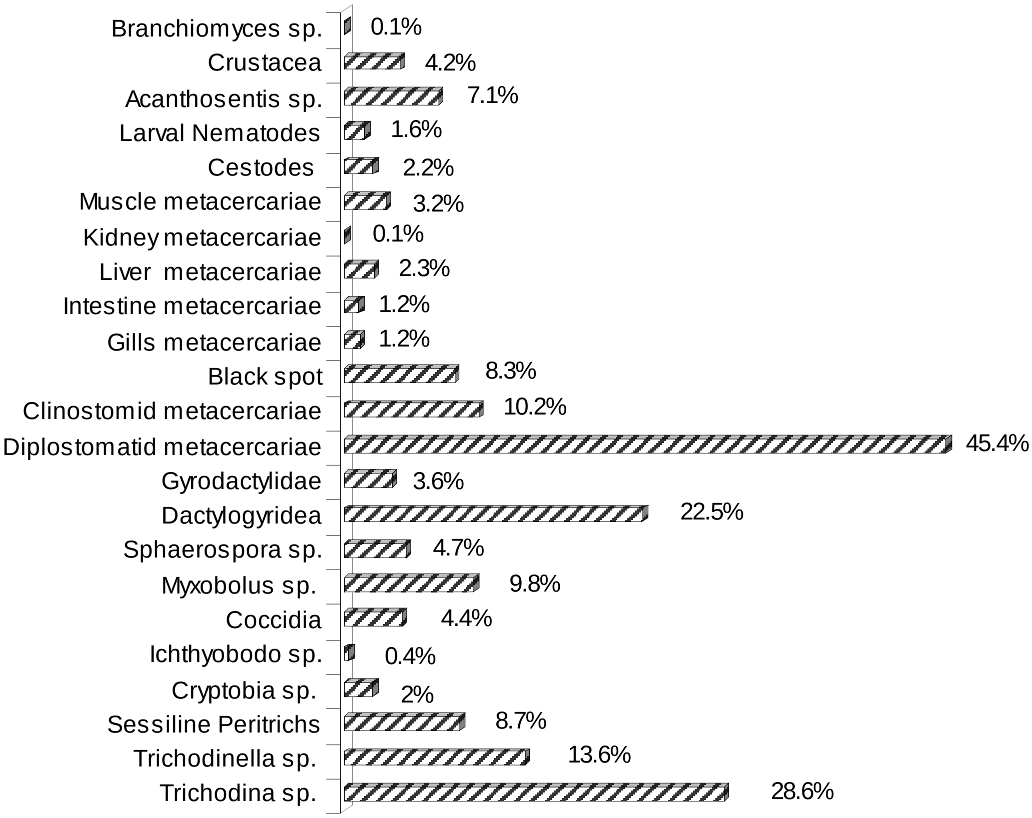


Figure 5 - Kenya: prevalence of different parasite groups in tilapia.

Figura 5 - Kenya: prevalenza di diversi gruppi parassitari in tilapia.

The highest value was referred to digenean diplostomatid metacercariae (45.4%), followed by ciliates (*Trichodina* sp. 28.6%) and monogenean dactylogyrids (22.5%), while the other parasite groups showed always prevalence values <13.6%, with sporadic findings of *Branchiomyces* sp. fungi, *Ichthyobodo* sp. flagellates and unidentified metacercariae from different body districts.

In Table 3 prevalence, mean intensity and abundance of the parasites found in tilapia from the different aquatic systems in Kenya are presented.

Parasite	Prevalence			Mean Intensity			Abundance		
	Caged	**Wild**	**Pond**	**Caged**	**Wild**	**Pond**	**Caged**	**Wild**	**Pond**
Trichodina sp.	22.2%	1.8%	34.6%	low	low	low	–	–	–
Trichodinella sp.	27.7%	1.8%	9.2%	low	low	low	–	–	–
Sessiline Peritrichs	13.0%	–	8.1%	low	–	low	–	–	–
Cryptobia sp.	–	–	3.0%	–	–	low	–	–	–
Ichthyobodo sp.	0.5%	–	0.4%	low	–	low	–	–	–
Coccidia	10.3%	1.8%	2.2%	high	high	medium	–	–	–
Myxobolus sp.	16.3%	–	8.3%	low	–	low	–	–	–
Sphaerospora sp.	11.4%	1.8%	2.2%	low	very low	low	–	–	–
Dactylogyridea	35.3%	16.1%	18.0%	3.7	1.3	4.9	1.3	0.2	0.8
Gyrodactylids	8.1%	–	2.2%	3	–	5.1	0.2	–	0.1
Diplostomatid metacercariae	28.3%	50.0%	52.0%	11.6	3.8	6.5	3.3	1.8	3.4
Clinostomid metacercariae	6.0%	1.8%	13.0%	5.25	32	6.9	0.2	0.6	0.62
Black spot	16.3%	1.8%	5.8%	2	29	3.1	0.3	0.52	0.2
Gills metacercariae	4.3%	1.8%	0.9%	3	2	2	0.13	0.03	0.01
Intestine metacercariae	1.6%	3.6%	0.7%	4.6	30.5	39.6	0.07	1.1	0.26
Liver metacercariae	0.5%	25.0%	0.2%	44	42.5	8	0.2	10.6	0.01
Muscle metacercariae	–	–	0.2%	4.3	–	3	0.5	–	0.006
Cestodes	–	14.3%	0.4%	4.4	2.5	40	0.1	0.35	0.17
Larval Nematodes	–	8.9%	1.1%	1	4.2	1.6	0	0.4	0.01
Acanthosentis sp.	–	7.1%	7.9%	2.5	1	2.9	0.13	0.07	0.2
Crustacea	–	1.8%	5.2%	1.4	1	4	0.03	0.02	0.2
Branchiomyces sp.	–	–	0.2%	–	–	–	–	–	–

Table 3 - Kenya: prevalence, mean intensity and abundance of parasites in tilapia from different aquatic systems.
Tabella 3 - Kenya: prevalenza, intensità media ed abbondanza di parassiti in tilapia nei diversi sistemi d'allevamento.

In Figures 6, 7 and 8 the prevalence values of different parasite groups found in tilapia from the different aquatic systems are shown.

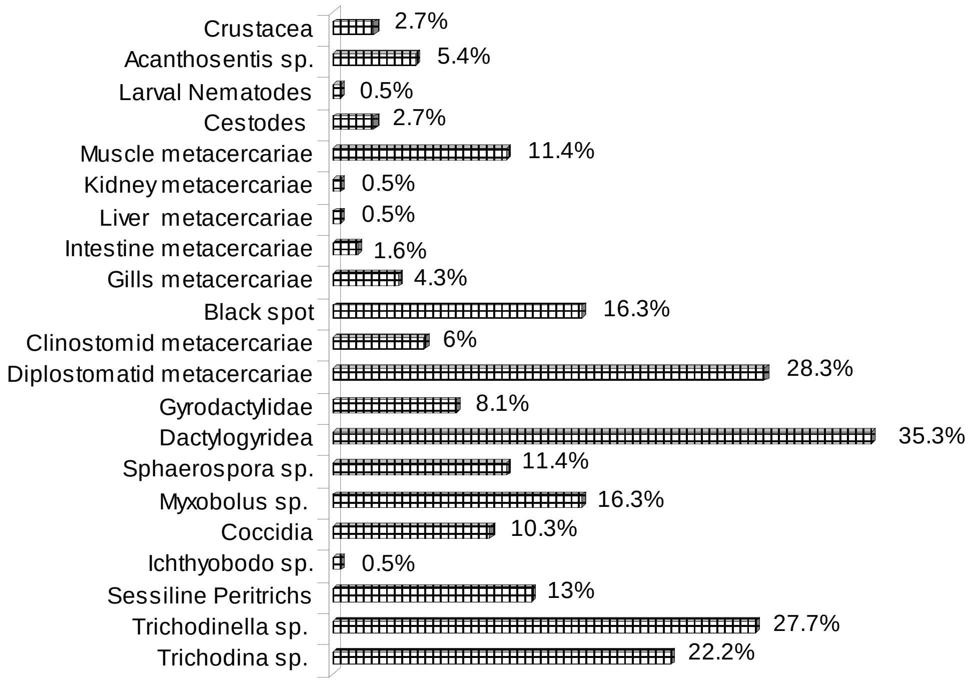


Figure 6 - Kenya: prevalence of different parasite groups in caged tilapia.
Figura 6 - Kenya: prevalenza di diversi gruppi parassitari in tilapie allevate in gabbia.

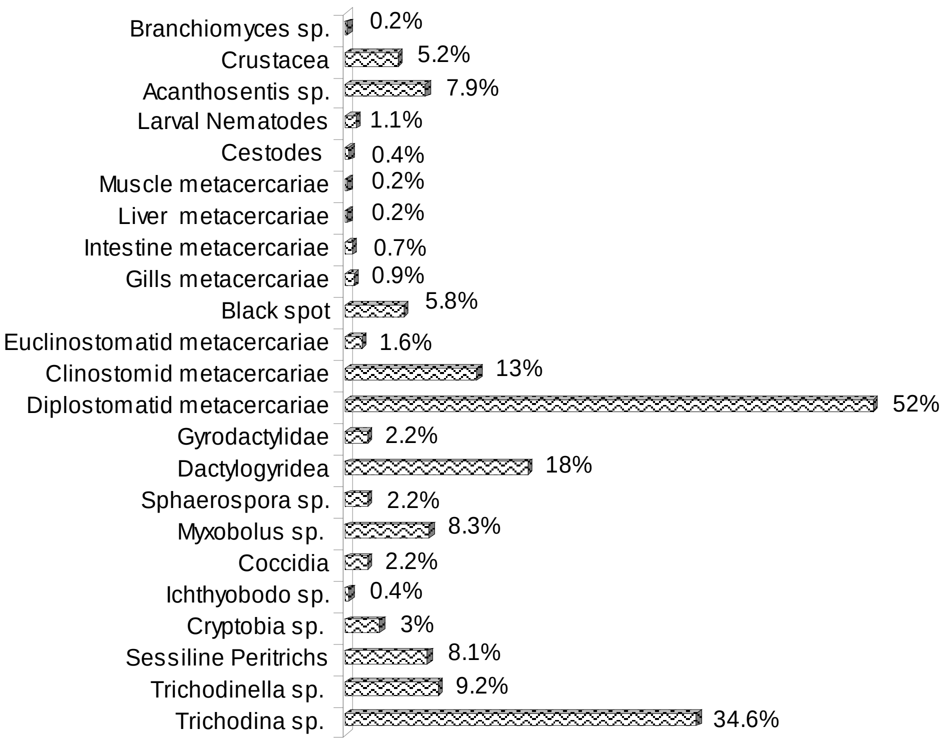


Figure 7 - Kenya: prevalence of different parasite groups in pond farmed tilapia.
Figura 7 - Kenya: prevalenza di diversi gruppi parassitari in tilapie allevate in bacini in terra.

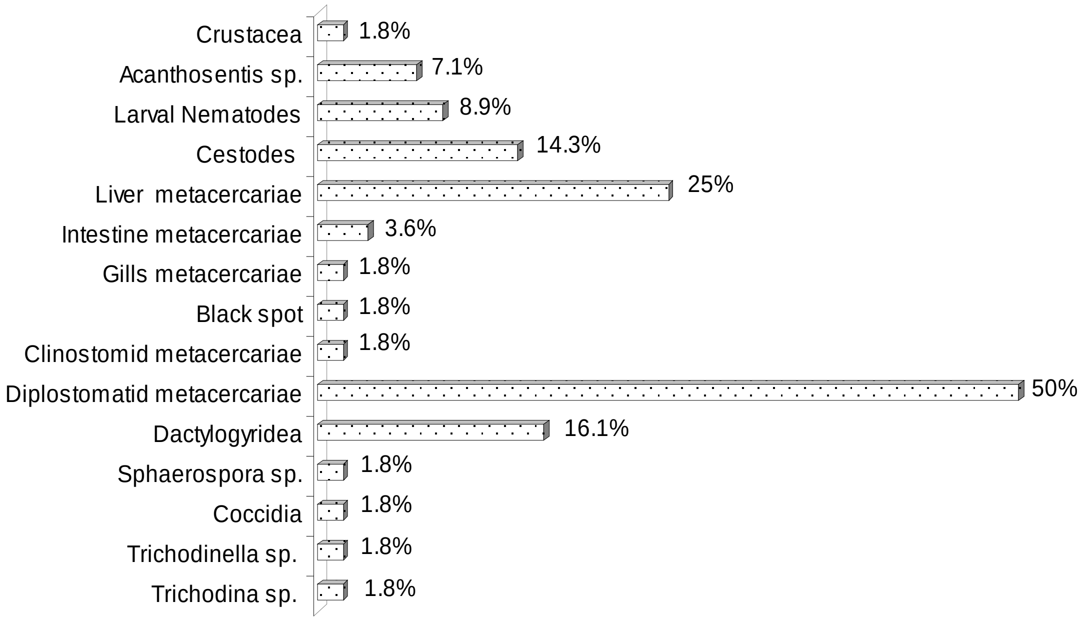


Figure 8 - Kenya: prevalence of different parasite groups in wild tilapia.
Figura 8 - Kenya: prevalenza di diversi gruppi parassitari in tilapia selvatiche.

In order to better compare the differences among prevalence values and composition of the parasitofauna recovered in tilapia from different aquatic systems, in the following figures (Figures 9-14) the results are presented in a comparative scheme for all the main groups of parasites.

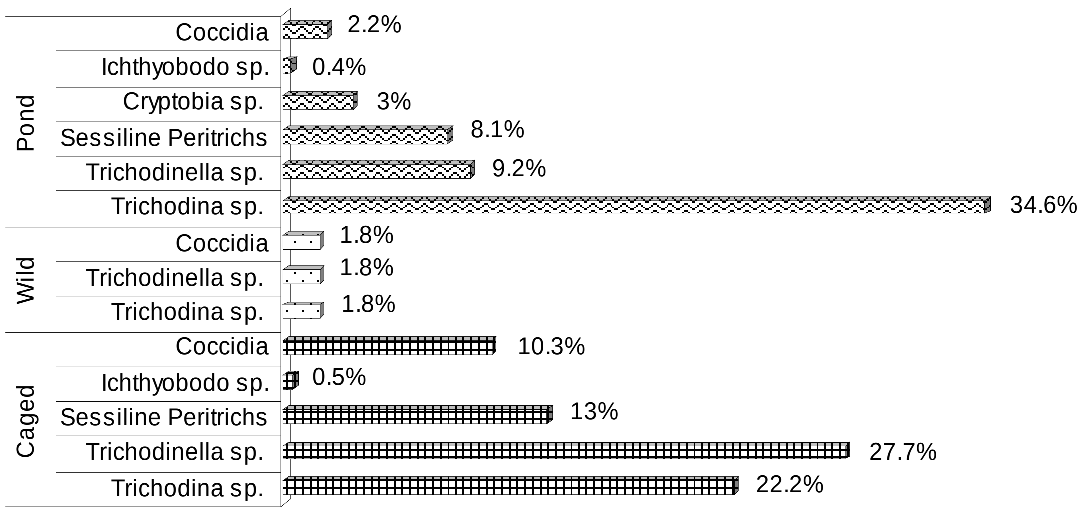


Figure 9 - Kenya: prevalence of protozoan parasites in different aquatic systems.
Figura 9 – Kenya: prevalenza di parassiti protozoi nei diversi sistemi acquatici.

Concerning the protozoan parasites, striking differences were observed concerning the aquatic systems, with positivity percentages always higher in farmed fish than in wild ones. Only very low prevalence of coccidia and mobiline ciliates (*Trichodina* sp. and *Trichodinella* sp.) were detected in wild fish, where flagellates and sessiline ciliates were never found.

This results confirm the opportunistic behavior of ciliates as trichodinids and peritrichs which are strongly influenced by environmental and zootechnical factors, such as biomass density, organic matter load, etc. (see section "qualitative data").

Furthermore all the protozoans found in tilapias during this survey show a direct life cycle, with an enhanced propagation in confined and crowded systems such as aquaculture facilities.

Unlike the above described protozoans, myxozoan parasites show a complex life cycle involving benthic invertebrates as intermediate/alternate hosts (see section "qualitative data"). Therefore their different distribution among aquatic systems is mainly regulated by the presence/absence of the invertebrates hosting infective stages.

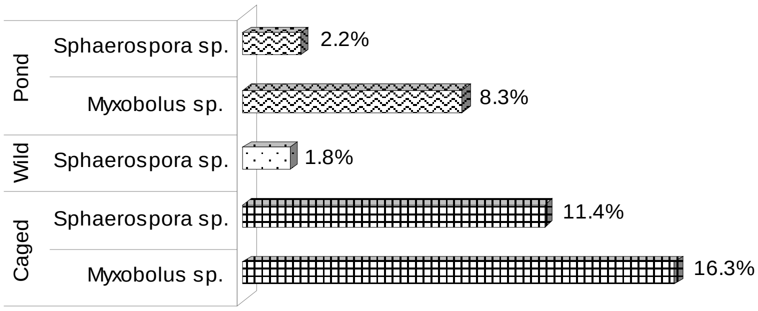


Figure 10 - Kenya: prevalence of Myxozoan parasites in different aquatic systems.
Figura 10 – Kenya: prevalenza di parassiti Myxozoa nei diversi sistemi acquatici.

Monogeneans are direct life cycle ectoparasites and their distribution is strongly influenced by stocking densities (mainly for gyrodactylids) and environmental factors (see section "qualitative data"). So the absence of gyrodactylid infections in wild fish can be related to the rare contact fish-to-fish, more frequent in farmed fish. On the contrary dactylogyrids show transmission strategies feasible both in wild and farmed fish, even if enhancing conditions are mainly present in farming systems.

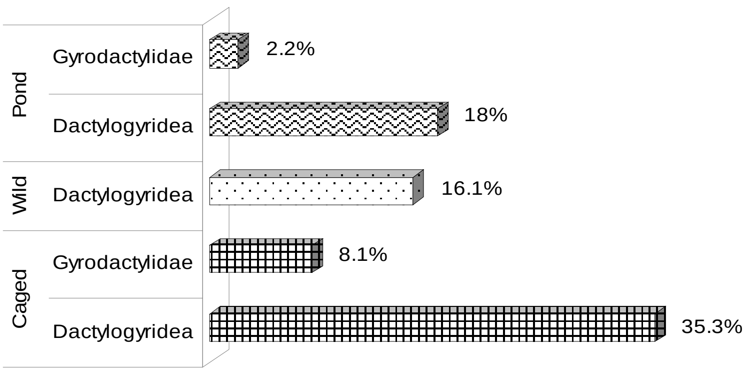


Figure 11 - Kenya: prevalence of monogenean parasites in different aquatic systems.
Figura 11 - Kenya: prevalenza di parassiti monogenei nei diversi sistemi acquatici.

Digeneans show a very complex life cycle and their distribution in fish as metacercariae follows the diffusion of intermediate and definitive hosts (see section “qualitative data”). Therefore the differences observed among prevalence of digenean metacercariae in tilapias from different aquatic systems is to be referred mainly to biotic factors such as presence/absence of snails and piscivorous birds.

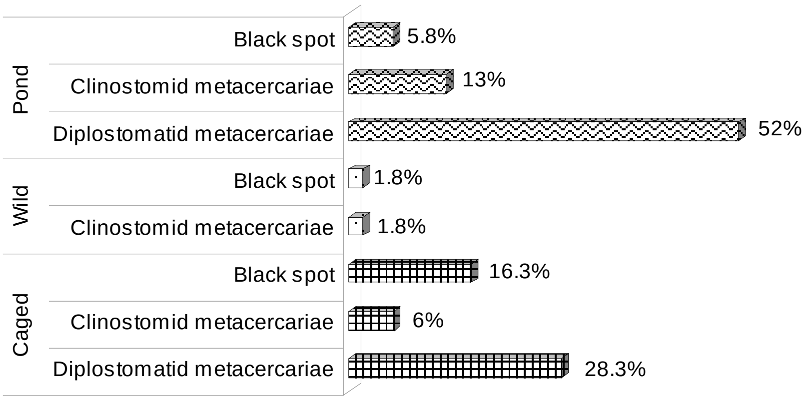


Figure 12 - Kenya: prevalence of digenean metacercariae in different aquatic systems.
Figura 12 - Kenya: prevalenza di metacercarie di digenei nei diversi sistemi acquatici.

Metacercariae of diplostomatids were the most frequent digeneans found in tilapias from all the aquatic systems, maybe for a huge diffusion of suitable snail hosts in the environments under study. On the contrary, Black Spot and clinostomid infections showed higher prevalence in farmed fish, probably in relation also to the abundance of birds suitable definitive hosts around farming sites (e.g. herons for clinostomids).

Cestodes, nematodes and acanthocephalans (Figure 13) are all parasites characterized by an indirect life cycle with crustaceans (copepods and amphipods) as intermediate host. The highest prevalence values observed in wild tilapias can be explained with the different feeding habits, based on natural zooplankton. On the contrary, caged tilapias feed on pelleted food with a reduced intake of plankton and a consequent reduced risk of ingestion of infective larval stages of these parasites.

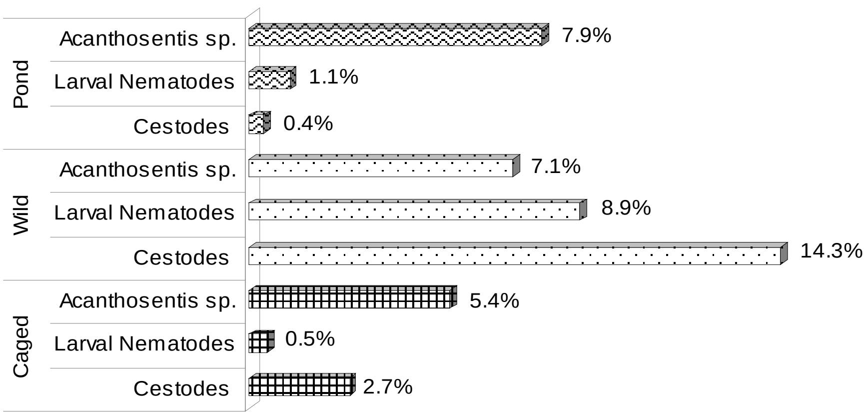


Figure 13 - Kenya: prevalence of cestodes, nematodes and acanthocephalans in different aquatic systems.
Figura 13 - Kenya: prevalenza di cestodi, nematodi e acantocefali nei diversi sistemi acquatici.

Crustacean parasites of fish show a direct life cycle strongly influenced by environmental factors (see section "qualitative data"). Copepod crustaceans were always found at low prevalence, but with a higher value in pond farmed fish for the presence of conditioning factors as shallow waters, low water exchange, presence of algae, etc.

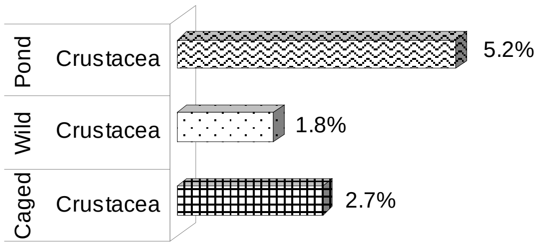


Figure 14 - Kenya: prevalence of crustacean parasites in different aquatic systems.
Figura 14 - Kenya: prevalenza di crostacei parassiti nei diversi sistemi acquatici.

Uganda

In Figure 15, the parasite groups recovered in tilapias from Uganda are shown.

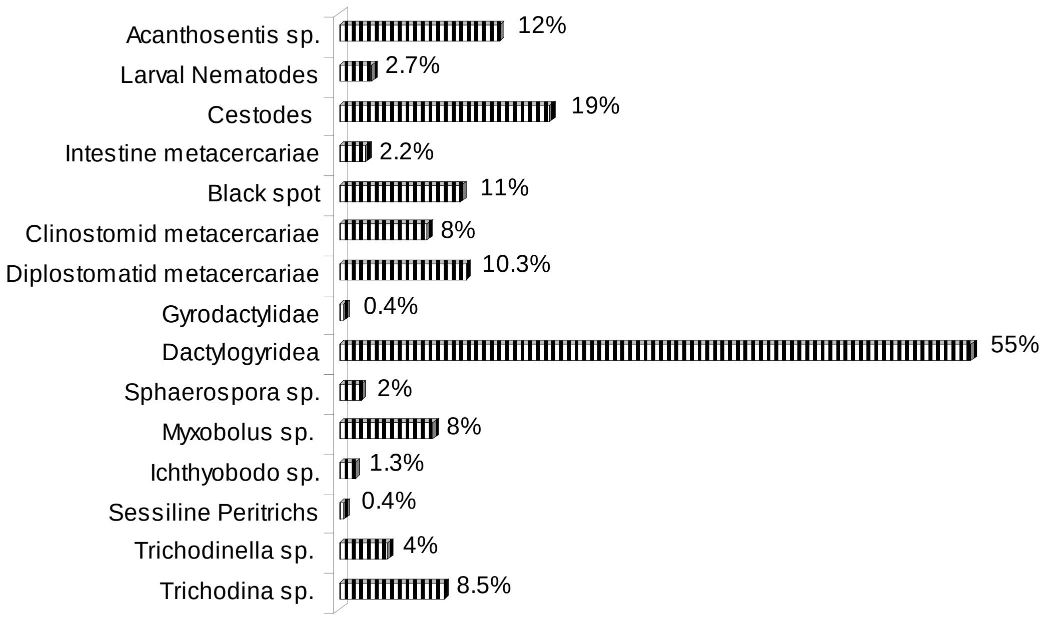


Figure 15 - Uganda: prevalence of different parasite groups in tilapia.
Figura 15 - Uganda: prevalenza dei diversi gruppi parassitari in tilapia.

The highest value was referred to monogenean dactylogyroids (55%), followed by cestodes at the larval stage (19%) and acanthocephalans owing to *Acanthosentis* genus (12%), while the other parasite groups showed always percentage of positivity values <11%, with sporadic findings of gyrodactylid monogeneans and sessiline peritrichs.

In Table 4 prevalence, mean intensity and abundance of parasites found in tilapia from the different aquatic systems in Uganda are presented.

Parasite	Prevalence		Mean Intensity		Abundance	
	Caged	Wild	Caged	Wild	Caged	Wild
Trichodina sp.	18.8%	2.2%	low	low	–	–
Trichodinella sp.	5.9%	2.9%	low	low	–	–
Sessiline peritrichs	1.2%	–	low	–	–	–
Ichthyobodo sp.	2.3%	0.7%	low	low	–	–
Myxobolus sp.	9.4%	6.5%	low	low	–	–
Sphaerospora sp.	2.3%	1.4%	low	low	–	–
Dactylogyridea	31.7%	63.5%	2.5	6.1	0.8	3.9
Gyrodactylidae	1.2%	–	2	–	0.03	–
Diplostomatid metacercariae	2.3%	15.3%	1	2.9	0.02	0.46
Clinostomid metacercariae	–	11.6%	–	3.4	–	0.4
Black spot	3.5%	13.8%	1.6	2.6	0.07	0.4
Intestine metacercariae	–	3.6%	–	30.4	–	1.15
Cestodes	4.7%	–	2.5	–	0.14	–
Larval Nematodes	5.9%	–	2.6	–	0.15	–
Acanthosentis sp.	17.6%	10.2%	1.7	1.6	0.3	0.16

Table 4 - Uganda: prevalence, mean intensity and abundance of parasite groups in caged and wild tilapia.
Tabella 4 - Uganda: prevalenza, intensità media e abbondanza dei diversi gruppi parassitari in tilapie selvatiche e allevate in gabbia.

In Figures 16 and 17 the prevalence values of parasite groups found in wild and caged tilapia are shown respectively.

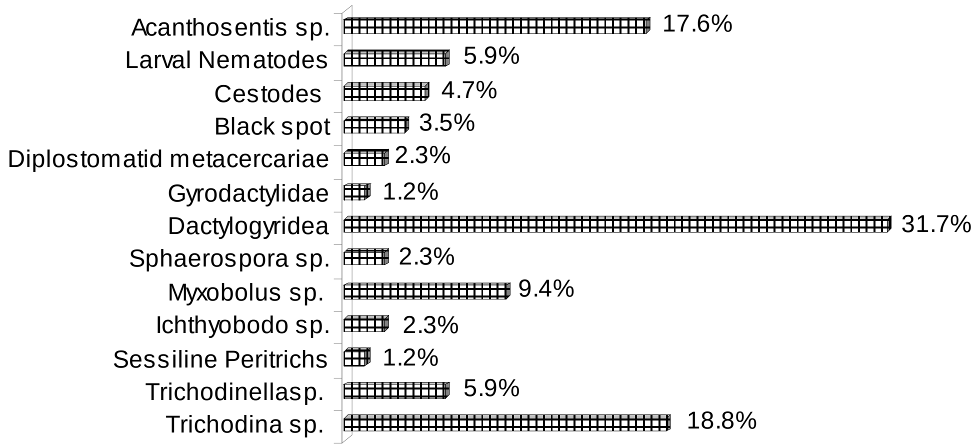


Figure 16 - Uganda: prevalence of parasites in caged tilapia.
Figura 16 - Uganda: prevalenza di parassiti in tilapie allevate in gabbia.

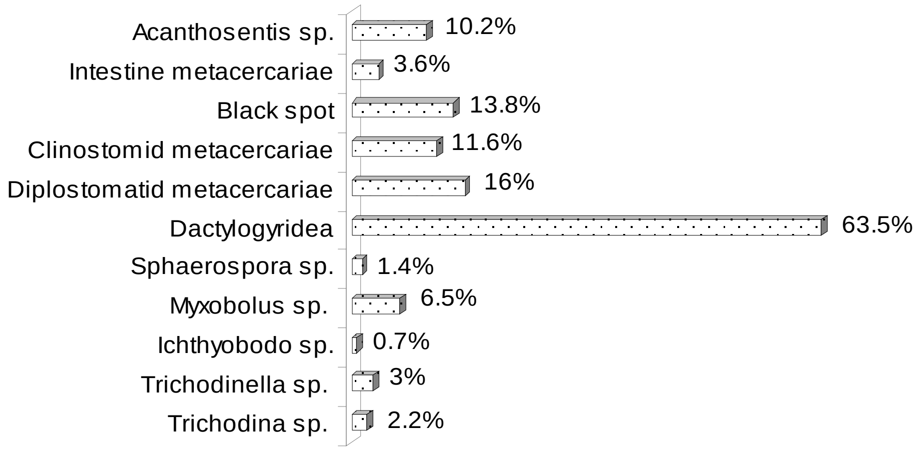


Figure 17 - Uganda: prevalence of parasites in wild tilapia.
Figura 17 - Uganda: prevalenza di parassiti in tilapie selvatiche.

Similarly to Kenya, protozoan parasites were detected in farmed fish with prevalence values higher than those observed in wild fish, in which only trichodinid ciliates were found (except for sporadic finding of *Ichthyobodo* sp. flagellates) always at low intensity.

Dactylogyrids occurred at high prevalence both in farmed and wild fish, indicating the existence of conditioning factors such as shallow waters and low water exchange in both the aquatic systems. The percentage of positivity for digenean metacercariae were higher in wild fish, as expected, while *Acanthosentis* sp. acanthocephalans showed a huger diffusion in caged fish, may be for a different composition of crustacean populations in the two systems.

In order to better compare the differences among prevalence values and composition of the parasitofauna recovered in tilapia from different aquatic systems, in the following figures (Figures 18-22) the results are presented in a comparative scheme for all the main groups of parasites.

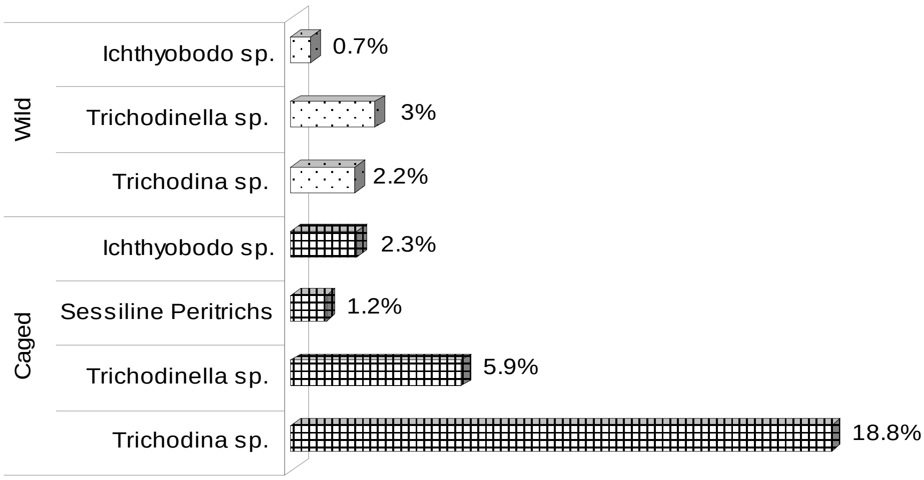


Figure 18 - Uganda: prevalence of protozoan parasites in different aquatic systems.
Figura 18 - Uganda: prevalenza di parassiti protozoi nei diversi sistemi acquatici.

The higher prevalence values of protozoan parasites detected in farmed fish - always at low intensity - were always low can be easily explained with the confined conditions of the caged fish which could enhance the transmission dynamics of these opportunistic ectoparasites.

The differences among prevalence of myxozoan parasites in wild and caged tilapias were not remarkable, indicating the presence of suitable invertebrate hosts (benthic annelids) in all the environments taken into considerations.

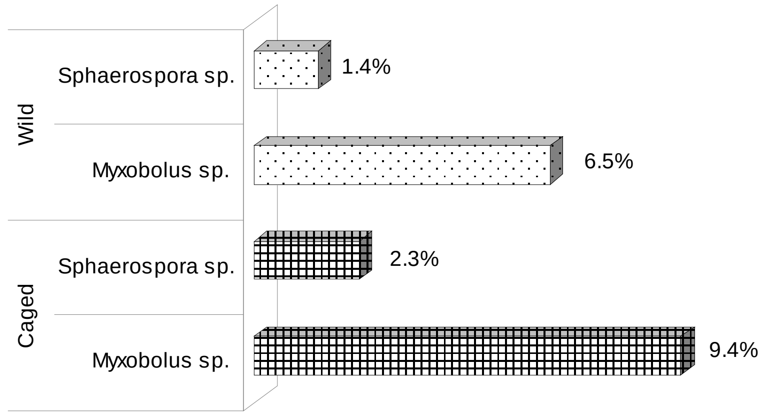


Figure 19 - Uganda: prevalence of Myxozoan parasites in different aquatic systems.
Figura 19 - Uganda: prevalenza di parassiti Myxozoa nei diversi sistemi acquatici.

Concerning monogenean ectoparasites, gyrodactylids were not detected in wild fish, probably for the rare contact fish-to-fish requested for the transmission of these monogeneans. On the contrary dactylogyrids showed high prevalence both in caged and wild fish (even if with higher values in the latter, 63.5%), maybe for the presence of condition enviromental factors enhancing the transmission dynamics (see section "qualitative data").

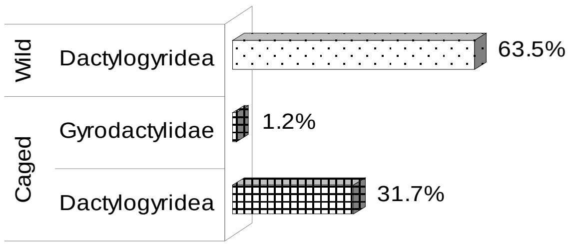


Figure 20 - Uganda: prevalence of monogenean parasites in different aquatic systems.
Figura 20 - Uganda: prevalenza di parassiti monogenei nei diversi sistemi acquatici.

Digenean metacercariae were found mainly in wild fish. In caged fish a very low percentage of positivity were recorded, and clinostomid larval stages were not detected at all. In order to explain these data, a survey on the distribution of gastropod species suitable as intemediate host should be carried out in the sites where plots are present.

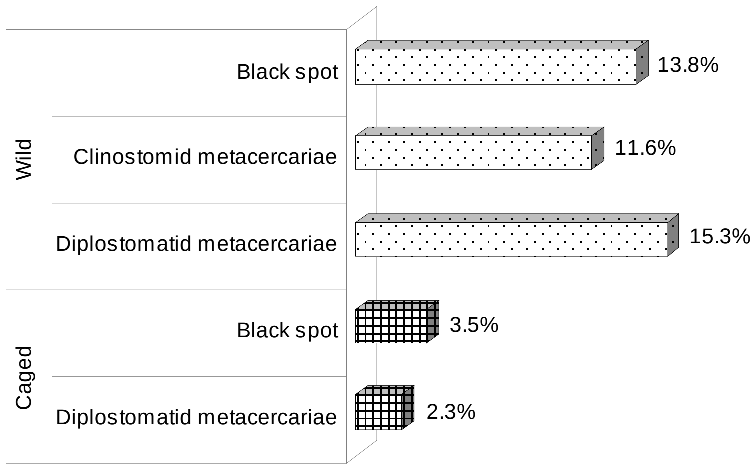


Figure 21- Uganda: prevalence of digenean metacercariae in different aquatic systems.
Figura 21 - Uganda: prevalenza di metacercarie di digenei nei diversi sistemi acquatici.

Surprisingly, cestodes and nematodes at the larval stage were detected only in caged fish, while acanthocephalans owing to *Acanthosentis* genus were present in both caged and wild fish even if with different values (Figure 22).

At this regard, the piscivorous birds acting as definitive hosts of cestodes and nematodes may be more abundant in the crowded plot area, leading to a massive contamination of the planktonic population in the environment. Furthermore, a not correct feeding procedure (qualitative or quantitative) of the caged fish could have caused an active predation of planktonic invertebrates by the farmed fish population.

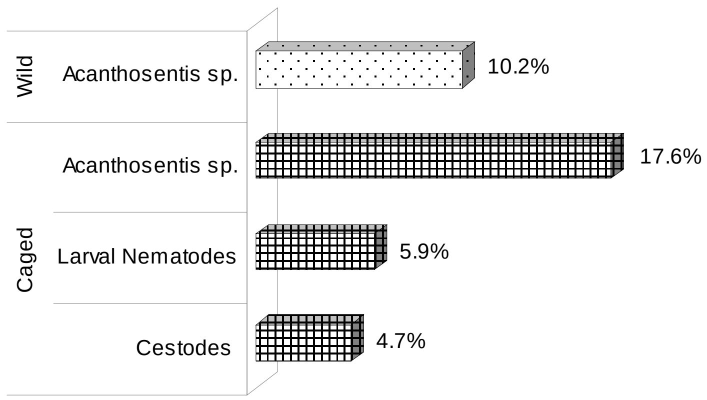


Figure 22 - Uganda: prevalence of cestodes, nematodes and acanthocephalans in different aquatic systems.
Figura 22 - Uganda: prevalenza di cestodi, nematodi e acantocefali nei diversi sistemi acquatici.

Ethiopia

In Figure 23 the parasite groups recovered in tilapias from Ethiopia are shown.

The prevalence values of several parasite groups show remarkable differences in comparison to those observed in tilapias from Kenya and Uganda, in fact the highest value was referred to clinostomid metacercariae (32.0%) and larval nematodes (17.3%), followed by monogenean dactylogyrids (16.0%).

The other parasite groups showed always percentage of positivity values <6.6%, with sporadic findings of *Cryptobia* sp. flagellates, monogenean gyrodactylids, *Trichodina* sp. ciliates and crustaceans.

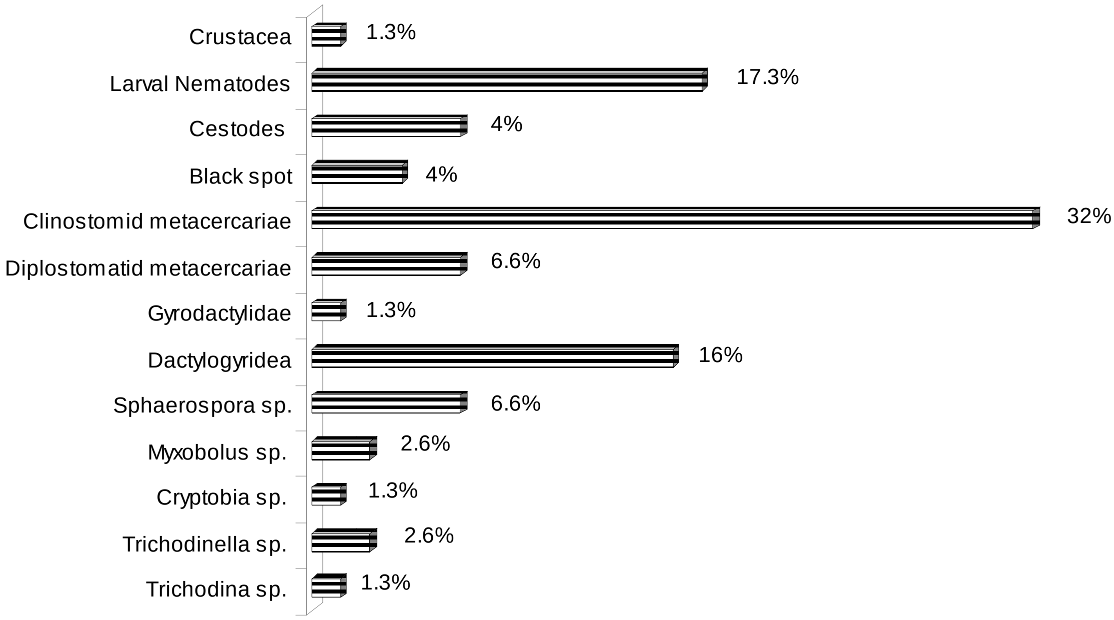


Figure 23 - Ethiopia: prevalence of parasites in tilapia.
Figura 23 - Etiopia: percentuale di positività per parassiti in tilapia.

In Table 5 prevalence, mean intensity and abundance of parasites found in tilapia from the different aquatic systems in Ethiopia are presented.

Parasite	Prevalence		Mean Intensity		Abundance	
	Caged	Wild	Caged	Wild	Caged	Wild
Trichodina sp.	–	1.5%	–	very low	–	–
Trichodinella sp.	–	3.1%	–	low	–	–
Cryptobia sp.	–	1.5%	–	low	–	–
Myxobolus sp.	–	3.1%	–	low	–	–
Sphaerospora sp.	–	7.7%	–	low	–	–
Dactylogyridea	–	18.5%	–	4.4	–	0.8
Gyrodactylidae	–	1.5%	–	5	–	1.4
Diplostomatid metacercariae	–	7.7%	–	5.2	–	0.4
Clinostomid metacercariae	30.0%	32.3%	1	3.9	0.3	0.9
Black spot	–	4.6%	–	3	–	0.1
Cestodes	10.0%	6.1%	1	3	0.1	0.2
Larval Nematodes	30.0%	15.4%	6	3.6	1.8	0.5
Crustacea	–	1.5%	–	1	–	0.01

Table 5 - Ethiopia: prevalence, mean intensity and abundance of the parasites found in caged and wild tilapia.
Tabella 5 - Etiopia: prevalenza, intensità media e abbondanza di parassiti in tilapie selvatiche ed allevate in gabbia.

In Figures 24 and 25 the prevalence values of the parasite groups found in caged and wild tilapias are shown.

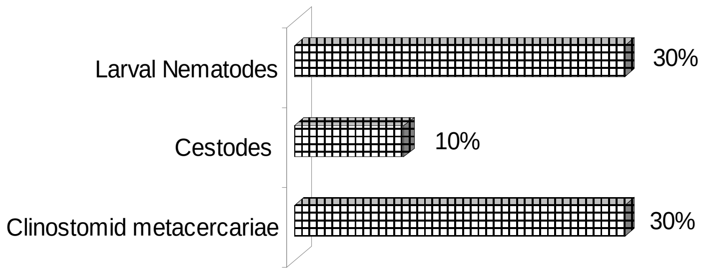


Figure 24 - Ethiopia: prevalence of different parasite groups in caged tilapia.
Figura 24 - Etiopia: prevalenza dei diversi gruppi parassitari in tilapie allevate in gabbia.

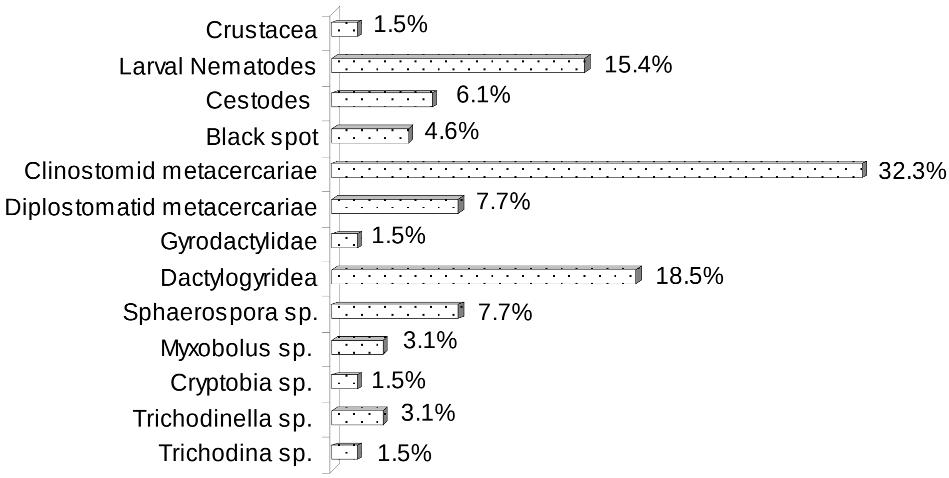


Figure 25 - Ethiopia: prevalence of different parasite groups in wild tilapia.
Figura 25 - Etiopia: prevalenza dei diversi gruppi parassitari in tilapie selvatiche.

The results of parasitological analyses obtained from caged fish are not significant, as only ten subjects from BOMOSA cages were examined. Clinostomids, larval cestodes and larval nematodes were found both in caged and wild fish indicating the presence of similar biotic risk factors in the aquatic environments.

The parasitofauna of wild tilapias showed a composition similar to those of tilapias from Kenya and Uganda.

The high prevalence values of larval stages of nematodes, cestodes and clinostomid digeneans have to be related to the abundance of piscivorous birds in all the sites under study. In fact fish-eating birds are the definitive hosts of all the recovered parasites owing to these groups and they disseminate the eggs in the water environment, where the intermediate hosts (crustaceans for cestodes and nematodes, snails for clinostomid digeneans) are widely present and spread the infection to fish population.

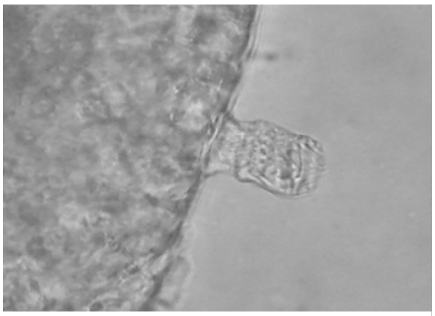
Qualitative data and pathological findings

In this section the main parasite groups found during the two year survey will be shortly presented and discussed with main reference to their occurrence and diffusion in the different aquatic environments taken into consideration, the histological lesions and their possible influence on host health and on the productivity of farmed fish.

During this survey members of four protozoan groups were found, belonging to ciliates (phylum Ciliophora), flagellates (phylum Euglenozoa) and coccidian (phylum Apicomplexa). Although the occurrence of protozoan ectoparasites can be considered a common finding in farmed fish, as most of them are ubiquitous in aquatic environment, in particular conditions they could reach very high intensities and act as opportunistic pathogenic agents on stressed or traumatized fish (Paperna, 1996).

Ciliates

Sessiline peritrichs are ectocommensal ciliates not feeding on fish tissue but filtering their nutrients from the water. They don't usually inflict serious injuries to the host and are not primary agents of disease, but sometimes in debilitated fish they proliferate and could mechanically affect the gas exchange of gill epithelium with resulting respiratory problems (Woo, 1995; Paperna, 1996). There were several reports on poor condition and mortality linked to massive infections by sessilians (Fijan, 1961; Meyer, 1970; Paperna *et al.*, 1984; Lightner *et al.*, 1988; Paperna, 1991).



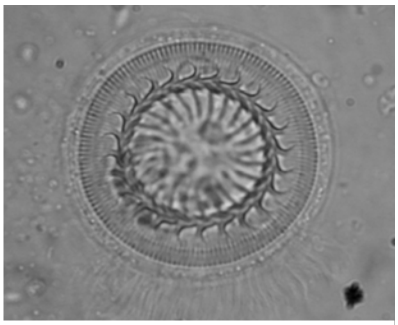
Sessiline peritrich attached to tilapia gill.
Peritrico sessile attaccato a branchie di tilapia.

Sessiline peritrichs were detected only in gills of caged and pond farmed fish, always at low level of infection. Their occurrence only in farmed fish could be related to a less active swimming behavior of caged fish and to high organic load, as typical in farming systems carried out in earth-based pond. Histology did not show any lesion referable to these parasites in the gills of infected fish.

Trichodinids are the most common ectoparasites of fish and in some cases they are able of inflicting heavy damages to stressed host with resulting mortality. Trichodinids are commensals and they use their host as a convenient feeding ground for waterborne particles, bacteria and debris from the fish surface (Woo, 1995). There are data on the presence of trichodinids in a large number of aquatic systems in tropical Africa (Fryer & Iles, 1972), but data on taxonomy are limited only to a few sites (El Tantawy & Kazubsky, 1986).

Heavily infected fish may show hypermelanosis for mucus secretion and epithelial sloughing off, with fin erosion frequently due to secondary bacterial infections, and respiratory distress in the case of heavy gill infections.

Trichodina spp. and *Trichodinella* spp. were found in all the systems examined, with high prevalence only in caged and pond farmed fish (e.g. 49.9% and 43.8% in caged and pond farmed tilapia from Kenya; 30.5% in caged fish from Uganda). Anyway massive infections were never detected and infection intensity was always low. In our study the low biomass density present in the



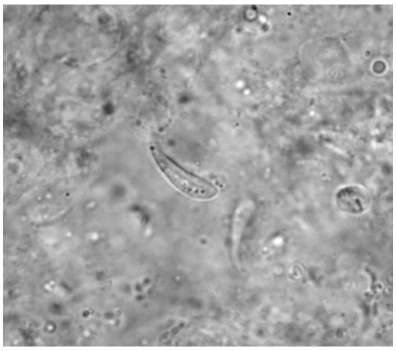
Trichodina sp. from gills of tilapia.
Trichodina *sp. da branchie di tilapia.*

farming systems taken into consideration allowed to avoid the conditions favoring the colonization and diffusion of these opportunistic protozoans potentially pathogenic for fish. No lesions were observed in the histological sections from gills of positive tilapia.

Flagellates

Bodonids are flagellates with oval or elongated body usually less than 10 µm and with 2 flagella. In the genus *Cryptobia* the species colonizing skin and gills are considered as ectocommensals, while other species are able to parasitize intestines or circulatory system (Lom & Dyková, 1992). Unlike haematozoic congeneric species, ectozoic *Cryptobia* have a direct life cycle and fish-to-fish transmission is favored by conditions such as low water exchange, high organic load and high biomass density. The attack of the parasite through the flagella in general does not induce any cellular damage or pathological alterations (Lom, 1980), even if some data exist on high morbidity associated with this parasite (Woo, 1987).

In the fish examined during this survey the prevalence, mean intensity and abundance values for *Cryptobia* spp. were generally low and referred only to open pond farmed fish, except for one wild subject from Ethiopia. The intensity was always low and no histological lesions were referred to the presence of the flagellate.



Ichthyobodo necator from tilapia gill.
Ichthyobodo necator *in branchie di tilapia.*

Ichthyobodo necator is a true ectoparasite presenting a free-swimming stage alternating with an attached trophic stage. It represents a serious sanitary risk for farmed fish, mainly fry and fingerlings, where it can reach high intensity and cause destruction of epithelial or epidermal cells with serious pathological changes also due to secondary bacterial infections (Lom & Dyková, 1992). Fish massively parasitized show anorexia and mucus hypersecretion. This parasite was sporadically found mainly in caged fish, always at low intensity and with no correlations to pathological changes.

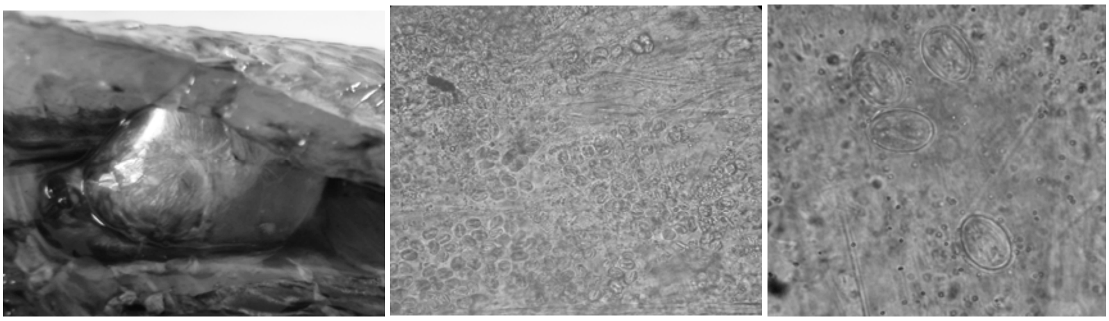
I. necator has to be considered as a potential threat to the sanitary conditions of tilapia fingerlings, mainly in overcrowding and poor hygienic situation or when stressful events occur (e.g. hatchery stocking, transportation, handling, etc.).

Coccidia

Intracellular protozoans parasitizing numerous animal species. They also infect numerous species of freshwater and marine fish, but information concerning the detrimental effects on fish is limited. Coccidia may be significant pathogens of cultured fish that are subjected to overcrowding, intensive management and factors contributing to diseases in fish may parallel those noted in other animals, i.e., age, immune status, nutrition, and stress (Dyková & Lom, 1981). The coccidia have been frequently found in African cichlids in Africa, i.e. *Eimeria vanas* and *Goussia cichlidarum* in tilapias from Uganda and South Africa (Paperna, 1996). *G. cichlidarum* has been recently described in tilapias from Egypt (El-Mansy, 2008). Although fish coccidia often show extraintestinal tropism, most of the infections are described in epithelial cells of intestines and swim bladder.

In this survey swim bladder coccidian referable to the species *G. cichlidarum* were found almost exclusively in farmed (cages and ponds, with higher prevalence and intensities in the former) tilapia from Kenya. The high fish density in cages may facilitate the direct oro-fecal transmission of oocysts among fish.

Histology allowed to detect *G. cichlidarum* oocysts in all the layers of positive swim bladder wall, in particular in the compact fibrous layer, in the lumen, in the connective tissue and in the *rete mirabilis*. Erythrocytes were infiltrated between gas gland epithelial cells with presence of eosinophilic cells. In heavy infected swim bladder, necrosis was present in the thin layer of the gas bladder epithelium. Eosinophilic granular cells were also scattered in the swim bladder serosa.



Left: gross lesions such as thickening and hemorrhages in swim bladder heavy infected by *Goussia cichlidarum*; Middle: several oocysts in fresh scraping from infected tilapia swim bladder; Right: a particular of *Goussia cichlidarum* mature oocysts.
A sinistra: lesioni macroscopiche (ispessimento ed emorragie) in vescica natatoria massivamente infetta da Goussia cichlidarum*; in mezzo: numerose oocisti in preparato a fresco da vescica natatoria parassitata; a destra: oocisti mature di* Goussia cichlidarum.

On the basis of histological observations and of gross lesions (heavy infected swim bladder showed wall thickening and haemorrhages) it could be stated that coccidiosis by *G. cichlidarum* may represent a potential risk for the health of farmed tilapia, mainly when environmental and farming factors enhance the survival and transmission of mature oocysts among fish.

Although the functional impairment of the swim bladder could not be relevant in the life of farmed fish, the indirect effect of this coccidiosis on general health status and productivity of farmed tilapias should be better assessed.

Myxozoa

Myxozoa are multicellular parasites of organ cavities (celozoic) and tissues (histozoic) of fish (Lom & Dyková, 1992). Except for *Enteromyxum leei*, myxozoan parasites show an indirect (alternate) cycle involving invertebrates (annelids or bryozoans) and vertebrates (mainly fish) hosts.

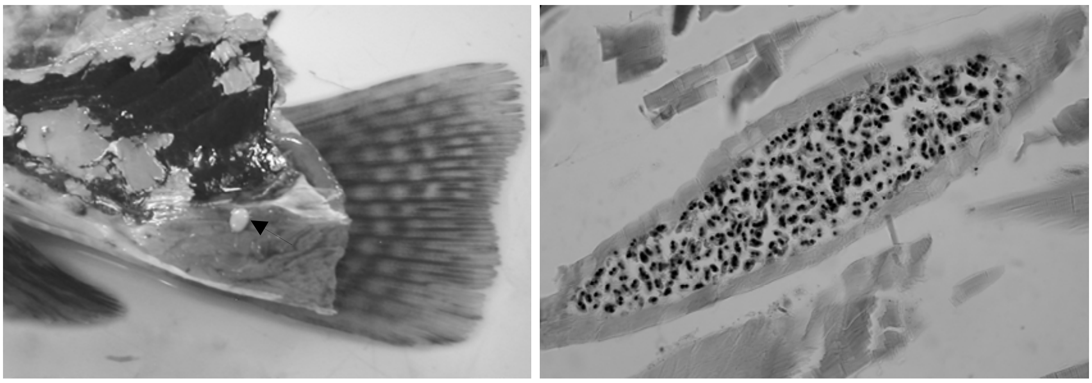
Data beyond African myxozoan species descriptions are available from Egypt (Fahmy *et al*., 1975), East Africa (Baker, 1963; Paperna, 1973), Ghana (Paperna, 1968; 1973), Cameroon (Fomena *et al*., 1985; Fomena & Bouix, 1987). Infections due to *Myxobolus, Thelohanellus* and *Henneguya* have been reported in both East African and West African waters and the species are apparently host specific (Paperna, 1996). The life cycles of myxozoans detected in African fish are not yet completely clarified.

The occurrence of myxozoan parasites has been investigated extensively also in fish farms because their potential pathogenic effect on their hosts (Okaeme *et al*., 1988; Obiekezie & Okaeme, 1990; Tarer *et al*., 1996), mainly in case of histozoic localizations in functionally relevant organs.

In African fish, histozoic myxosporidians have been described mainly as skin and gill infections (Paperna, 1973; Abolarin, 1974; Fomena *et al.*, 1985; Landsberg, 1986). *Myxobolus* cysts occur also in the interior organs, muscles and eyes mainly in cichlids, but also in fish from other families (Paperna, 1973; El-Mansy, 2005). Infections (even heavy infections) often occur in fish otherwise in good condition. Nonetheless, multiple cysts and particularly large cysts in vulnerable organs such as the gills, may compromise fish health, and macroscopically visible cysts in the muscle may compromise the market value of fish products. Prevalence of infections is variable on the basis of host species and seasonality (Fryer, 1961; Baker, 1963; Paperna, 1973).

During this survey members of two myxozoan genera (*Myxobolus* and *Sphaerospora*) were found. In particular *Myxobolus* spp. spores were detected in gills, muscle, intestine and kidney, sometimes encysted in macroscopically visible cysts, of farmed fish in Kenya and Uganda, and in few wild fish in Uganda and Ethiopia. Prevalence values were extremely variable among sites, may be due to the different distribution of the benthic annelids which represent suitable alternate host for *Myxobolus* spp. A correct identification of the parasite species by morphological and molecular techniques, and the detection of infected invertebrate hosts may allow in future to clarify the introduction/diffusion dynamics and define preventive measures.

The prevalence of *Myxobolus* spp. observed in this study is lower than that reported by Gbankoto *et al.* (2001) who found the same genus in gills of *Tilapia zillii* and by El-Mansy (2005), who reported *M. heterosporus* from the eyes of *O. niloticus* in River Nile, Egypt. In Africa, the first description of myxosporeans was given by Baker (1963), followed by a number of studies, such as those of Paperna (1968), Abolarin (1974), Obiekezie & Okaeme (1990), Fomena & Bouix (1994), Kostoingue & Toguebaye (1994), Fomena & Bouix (1997).



Left: macroscopically visible cyst of *Myxobolus* (arrow) in the posterior lateral muscle of tilapia;
right: histological section of a muscular fiber filled with *Myxobolus* spores.
A sinistra: cisti macroscopica di Myxobolus *sp. nel muscolo posteriore laterale di tilapia;*
a destra: sezione istologica di fibra muscolare ripiena di spore di Myxobolus *sp.*

Histological lesions were different with reference to the localization of *Myxobolus* cysts. In gills heavy infected by *Myxobolus* spp. mild to severe gill hyperplasia in the distal portion of filaments, vascular congestion and teleangectasia were observed. Furthermore necrosis and hemorrhages with cellular debris and bacteria were also noticed. The myxozoan cysts were encapsulated by a double layered thick wall. In the kidney the spores were often surrounded by melanomacrophages in the interstitial tissue with rarefaction of hematopoietic component. In the intestine no lesions were referred to the presence of spores, always sparse and in low number, while in the muscle the cysts were in general represented by fibers engulfed of a plenty of spores without clear pathological changes except for a slight atrophy in the surrounding muscular tissue.

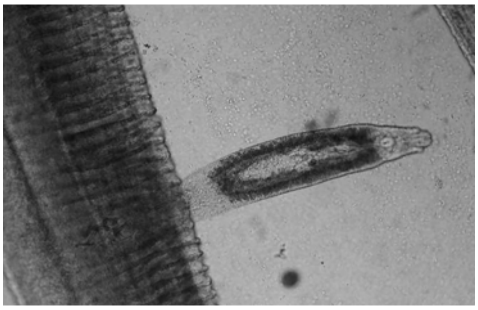
With reference to *Sphaerospora* spp., spores were found in the kidney of farmed and wild tilapias from Kenya, Uganda and Ethiopia, in general at low/medium intensity. The lack of descriptions of *Sphaerospora* sp. from African tilapia makes the identification at species level difficult and need further studies aimed also to clarify the potential pathogenic effect of the parasite. In fact other renal *Sphaerospora* such as *S. renicola* in cyprinids may cause heavy kidney infections and inflammation in affected organs. In our study histology allowed to detect the presence of immature stages of *Sphaerospora* spp. in kidney interstitial tissue and in the wall of tubules, and of mature spores into the tubular lumen. In some cases spores seem to be strongly attached to tubular epithelial cells and associated to severe degeneration and necrosis of epithelium.

Myxozoan parasites may represent a risk factor for health and productivity of farmed tilapia. For this reason preventive measures should aim to reduce the colonization of farming environments by benthic annelids, for example by the application of fallowing procedures in order to clean the bottom and break the infection transmission dynamics of these parasites.

Monogeneans

Monogeneans are small flattened ectoparasites (Platyhelminthes) attached by characteristic organs (opisthaptor) to the host skin, fins or gills. Among monogenean dactylogyroids, the genus *Cichlidogyrus* Paperna, 1960, infecting the gills, and *Enterogyrus* Paperna, 1963, endoparasitic in the stomach, are host specific to diverse cichlid fish species from Africa (Paperna, 1996). Monogeneans have been reported to cause severe mortalities in fish hatcheries in Nigeria (Obiakezie & Taege, 1991) and South Africa in catfish, black bass and freshwater ornamental fish. Overcrowding of fish into culture ponds or tanks, together with different environmental and management factors, promote heavy infestations, which can lead to productive losses, tissue damages and in some cases mortality (Hecht & Endemann, 1998).

The species recorded so far on wild and cultured Nile tilapia (*Oreochromis niloticus*) are 19, but none of these has ever been reported in Kenya. These monogeneans are common in different species of tilapia and displays a wide geographic distribution (Pariselle & Euzet, 1995; 2003), the majority of species descriptions were from Egypt, Senegal, Ivory Coast, Zimbabwe, Uganda, Nigeria, Philippines, Bangladesh and Mexico (Ergens, 1981; Okaeme *et al.*, 1988; Flores-Crespo *et al.*, 1992; Douëllou, 1993; Ferdousi & Chandra, 2002).



Cichlidogyrus sp. in tilapia gills.
Cichlidogyrus *sp. in branchie di tilapia.*

Among the fish examined in this survey, *Cichlidogyrus* specimens collected from tilapias belonged to four different species: *C. tubicirrus magnus* Paperna & Thurston, 1969, *Cichlidogyrus* sp. *sensu* Ergens, 1981, *Cichlidogyrus* sp. type 1 and *Cichlidogyrus* sp. type 2.

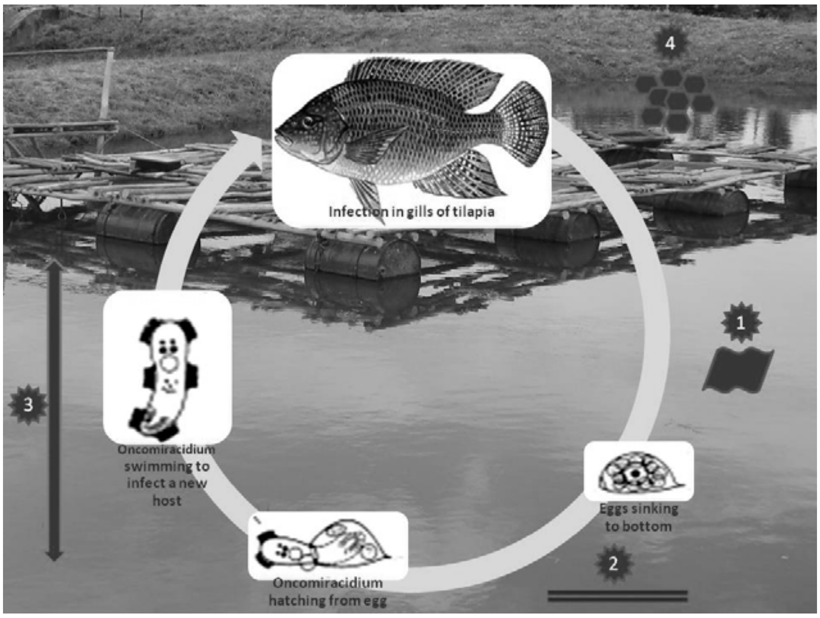
C. tubicirrus magnus was found for the first time on gills of *O. niloticus* in Uganda by Paperna & Thurston (1969) and then has been reported from *Tilapia zilli* Gervais, 1848 in Egypt by Ergens (1981).

Regarding the control of these monogeneans in Africa, spontaneous immunity responses have been documented (Paperna, 1996). In fact, some studies on humoral antibody response of the tilapia against *Cichlidogyrus* spp. suggest that tilapia is capable of producing an induced humoral immune response (Sandoval-Gío *et al*., 2008). It's always better to apply good management practices to avoid parasite diffusion and consequently improve fish ponds productivity.

Dactylogyroids referable to *Cichlidogyrus* spp. were found in all the environments investigated; the highest prevalence was found in wild fish from Uganda (65.1%) but relevant percentage of positivity were also observed in caged and pond farmed tilapias from all the considered countries. Anyway the infection intensity was generally low with mean values around 4 parasites/host.

Histological observations did not allowed to refer relevant pathological alterations to infestations due to *Cichlidogyrus* spp. at low intensities, except for a mild gill disease with light hyperplasia and oedema and slight epithelial sloughing off.

On the basis of the life cycle pattern of dactylogyroid parasites, their presence in both wild and farmed fish is easily explained. In fact the risk factors favoring the spread of infection are related to water environment characterized by low water exchange, poor bottom hygiene and short water column depth. Stocking density may represent a further conditioning factor leading to heavy gill infestation in farmed fish. Biosecurity measures aimed to prevent and control this parasitosis should take into consideration all these risk factors.

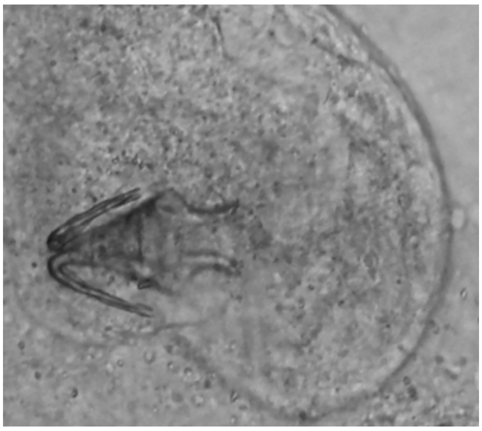


Life cycle of dactylogyroid parasites with main risk factors favouring their transmission/spread: 1) low water exchange; 2) poor bottom hygiene; 3) short distance cage-bottom; 4) overcrowding.

Ciclo biologico di parassiti dactilogiroidei con i principali fattori che ne influenzano la trasmissione/diffusione: 1) basso ricambio idrico; 2) scarsa igiene del fondo; 3) ridotta distanza gabbia-fondale; 4) sovraffollamento.

Monogeneans belonging to the family Gyrodactylidae are ubiquitous ectoparasites of teleost fish that have been extensively studied since the late 19th Century, mainly due to their unique reproductive adaptations. In fact the Gyrodactylidae are the only viviparous family within Monogenea; worms give birth to fully developed adults and intra-uterine embryos already contain second and often third generation embryos, recognized by their developing marginal hooks and two big anchors called *hamuli* (Paperna, 1996). This "Russian-Doll" mode of reproduction, termed hyperviviparity, combined with a rapid generation time, can result in exponential parasite population growth on a single host.

The life cycle is direct and the transmission can occur directly from host to host by contact, or by free swimming in the water column for few hours until they find a new host (Soleng et al., 1999). They feed on mucus and epithelial cells, and can move freely on the host. Even if high infection levels can cause host mortality, low parasite burdens can cause secondary pathology, such as fin clamping, which presumably inflicts severe fitness costs to the host, and alteration in feeding behavior up to anorexia, with productive losses.



Posterior part of *Gyrodactylus* spp. from tilapia skin.
Porzione posteriore di Gyrodactylus *spp. da cute di tilapia.*

Among the fish examined in this survey, *Gyrodactylus* spp. were found at low intensities on tilapias from cages and open ponds in Kenya and Uganda, and just in one wild fish from Ethiopia. The specimens recovered in this survey are probably new species and their description is in progress. Anyway, in literature the gyrodactylids described up to now in *O. niloticus* are *G. cichlidarum* Paperna, 1968 and *G. shariffi* Cone, Arthur & Bondad-Reantaso, 1995 (www.gyrodb.net).

The occurrence of gyrodactylids almost exclusively in farmed tilapia is well explained by the transmission route of these parasites, which spread among fish mainly by direct contact. For this reason, high stocking density is the most important risk factor for gyrodactylosis in aquaculture. Necroscopical and histological exams did not show any lesion in the examined fish, where very low intensity was always observed.

Digeneans

Digeneans are Platyhelminthes characterized by indirect life cycle and an extremely variable morphology. Fish can be intermediate or definitive host of digenean parasites, but the main sanitary problems in fish are caused by infections due to larval stages (metacercariae).

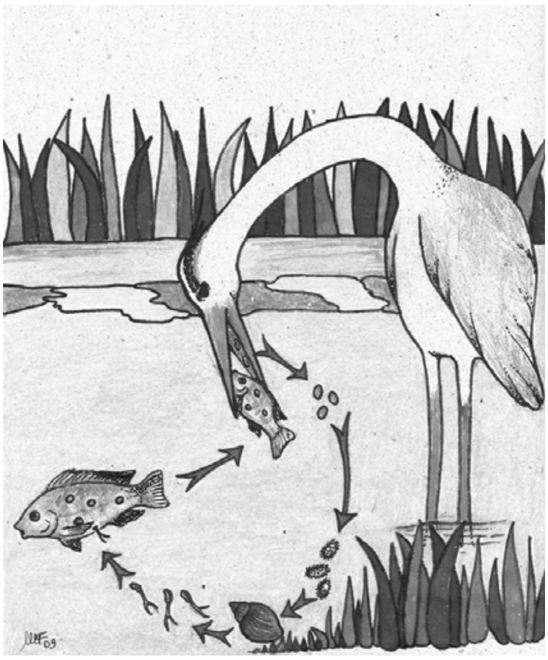
The life cycle of clinostomids and strigeoids (diplostomids and strigeids), which represent the most abundant digeneans found in tilapia during this survey, involves a fish-eating bird as definitive host, a gastropod as invertebrate first intermediate host and many fish species as second intermediate host.

The snail host of *Neascus* causing blackspot in Lake Victoria cichlids is the local bulinid, *Bulinus ugandae*, while in Israel, *B. truncatus* was shown to be the intermediate host for *Clinostomum tilapiae* (Finkelman, 1988). Elsewhere in Africa, where *B. truncatus* is absent, *C. tilapiae* is likely to be transmitted by other bulinids. Another bulinid, *Bulinus (Physopsis) globosus*, is the intermediate host for *Euclinostomum heterostomum* (Donges, 1974). *Clinostomum complanatum* develops through species of *Lymnaea (Radix)* (Lo *et al.*, 1982;

Finkelman, 1988). In diplostomids and clinostomids the cercariae hatched from eggs are fork-tailed (furcocercariae) and penetrate actively in the fish host mainly through the skin, going to develop in metacercaria in different organs with regard to parasite species.

Metacercarial infections were found in fish from all the studied inland water bodies in Africa and the Near East (Fahmy & Selim, 1959; Paperna, 1964a; Williams & Chaytor, 1966; Paperna & Thurston, 1968; Khalil, 1969; 1971; Van As & Basson, 1984).

Strigeoid metacercariae (Strigeidae, Diplostomatidae and Cyathocotylidae) encyst in a variety of organs, including the eye, remaining sometimes unencysted in the eye lens and retina. The cysts of some species (e.g. the larval genus *Neascus*, *Bolbophorus*, etc.) occurring in the skin, accumulate melanophores or other skin chromatophores causing "black spot" syndrome.



Life cycle of clinostomid digeneans.
Ciclo biologico di digenei clinostomidi.

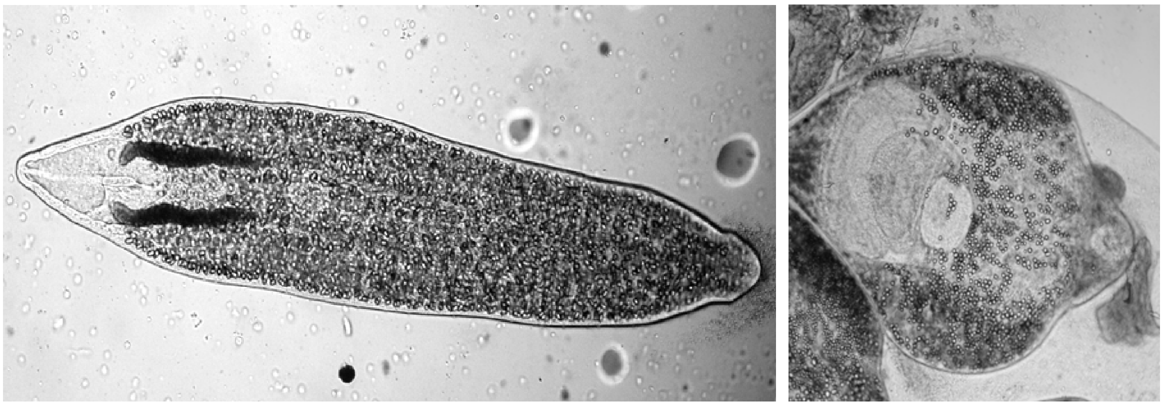
Clinical effects of infection are often not obvious, even if morbidity and mortality have been often associated with heavy metacercarial infection, mainly in young fish. The large (0.5-0.8 mm in diam.) and numerous (over 50) cysts of *B. levantinus*, established in muscles of young cichlids (<50 mm long), could produce severe body deformities (Paperna & Lengi, 1963; Yekutiel, 1985). Massive infections with metacercariae in juvenile (O-class) fish have been considered as an important cause of natural mortalities at this life stage, i.e. *Centrocestus* spp. in gills and *B. levantinus* in muscles of cichlid fish (Yekutiel, 1985; Farstey, 1986; Paperna, 1991).

In spite of the large size (3-7 mm) of the clinostomid cysts, neither skin infection nor muscle and visceral infections induce severe histopathology or gross pathological effects in fully grown or even juvenile fish. Seemingly healthy looking cichlids (*Tristramella simonis* in Lake Kinneret, Israel) are occasionally found virtually covered by cutaneous cysts of *Clinostomum* spp. (Paperna, 1964a; 1964b). Very young fish (*O. mossambicus*, 40-60 mm long), however, succumbed to infection by 3-5 cysts of *Euclinostomum heterostomum* in the viscera.

Eye metacercariae may impair eye vision, mainly when located in the lens. Although the specific lens parasite *Diplostomum spathaceum* is unknown till now from African waters, several infections by other diplostomatid metacercariae have been reported, usually invading the anterior or vitreous humor. Severe infection may lead to exophthalmia, cataracts, and even complete collapse of the eye, up to blindness (Thurston, 1965; Lombard, 1968; Douëllou, 1992).

Most of the metacercarial infections of fish eyes are referred to diplostomid and strigeid digeneans. In particular, *Diplostomum* spp. and *Tylodelphys* spp. are respectively reported from lens and vitreous humour of fish eyes over the world. The greatest damage is caused when the lenses are infected by non-encysted metacercariae of *D. spathaceum*, while eye flukes that live in the vitreous humour as *Tylodelphys* spp. or other *Diplostomum* species, are not yet clearly correlated to pathological effect. In general eye infections by diplostomatid metacercariae are considered of detrimental impact to fish activity and productivity, as feeding and therefore growth rates of the infected fish are negatively influenced.

In this survey, lens infections by *D. spathaceum* have never been detected, confirming the unsuitability of African aquatic environments for the development of this parasite, suggesting the absence of suitable first intermediate hosts and adverse environmental conditions. On the contrary, high prevalence of *Tylodelphys* spp. (diplostomatid unencysted metacercariae) and *Apharyngostrigea* spp. (strigeid encysted metacercariae) has been observed in Kenya with higher values in pond farmed (52%) and wild fish (50%).



Left: *Tylodelphys* sp. metacercaria from vitreous humour; right: *Apharyngostrigea* sp. metacercaria encysted in soft tissues back eyes.
A sinistra: metacercaria di Tylodelphys *sp. da umor vitreo; a destra: metacercaria di* Apharyngostrigea *sp. incistata nei tessuti molli della porzione posteriore dell'occhio.*

It has to be noticed how the infection intensity showed highest values (11.6 parasites/host) in caged fish where the percentage of positivity was lower (28.3%) than that registered in pond farmed and wild fish. This could be due to the confinement and reduced swimming activity of caged fish which are more susceptible to infection by cercariae shed by snails. In Uganda and Ethiopia only *Tylodelphys* spp. was found, always unencysted in the vitreous humour.

Histology did not show any lesions in the eyes infected by metacercariae collected during this study. Further surveys are necessary to identify the species of *Apharyngostrigea* here described and clarify its possible pathogenicity to the host, as it represents a never described parasite in tilapia.

Flukes of the family Clinostomidae contain the most important fish-eating bird parasites (Matthews & Cribb, 1998). The genus-type of this family is *Clinostomum* Leidy, 1856 (Kanev *et al*., 2002) that is characterized by a long and confusing taxonomic history.

Clinostomids are very widespread in Africa (Manter & Pritchard, 1969; Yimer & Enyew, 2003) and most of the researches on fish, second intermediate host of these digeneans, were carried out in Southern, West and Central Africa and just few in the North and East part of Africa (Aloo, 2002).

Humans may be an accidental host of these parasites as a consequence of consumption of raw or slightly cooked parasitized fish (Euzeby, 1984; Kakizoe *et al*., 2004).

During this study different species of clinostomids were isolated from tilapia, in particular at least two species belonging to the genus *Clinostomum* and *Euclinostomum heterostomum*, the last one generally found in the kidney as big encysted metacercariae protruding from the renal tissue. The two *Clinostomum* species differed significantly in morphology and localization: a smaller and yellow-whitish one from the skin and a bigger and yellowish one from the basis of gill arch could be observed. The morphological study of the collected specimens allowed to refer the skin metacercariae to *Clinostomum* "*cutaneum* type" described by Paperna (1964a; 1964b) and the gill arch metacercariae to *C. phalacrocoracis* Dubois, 1931.

Left: *Euclinostomum heterostomum* metacercariae from kidney; middle: *Clinostomum "cutaneum"* type metacercaria from skin; right: *C. phalacrocoracis* metacercaria from gill arch.
A sinistra: metacercaria di Euclinostomum heterostomum *da rene; in mezzo: metacercaria di* Clinostomum "cutaneum" *type da cute; a destra: metacercaria di* C. phalacrocoracis *da arco branchiale.*

The PCR confirmed the separation between the samples collected from the skin and the ones from the gills and allowed for the first time to obtain the sequence from these species, as till now only *C. complanatum* and *C. marginatum* partial sequences were available. Other partial sequence of *Clinostomum* sp. are available in GenBank but with any correlation to the morphological features of the specimens.

The presence of *Euclinostomum heterostomum* metacercariae has been recorded from the kidney of tilapias in Kenya (1.4%) and Ethiopia (9.3%) both in farmed and wild fish, but always with a mean intensity <3 parasites/host. This clinostomatid was never found in Ugandan fish. Histology showed the presence of big parasitic cysts in the kidney surrounded by a thick fibrous capsule with an external layer composed of eosinophilic granular cells and melanomacrophages. In the surrounding tissue vacuolar degeneration of tubules, congestion of glomeruli, hemorrhages of interstitial hematopoietic tissue with rarefaction were observed. These pathological alterations were limited to the infected area so low infection should not induce functional alteration in kidney of adult fish. On the contrary, in young fish also mild infections could be detrimental for the host health.

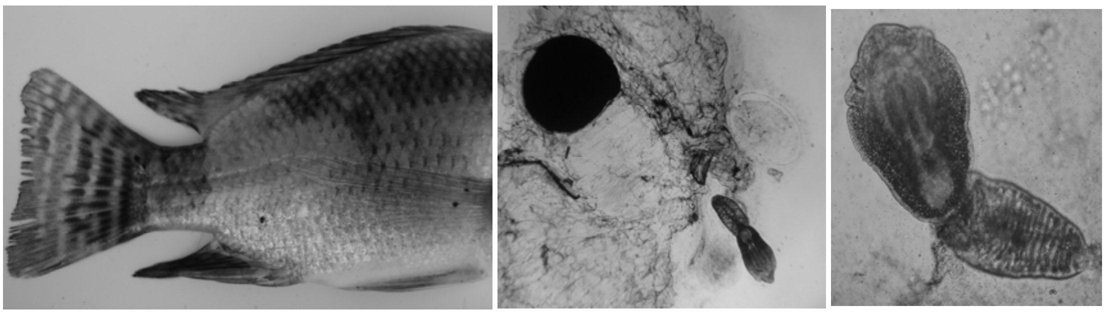
Histological section of tilapia kidney: encysted metacercaria of *Euclinostomum heterostomum*.
Sezione istologica di rene di tilapia: metacercaria incistata di Euclinostomum heterostomum.

Clinostomum spp. metacercariae were found in all the water environments in Kenya, Uganda and Ethiopia with the highest prevalence in the last country (22.6%) and mean intensity value always between 1 and 5.6 except for wild fish from Kenya where only one subject was parasitized by 32 skin metacercariae. The distribution of clinostomids in fish follows the diffusion of the suitable first intermediate host (gastropods) and definitive host (several piscivorous birds). For this reason the taxonomic identification of metacercariae is essential to better understand their biology and epidemiology and to define biosecurity procedures aimed to prevent and control the parasitosis in farmed fish.

Histology carried out on skin infected by metacercariae did not reveal any pathological changes in the tissue around the parasites, as already observed by Paperna (1995). It was not possible to carry out histological exam on gill arch metacercariae for the difficulty in sampling the affected tissue. Anyway massive metacercarial infections have sometimes resulted in mortalities of young fish (Paperna, 1995).

Despite their limited pathogenic effect, *Clinostomum* spp. are of economic importance since heavily infected fish, particularly those with cutaneous infections, when marketed are often rejected by consumers (Kabunda & Sommerville, 1984). Furthermore some human cases caused by clinostomids have been reported, pointing out the potential zoonotic role of these parasites (Euzeby, 1984; Kakizoe *et al*., 2004).

The "black spots" in fishes are caused by metacercariae of at least six species of strigeoid digeneans among which *Neascus* sp., *Posthodiplostomum* sp. and *Bolbophorus* sp. have been frequently reported in African fish. The parasites are encysted generally in skin and muscle, but also in visceral districts and gills. Cysts consolidating around these metacercariae incorporate dermal melanophores and other chromophores with a resulting "black spot" appearance. Even if infestations of these parasites do relatively little damage to the fish, there is some evidence that heavily infected juvenile fish may suffer stress, weight loss and even death (Baker & Frank, 1985). Black spot metacercariae were found in all the aquatic environments with prevalence values rarely exceeding 10% and quite low mean intensities (3.1) in farmed fish. Only in a single case represented by one wild fish in Kenya 29 black spots were found. Histology allowed to observe just a mild atrophy in muscular tissue surrounding the parasite cyst, sometimes with focal hemorrhages.



Left: "black spots" in tilapia skin; middle: "black cyst" with an excisted metacercaria referable to *Bolbophorus* spp.; right: excisted *Bolbophorus* spp. metacercaria.
A sinistra: "black spots" nella cute di tilapia; in mezzo: "black cyst" con una metacercaria excistata riferibile a Bolbophorus *spp.; a destra: metacercaria di* Bolbophorus *spp. excistata.*

Furthermore unidentified digenean metacercariae were sporadically found in gills, liver, kidney, intestine and muscle of the examined fish.

All the above described digeneans recognize snails as first intermediate hosts. For this reason, the presence in the tilapia farming site of omnivorous fish, such as black carp and siluroid catfish, could reduce the snail population as they eat thin-shelled snails and their spawn. Furthermore the presence of piscivorous birds, definitive hosts of several digeneans parasitizing fish at the larval stages, should be avoided or reduced in the farming sites.

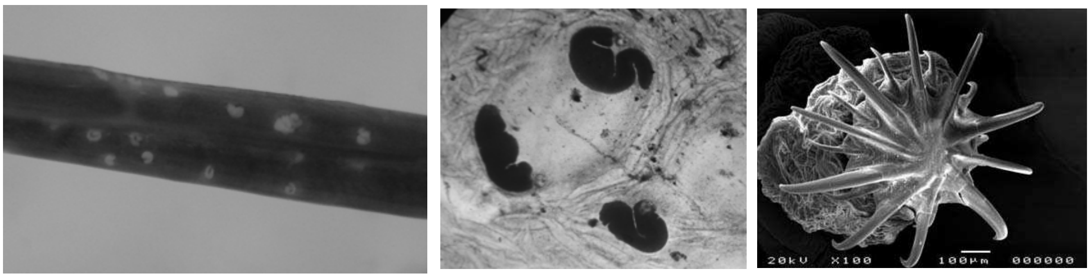
Finally several environmental factors may favor the colonization by intermediate hosts and the propagation of the infection.

Cestoda

Cestodes are endoparasites of vertebrates with over 5000 species so far described. Most of them require at least one intermediate host and complete their life cycle as adults in the definitive hosts. Two stages can be found in fish: adults in the intestine, when fish is the definitive host, and plerocercoid larvae in the viscera and musculature; the first larval stage (procercoids) is generally found in aquatic crustaceans (Woo, 1995). Numerous cestodes cause disease in fish (mainly at the plerocercoid larval stage) and in some cases they can be transmitted to humans, as in the case of *Diphyllobothrium* spp., causing a serious fish-borne zoonosis called Diphyllobothriasis.

A variety of adult and larval tapeworms (over 40 species) occur in native African fish; unsegmented forms, notably Caryophyllaeidae as well as one amphilinid representative, and the segmented pseudophyllideans and Proteocephalidae (Khalil, 1971; Van As & Basson, 1984). Although tapeworms are widespread in Africa there are only few records of tapeworms from cichlid fish, e.g. *Proteocephalus bivitellatus* (Paperna, 1996) and several gryporhinchyd cestodes (Scholz *et al*., 2004).

Amirthalingamia macracantha is the only species found during this survey both in tilapia and birds. The main characteristic feature of the species (and the monotypic genus as well) is the presence of 20 hooks of three sizes, arranged in two rows in a bilaterally symmetrical pattern (Bray, 1974). Adults have been found in *Phalacrocorax africanus* (type host) and *P. carbo* in Africa (Mali, Sudan) (Joyeux & Baer, 1935; Bray, 1974). Plerocerci were found by Aloo (2002) in the intestinal wall of *T. zillii*, previously designated as *Amirthalingamia* sp., then considered conspecific with *A. macracantha* (Scholz *et al*., 2004). Regarding the pathogenicity of *A. macracantha*, there were no data on the impact of this parasitic species in *O. niloticus*.



Left: intestinal wall of tilapia with several larval stages of cestodes; middle: three cestode larvae in the gut wall observed by light microscope; right: SEM detail of the armed scolex of *Amirthalingamia macracantha*.
A sinistra: parete intestinale di tilapia con numerosi stadi larvali di cestodi; in mezzo: tre larve di cestodi in preparato microscopico da intestino di tilapia; a destra: dettaglio dello scolice armato di Amirthalingamia macracantha *al SEM.*

In most cases heavily infected fish were found with many small plerocerci in the gut wall. Cestodes occurred in the three countries with quite low prevalence values, except for caged fish in Kenya (14.2%) probably for the different feeding, as they are fed on by the farmer and usually do not prey actively on zooplankton. Concerning infection intensities the positive fish showed heavy infestation with several larval stages well visible in the gut wall.

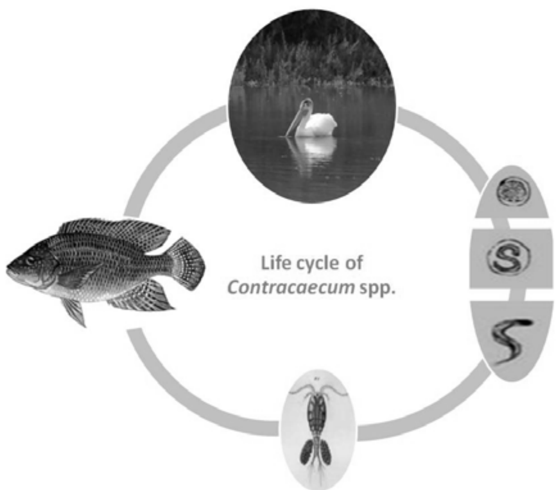
One interesting observation was the presence of cestode larvae frequently in association with Acanthocephala cystacanths, due to the presence of both parasites in small crustaceans (copepods and amphipods respectively) at the same level of food chain.

Histology showed numerous cysts in the intestinal wall containing parasitic larval stages of tapeworms with armed scolex; cyst had fibrous structure and contained red blood cells. The cysts were present in the submucosal and muscular layer, and were surrounded by epithelioid cells and sometimes by fibroblasts and lymphocytes. More often the cyst had jagged edges with cellular exfoliation and presence of red blood cells, may be for a mechanical erosion caused by the larvae. The wall around the parasite was hypertrophic for chronic inflammation. The cyst often protruded on the outer surface and on the lumen.

Nematodes

This "roundworms" infect many different species of farmed and wild fish. Small numbers of nematodes often occur in healthy fish, but high numbers cause illness or even death. In aquaculture systems, brood stock infected with a small number of nematodes may not even show signs of illness, but they often show a reduced reproductive capacity. On the other hand, juvenile fish infected by small numbers of nematodes are more likely to show signs of illness and also have reduced growth rates. In aquaculture, fish become infected with nematodes if they fed on live foods (mainly crustaceans) containing infective larval stages or if they are farmed in culture systems that promote the growth of other animals hosting infective stages of the nematode (vector or paratenic host, and intermediate hosts). Some nematodes can be transmitted directly from fish to fish, but the common life cycle pattern of fish nematodes is indirect, with at least one intermediate host. Adult nematodes are typically found in fish digestive tracts, depending from the species of nematode and the species of infected fish, adult and other life stages of nematodes can be found in almost any part of the fish, including the body cavity, internal organs, swim bladder, deeper layers of skin or fins, and external muscle layers (Yanong, 2006).

Some nematodes (Anisakidae) are characterized by a complex life cycle where carnivorous fish, piscivorous birds and aquatic mammals are definitive hosts, and crustaceans and fish first and second intermediate hosts respectively. Some Anisakid genera (*Anisakis*, *Pseudoterranova* and, at a less extent, *Contracaecum*) parasitizing fish at the larval stage can be accidentally transmitted to humans by eating infected raw fish and cause serious diseases. Among these genera, only *Contracaecum* larvae having piscivorous birds as definitive hosts occur in freshwater fish.



Contracaecum infestations have been described in fish from Israel (Paperna, 1964a), Egypt, most of the large and small East African lakes (Rift Valley), Zaire, Mali (Niger basin) (Khalil, 1971) and South Africa, where it was also reported from brackish water hosts (Boomker, 1982; Van As & Basson, 1983). Infections of the pericardia in cichlid fish occur in Israel (Landsberg, 1989) and in lakes Victoria, George, Nakuru, Naivasha, Baringo and Magadi (Paperna, 1974; Malvestuto & Ogambo-Ongoma, 1978). Larval nematodes occur either encysted in tissues or free in body cavities, most often in the abdominal or pericardial cavity. Larvae of *Contracaecum* and *Eustrongylides* tend to escape from their cysts, and crawl out of their host body after its death.

Definitive hosts of *Contracaecum* are pelicans, cormorants and herons. Pelicans (*Pelecanus onocrotalus*), incriminated as the definitive hosts of *Contracaecum* (found in the pericardial cavity of farmed tilapia, *Oreochromis* hybrids), were found to be infected by two species, *C. multipapillatum* and *C. micropapillatum*, but only the former appears to be implicated in infections of tilapia (Paperna, 1996). In South Africa, *C. spiculigerum* is found also in herons (Prudoe & Hussey, 1977).

Fish seem not to be severely affected by *Contracaecum* larvae neither encysted nor free, as inflammation, epithelioid formation and fibrous encapsulation around encysted larvae is generally localized. Multiple infection of the mesenteries resulting in extensive inflammation, fibrosis and even some visceral adhesions, were seen only in large fish, with no apparent impact on their body condition (Mbahinzireki, 1980). Worms inhabiting the pericardial cavity do not induce any visible damage despite localization. Large (200-350 g) tilapia can host up to 12 worms, which may reach a length of 6 cm and 2-3 mm in diameter. However, these infections, which affect the large fisheries of Lake Naivasha in Kenya and intensive tilapia pond cultures in Israel, cause significant economic losses, as larvae tend to migrate to the surface once fish die, making the fish not marketable.

Prevalence of pericardial *Contracaecum* infection among tilapia in a contaminated pond often reaches 100%, usually with 1-4 worms per fish. In Lake Naivasha, Kenya, 85% of *Tilapia leucosticta* were reported infected with a mean of 9 worms per fish, in Lake Baringo 70% of *O. niloticus* with 5 worms per fish, in Lake Magadi 30% of *T. grahami* with a mean of 2 worms per fish and in Lake George 30% of (270 mm long) *O. niloticus* with a mean of 1 worm per fish (Paperna, 1974; Malvestuto & Ogambo-Ongoma, 1978).

In this survey *Contracaecum* infections were observed mainly in wild tilapia from Ethiopia, with an overall prevalence of 17.3% and a mean intensity of 4.1 parasites/host. In the other countries both prevalence and intensity showed low values. The larvae were always found in pericardial cavity, as already reported by the above mentioned authors. The body condition of the infected fish seemed to be good, and the migration of larvae in muscle was never observed. The low prevalence observed in caged fish may be related to their different feeding behaviour as pelleted feed is given by the farmer and fish don't feed actively on zooplankton, where infective stages could be present. The prevention and control of the infection by *Contracaecum* spp. should be based on avoiding the presence of the host involved in the life cycle of the parasite. Prevention by keeping away piscivorous birds is impractical not only in natural habitats or reservoirs but even in fish ponds. Similarly the elimination of copepod crustaceans, first intermediate hosts, from the farming environment is not feasible. All the *Contracaecum* larvae found in the pericardial cavity of tilapia from Ethiopia were identified as *C. multipapillatum* on the basis of morphological study. Experimental trials have demonstrated that *Contracaecum*

Contracaecum spp. larvae in the pericardial cavity in tilapia.
Larve di Contracaecum *spp. nella cavità pericardica di tilapia.*

multipapillatum larvae can cause pathology in cats and rabbits (Vidal-Martinez *et al*., 1994; Barros *et al*., 2004), pointing out the zoonotic potential of this anisakid species.

Acanthocephalans

Also called "spiny" or "thorny-headed worms", these helminths belong to the separate distinct phylum Acanthocephala with about 1200 species divided into three classes. They are all intestinal parasites of vertebrates. Their major hosts are fish, amphibians, birds and mammals (Grabda, 1991). Morphologically they are cylindrical worms (from few mm to 70 cm long), with the anterior part provided with an eversible hooked proboscis, without digestive system (they absorb nutritive materials with the whole surface of the body). The acanthocephalans are dioecious, with males usually smaller than females, and oviparous. They exhibit an indirect life cycle with crustaceans (usually amphipods) as intermediate hosts. They infect host through the alimentary tract by ingestion of the infective larva via food. Several fish species can act as parathenic host.

More than 40 species of *Acanthosentis* are described throughout the world, 6 of which in Africa where the distribution of these species cannot be accounted for by the dispersal of their fish hosts, except for *A.* (*A.*) *tilapiae*. The other five species have regional distributions in geographically distant and unrelated parts of the continent: *A.* (*A.*) *malawiensis* in Malawi, *A. (A.) maroccanus* in Morocco, *A.* (*A.*) *nigeriensis* in Niger, *A.* (*A.*) *papilio* in Senegal and *A.* (*A.*) *phillipi* in South Africa. In contrast, *A.* (*A.*) *tilapiae* has been reported from 30 species of cichlid (28 of the genus *Tilapia*) and three non-cichlid species in Tanzania, Congo, Uganda, Chad, Nigeria, Egypt and Malawi (Amin & Hendrix, 1999). Much of this distribution is attributed to the dispersal of the ubiquitous cichlid and intermediate hosts via the waterways of the River Nile (Amin, 2005). Low to moderate infections with larval stages in visceral organs caused only local changes while heavy infections led to granuloma, fibrosis and atrophy of the affected portion (Paperna, 1996).

Results showed that this parasite has a tendency for a strong aggregation pattern. Aggregated parasite distributions are common among parasites (Rohde, 1993) and increase the chances of mating (Kennedy, 1976).

Acanthosentis spp. were present in Kenya (7.1%) and Uganda (13%) in all the systems considered with similar mean intensities ranging from 1.7 to 2.7 parasites/host. All the acanthocephalans found during this survey were identified as *Acanthosentis tilapiae*.

Acanthosentis spp. with everted proboscis.
Acanthosentis *spp. con proboscide estroflessa.*

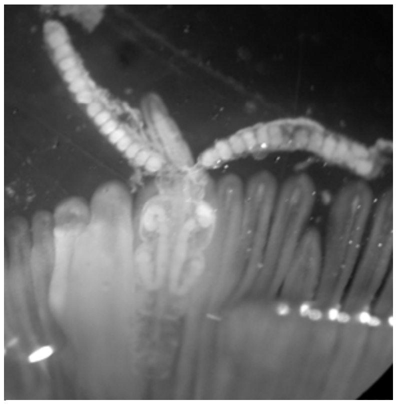
Histology did not show any relevant pathological sign in the host intestine, although in some case an eosinophilic granular cells infiltration in the submucosa was observed.

Crustaceans

Are a large group of arthropods, comprising almost 52000 described species, the majority of which are aquatic, living in either marine or freshwater environments. A few groups have adapted to life in terrestrial environments. Most of the crustaceans are motile, moving about independently, although a few taxonomic units are parasitic and live attached to their hosts (including sea lice, tongue worms, anchor worms, etc.). Crustacea parasitic on fish are numerous as species and abundant as individuals, showing strong structural modifications due to the adaptation to parasitism. Three major divisions of Crustacea have to be considered in fish parasitology: Branchiura, Copepoda and Isopoda, the latter found mainly in marine environment (Woo, 1995). The majority of the 2000 species that have been described so far as fish parasites belong to the class Copepoda. This group is very heterogeneous, showing different adaptations to various habitats and to the parasitic life; the range of forms extends from epizoic organisms and commensals to proper parasites.

The parasitic copepoda have a direct life cycle, generally shortened if compared to the free-living copepods, with several different larval stages and an adult stage. Copepods are dioecious, with males usually smaller than females. A female carries eggs in egg sacs, either single or paired, attached to the genital segment. The eggs hatches into a nauplius-type larva that will undergo several moults into different larval stages (it depends on the species). The crustacean parasites may harm the fish in three ways: they can cause pressure atrophy of soft tissues with their hard body, they can determine mechanical damage with their attachment structures and they can inflict damages of different degrees by feeding at the expenses of the host (some species are hematophagous).

Parasitic *Lamproglena* species are lernaeid copepods specialized for attachment on the gills and were observed in all the examined sites with similar prevalence values but higher mean intensity in Sagana ponds. Egg sacs were elongated and eggs organized in a string rather than in a clump as in *Lernaea*. This copepod frequently moves from one to another gill septum, leaving behind thickenings and mechanical grasping that prevent the circulation and blood supply, thus decreasing gill capacity, i.e. the exchange of oxygen with the surroundings. This leads to respiratory problems and reduces viability of the fish (Öktener *et al.*, 2008). Paperna (1996) suggested that high infestations by *Lamproglena clariae, L. intercedens* and *L. monodi* may interfere seriously with respiration of their host fishes.



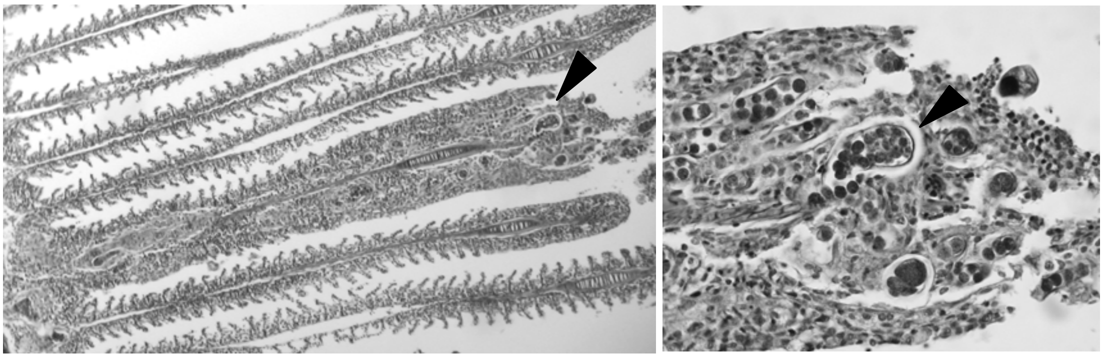
Copepod referable to *Lamproglena* spp. in tilapia gills.
Copepode riferibile a Lamproglena *spp. in branchie di tilapia.*

In our survey *Lamproglena* sp. was found in Kenya and Ethiopia in tilapias from all the aquatic systems taken into consideration, but always at low intensity (one parasite/fish in Ethiopia and 4.2 in Kenya). In the parasitized gills, the attachment area showed the presence of a thick layer of mucus, suggesting that the parasite can cause gill damage and hamper oxygen intake by heavy parasitized fish. In histology lesions such as mild to severe gill disease and epithelial sloughing off were observed, in association with the presence of the parasite, strongly attached to the gill lamellae.

Fungi

Branchiomycosis caused by *Branchiomyces* spp. was reported in various fish species, notably common carp, American catfish and eels. The only report in farmed tilapia is to date reported by Paperna & Smirnova (1997), detected in Israel in juveniles by histological examination. Infections are usually confined to the gills, colonizing gill vessels and capillaries. Infected areas become necrotic and brownish-grey (gill rot) for blockage, haemostasis and thromboses by the hyphae. Branchiomycosis occurs in eutrophic environments with lot of organic matter and temperature above 20°C; above 25°C infection may spread to most fish and cause heavy mortalities (Paperna, 1996).

Among the fish examined only one pond farmed tilapia from Kenya was positive for *Branchiomyces* spp., probably *B. sanguinis* for the lacking of expansion to extravascular system (Neish & Huges, 1980). At the histological examination the infection affect only few gill lamellae showing heavy proliferation of filaments and the presence of hyphae loaded with sporonts. Epithelial cells showed pycnosis and karyorrhexis. The observed lesions were similar to those described by Paperna & Smirnova (1997).



Left: gill filament affected by *Branchiomyces* spp. (arrow); right: detail of the affected filament with hyphae full of sporonts (arrow).
A sinistra: filamento branchiale colpito da Branchiomyces *spp. (freccia); a destra: particolare del filamento infetto con ife ripiene di sporonti (freccia).*

CONCLUSIVE CONSIDERATIONS

On the basis of the results presented, the parasitofauna did not show remarkable differences between wild and farmed (cages and earth-based ponds) fish, except for protozoan ectoparasites found mainly and with high infection intensities only in farmed tilapias.

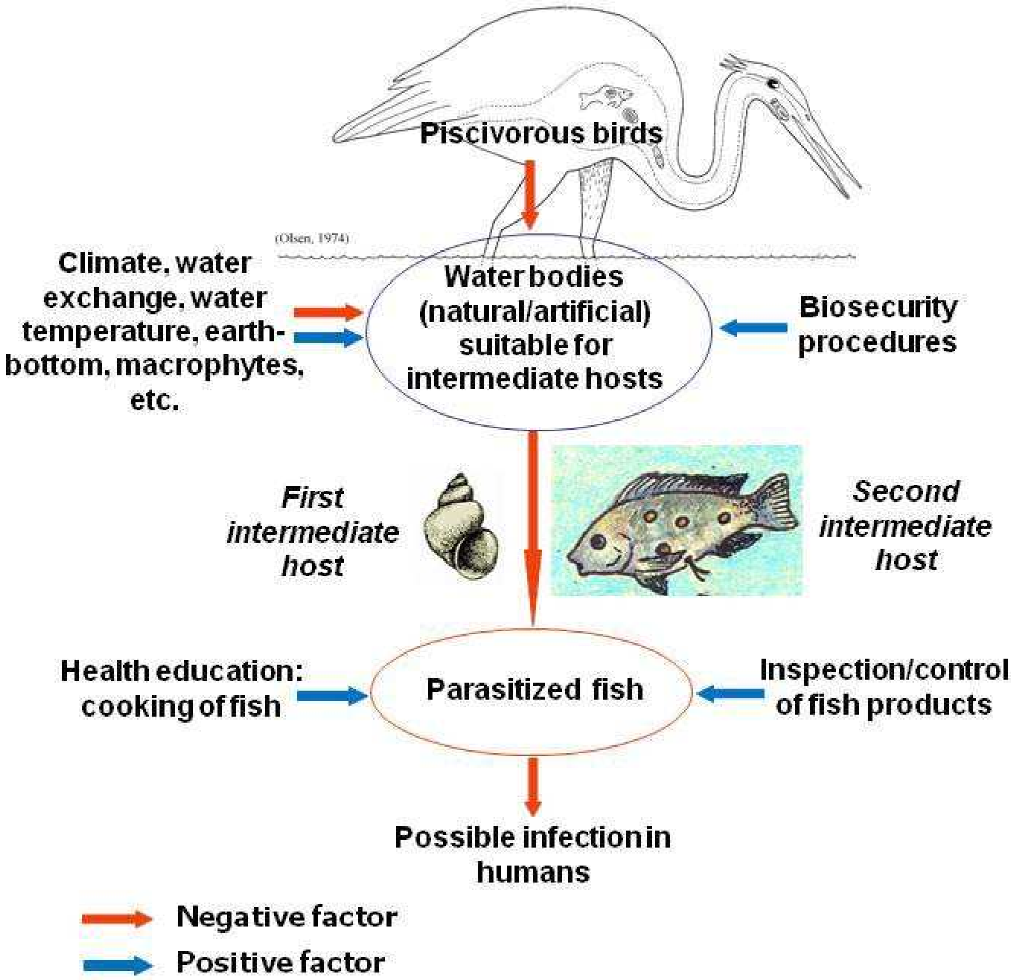
In a general view, parasitic infections due to protozoan ectoparasites are influenced by poor water parameters (pH, ammonia, low water exchange, high organic load, etc.) and husbandry conditions (overcrowding, etc.), requiring attention to these factors during environmental monitoring programmes. Biomass density will be an important factor to assess also with reference to monogenean (dactylogyroids and gyrodactylids) infections.

Concerning parasitic infections due to heteroxenous parasites, detected in all the sampling sites, biotic factors such as presence of invertebrates and piscivorous birds could represent important risk factors and should be evaluated and estimated for monitoring activities.

In general, the following risk factors and related preventive/control measures can be listed:

- Overcrowding and poor water quality parameters can predispose to parasitic infections due to protozoan and monogenean ectoparasites in hatchery and cage-farmed fish. *At this regard, the maintenance of good water parameters and low biomass density are important methods of prevention for this infections.*
- Invertebrates (gastropods, crustaceans, etc.) are often intermediate hosts of heteroxenous parasites (cestodes, nematodes, digeneans, acanthocephalans, etc.) and their presence is necessary for the transmission of these parasites to fish, mainly in natural water bodies but also in earth-based farming ponds. *At this regard, biosecurity actions aimed at reducing or avoiding the presence of these invertebrates (e.g. introducing a congruous number of siluroid fish) could be useful in limiting the colonization and spreading of some parasitic infections.*

- Piscivorous birds are definitive hosts of heteroxenous parasites (cestodes, nematodes, digeneans, etc.) harmful to fish, both farmed and wild; furthermore, they can spread several infectious pathogens among water bodies and within fish populations. *At this regard, their presence should be reduced as much possible in order to minimize the contamination of the farming systems (e.g. protecting by nets the farming area when feasible).*
- Only clinostomid digeneans and *Contracaecum* nematodes may represent a risk factor for humans when raw or slightly cooked parasitized fish are eaten. *At this regard the cooking of the fish easy inactivate the parasite, representing an useful individual control method. In the figure below the factors influencing in a positive/negative way the completion of the life cycle of clinostomid digeneans and their transmission to humans are schematized.*



Factors influencing in a positive/negative way the completion of the life cycle of clinostomid digeneans and their transmission to humans.
Fattori condizionanti, in senso positivo/negativo, il completamento del ciclo biologico di digenei clinostomidi e la loro trasmissione all'uomo.

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REFERENCES

Abolarin M.O. (1974). *Myxobolus tilapiae* sp. nov. (Protozoa: Myxosporidia) from three species of freshwater *Tilapia* in Nigeria. *J. West Afr. Sci. Assoc.*, 19: 106-114.

Aloo P.A. (2002). A comparative study of helminth parasites from the fish *Tilapia zillii* and *Oreochromis leucostictus* in Lake Naivasha and Oloidien Bay, Kenya. *J. Helminthol.,* 76: 95-102.

Amin O.M. (2005). Occurrence of the subgenus *Acanthosentis* Verma & Datta, 1929 (Acanthocephala: Quadrigyridae) in Japan, with the description of *Acanthogyrus* (*Acanthosentis*) *alternatspinus* n. sp. and *A.* (*A.*) *parareceptaclis* n. sp. from Lake Biwa drainage fishes and a key to the species of the subgenus. *Syst. Parasitol*., 60: 125-137.

Amin O.M. & Hendrix S.S. (1999). Acanthocephala of cichlids (Pisces) in Lake Malawi, Africa with a description of *Acanthogyrus (Acanthosentis) malawiensis* sp. n. (Quadrigyridae) from *Labeo cylindricus* Peters, 1852 (Cyprinidae). *J. Helm. Soc. Wash.*, 66: 47-55.

Baker J.R. (1963). Three species of *Myxosoma* (Protozoa: Myxosporidia) tilapias in Israel. *J. Protozool.*, 32, 1: 194-201.

Baker S.C. & Frank J.B. (1985). Effects of Black-Spot Disease on the condition of stonerollers *Campostoma anomalum*. *Am. Mid. Nat.*, 114, 1: 198-199.

Barros L.A., Tortelly R., Pinto R.M. & Gomes D.C. (2004). Effects of experimental infections with larvae of *Eustrongylides ignotus* Jäegerskiold, 1909 and *Contracaecum multipapillatum* (Drasche, 1882) Baylis, 1920 in rabbits. *Arq. Bras. Med. Vet. Zootec*., 56, 3: 325-332.

Boomker J. (1982). Parasites of South African freshwater fish. I. Some nematodes of the catfish [*Clarias gariepinus* (Burchell, 1822)] from the Hartbeespoort dam. *Onderstepoort J. Vet. Res.,* 49: 41-51.

Bray R.A. (1974). A new genus of dilepidid cestode in *Tilapia nilotica* (L., 1758) and *Phalacrocorax carbo* (L., 1758) in Sudan. *J. Nat. Hist*., 8: 589-596.

Bush A.O., Lafferty K.D., Lotz J.M. & Shostak A.W. (1997). Parasitology meets ecology on its own terms: Margolis *et al.* revisited. *J. Parasitol.*, 83: 575-583.

Coche A.D. & Pedini M. (1997). Establishment of the aquatic farming systems network for Africa. *FAN, FAO Aquacult. Newsl.* 15: 7-11.

Cribb T.H., Anderson G.R., Adlard R.D. & Bray R.A. (1998). A DNA-based demonstration of a three-host life-cycle for the Bivesiculidae (Platyhelminthes: Digenea). *Int. J. Parasitol.,* 28: 1791-1795.

Dawes B. (1956). The Trematoda. *Cambridge University Press, London*.

Donges J. (1974). The life cycle of *Euclinostomum heterostomum* (Rudolphi, 1809) (Trematoda: Clinostomatidae). *Int. J. Parasitol*., 4: 79-90.

Douëllou L. (1992). Parasites of *Oreochromis* (*Oreochromis*) *mortimeri* (Trewavas, 1966) and *Tilapia rendali rendali* (Boulanger, 1836) in Lake Kariba, Zimbabwe. *University of Zimbabwe Lake Kariba Research Station Bull., 2 (Proc. of seminar series)*: 14-31.

Douëllou L. (1993). Monogeneans of the genus *Cichlidogyrus* Paperna, 1960 (Dactylogyridae: Ancyrocephalinae) from cichlid fishes of Lake Kariba (Zimbabwe) with descriptions of five new species. *Sys. Parasitol*., 25: 159-186.

Dyková I. & Lom J. (1981). Fish coccidia: critical notes on life cycles, classification and pathogenicity. *J. Fish Dis.,* 4, 6: 487-505.

Dyková I. & Lom J. (1982). *Sphaerospora renicola* n. sp., a myxosporean from carp kidney, and its pathogenicity. *Z. Parasitenk*., 68: 259-268.

El-Mansy A. (2005). *Myxobolus heterosporus* Baker, 1963 (syn. *Myxosoma heterospora*) (Myxozoa: Myxosporea) in African records. *Dis. Aquat. Org*., 63: 205-214.

El-Mansy A. (2008). New exogenous stages of oocysts, sporocysts, and sporozoites of *Goussia cichlidarum* Landsberg and Paperna 1985 (Sporozoa: Coccidia) and impact of endogenous stages on the swim bladder of tilapias in Egypt. *Parasitol. Res*., 102, 2: 233-241.

El-Tantawy S.A.M. & Kazubski S.L. (1986). The trichodinid ciliates from fish, *Tilapia nilotica* from the Nile delta. *Acta Protozool*., 25: 439-444.

Ergens R. (1981). Nine species of the genus *Cichlidogyrus* Paperna, 1960 (Monogenea: Ancyrocephalinae) from Egyptian fishes. *Folia Parasitol*., 28: 205-214.

Euzeby J. (1984). Les parasitosis humaines d'origine animale: caractères épidémiologiques. *Flammarion Médecine-Sciences, Paris*.

Fahmy M.A.M., Mandour A.M. & El Naffar M.K. (1975). A survey of myxosporidia of freshwater (fish) collected from the river Nile at Assiut province. J. *Egypt. Soc. Parasitol*., 4/5: 93-102.

Fahmy M.A.M. & Selim M.K. (1959). Studies on some trematodes parasites of dogs in Egypt with special references to the role played by fish in their transmission. *Z. Parasitenk*., 19: 3-13.

Farstey V. (1986). *Centrocestus* sp. (Heterophyidae) and other trematode infections of the snail *Melanoides tuberculata* (Muller, 1774) and cichlid fish in Lake Kinneret. *MSc Thesis, Hebrew University of Jerusalem (Hebrew text, English summary).*

Ferdousi U.K. & Chandra K.J. (2002). New monogenean gill parasites of *Oreochromis niloticus* (Linnaeus) and *Oreochromis massambicus* (Peters) (Osteichthys, Cichlidae) from Mymensingh, Bangladesh. *Riv. Parassitol*., 19, 1: 49-60.

Fijan N. (1961). Massive invasion of carp fry (*Cyprinus carpio* L.) by protozoan of the genus *Glossatella* (Croatian text, English summary). *Vet. Arth. Zagreb*, 32: 30-33.

Finkelman S. (1988). Infections of Clinostomatidea in the Sea of Galilee fish. *MSc Thesis, Faculty of Agriculture, Hebrew University of Jerusalem (Hebrew text, English summary).*

Flores-Crespo J., Ibarra F., Flores-Crespo R. & Vasquez C.G. (1992). Variación estacional de *Dactylogyrus* sp. en dos localidades productoras de tilapia del Estado de Morelos. *Técnica Pecuaria en México*, 30, 2: 109-118.

Fomena A. & Bouix G. (1987). Contribution a l'etude des Myxosporidies des poisson d'eau douce du Cameroun. III. Especes nouvelle des genres *Henneguya* Thelohan, 1892 et *Thelohanellus* Kudo, 1933. *Rev. Zool. Africaine*, 101: 43-53.

Fomena A. & Bouix G. (1994). New Myxosporidea species (Myxozoa) from freshwater teleosts in Southern Cameroun (Central Africa). *Zool.*, 108: 481-491.

Fomena A. & Bouix G. (1997). Myxosporea (Protozoa: Myxozoa) of freshwater fishes in Africa: keys to genera and species. *Syst. Parasitol.*, 37: 161-178.

Fomena A., Bouix G. & Birgi E. (1985) Contribution à l'étude des Myxosporidies des poissons d'eau douce du Cameroun. II. Espèces nouvelles du genre *Myxobolus* Bütschli, 1882. *Bull. Inst. Fond. Afr. Noire, Série A*, 46: 167-192.

Fryer G. (1961). Observation on the biology of the cichlid fish *Tilapia variabilis* Boulenger in the northern waters of Lake Victoria (East Africa). *Rev. Zool. Bot. Afr.*, 64: 1-33.

Fryer G. & Iles T.D. (1972). The cichlid fishes of the great lakes of Africa. *T.F.H. Publications, Neptune City, N.J.*

Gbankoto A., Pampoulie C., Marques A. & Sakiti G.N. (2001). *Myxobolus dahomeyensis* infection in ovaries of tilapia species from Benin (West Africa). *J. Fish Biol.*, 58: 883-886.

Grabda J. (1991). Marine fish parasitology. An outline. *Warsaw. PVN, Polish Scientific Publishers.*

Harris P.D. & Cable J. (2000). *Gyrodactylus poeciliae* n. sp. and *G. milleri* n. sp. (Monogenea: Gyrodactylidae) from *Poecilia caucana* (Steindachner) in Venezuela. *Syst. Parasitol.*, 47: 79-85.

Hecht T. & Endemann F. (1998). The impact of parasites, infections and diseases on the development of aquaculture in sub-Saharan Africa. *J. Appl. Ichthyol.*, 14, 3/4: 213-221.

Hillis D.M. & Dixon M.T. (1991). Ribosomal DNA: molecular evolution and phylogenetic inference. *Q. Rev. Biol.*, 66, 4: 411-453.

Hoffman G.L. (1999). Parasites of North American freshwater fishes. *Cornell University Press, Ithaca USA*: 1-539.

Joyeux C. & Baer J.G. (1935). Notices helminthologiques. *Bull. Soc. Zool. Fr.*, 60: 482-501.

Kabata Z. (1970). Diseases of fishes: Book I. Crustacea as enemies of fishes. *New Jersey: T.F.H. Publications.*

Kabunda M.Y. & Sommerville C. (1984). Parasitic worms causing the rejection of tilapia (*Oreochromis* species) in Zaire. *Br. Vet. J.*, 140, 3: 263-268.

Kakizoe A., Washizaki M., Oda M., Kifune T. & Inokuchi A. (2004). A case of parasitic laryngitis caused by *Clinostomum complanatum*. *Larynx Japan*, 16: 52-55.

Kanev I., Radev V. & Fried B. (2002). Family Clinostomidae Lühe, 1901. *In: Gibson D.I., Jones A., Bray R.A. (Eds). Keys to the Trematoda, vol. 1. CABI Publishing, Wallingford, UK*: 113-120.

Kennedy C.R. (1976). Reproduction and dispersal. *In: Ecological aspects of parasitology (Ed. C.R. Kennedy). North Holland Publ. Co., Amsterdam*: 143-160.

Khalil L.F. (1969). Studies on the helminth parasites of freshwater fishes of the Sudan. *J. Zool. London*, 158: 143-170.

Khalil L.F. (1971). Checklist of the helminth parasites of African freshwater fishes. *Tech. Comm. 42, Comm. Inst. Helm., C.A.B. England*: 80 pp.

Kostoingue B. & Toguebaye B.S. (1994). Le genre *Myxobolus* (Myxozoa: Myxosporea) chez les poissons d'eau douce du Tchad avec la description de trois nouvelles especes. *Bull. Inst. Found. Afrique Noire*, 47: 63-71.

Landsberg J.H. (1986). Myxosporean parasites of the catfish, *Clarias lazera* (Valenciennes). *Syst. Parasitol.*, 9: 73-81.

Landsberg, J.H. (1989). Parasites and associated diseases of fish in warm water culture with special emphasis on intensification. *In: Shilo M. & Sarig S. (ed.) Fish culture in warm water systems: problems and trends. CRC Press Inc. Boca Raton, Flo.*: 195-252.

Lightner D., Redman R., Mahoney L., Dickerson G. & Fitzsimmons K. (1988). Major diseases encountered in controlled environment culture of tilapias in fresh water and brackish water over a three year period in Arizona. *In: Pullin R.S.V., Bhukaswan T., Tonguthai K. & Maclean J.L. (eds.) The second International Symposium on Tilapia in Aquaculture. ICLARM Conference Proceedings 15, Dep. Fish. Bangkok, Thailand, ICLARM Manila, Philippines*: 111-116.

Lo C.-F., Wang C.-H., Huber F. & Kou G.-H. (1982). The study of *Clinostomum complanatum* (Rudolphi, 1814) II. The life cycle of *Clinostomum complanatum*. *CAPD Fisheries Series (Taiwan) No. 8, Fish Disease Research (IV)*, 1: 26-56.

Lockyer A.E., Olson P.D., Ostergaard P., Rollinson D., Johnston D.A., Attwood S.W., Southgate V.R., Horak P., Snyder S.D., Le T.H., Agatsuma T., McManus D.P., Carmichael A.C., Naem S. & Littlewood D.T.J. (2003). The phylogeny of the Schistosomatidae based on three genes with emphasis on the interrelationships of *Schistosoma* Weinland, 1858. *Parasitology,* 126, 3: 203-224.

Lom J. (1980). *Cryptobia branchialis* Nie from fish gills: ultrastructural evidence of ectocommensal function. *J. Fish Dis.*, 3: 427-436.

Lom J. & Dyková I. (1992). Protozoan parasites of fishes. *Elsevier Science Publishers, Amsterdam.*

Lombard G.L. (1968). A survey of fish diseases and parasites encountered in Transvaal. *Newsletter Limnol. Soc. S. Afr.*, 11: 23-29.

Malvestuto S.P. & Ogambo-Ongoma A. (1978). Observation on the infection of *Tilapia leucosticta* (Pisces: Cichlidae) with *Contracaecum* (Nematoda: Heterocheilidae) in Lake Naivasha, Kenya. *J. Parasitol.*, 64: 383-384.

Manter H.W. & Pritchard M.H. (1969). Some digenetic trematodes of Central Africa chiefly from fishes. *Revue de Zoologie et de Botanique Africaines,* 80, 1-2: 51-61.

Mariaux J. (1998). A molecular phylogeny of the Eucestoda. *J. Parasitol.*, 84, 1: 114-124.

Matthews D. & Cribb T.H. (1998). Digenetic trematodes of the genus *Clinostomum* Leidy, 1856 (Digenea: Clinostomidae) from birds of Queensland, Australia, including *C. wilsoni* from *Egretta intermedia*. *Syst. Parasitol.*, 39: 199-208.

Mbahinzireki G.B. (1980). Observation on some common parasites of *Bagrus docmac* Forskahl (Pisces: Siluroidea) of Lake Victoria. *Hydrobiol.*, 75: 273-280.

Meyer F.P. (1970). Seasonal fluctuations in the incidence of diseases on fish farms. *Spec. Publ. Am. Fish. Soc.*, 5: 125-128.

Michel C. (1989). Pathology of tilapias. *Aquat. Living Resour.*, 2: 117-126.

Neish G.A. & Hughes G.C. (1980). Fungal diseases of fishes. *In: Disease of fishes, Book 6 (ed. by S.F. Snieszko & H.R. Axelrod). T.F.H., Neptune, N.J.*

Obiekezie' A.I. & Okaeme A.N. (1990). Myxosporea (Protozoa) infections of cultured tilapias in Nigeria. *J. Afr. Zool.*, 104: 77-91.

Obiakezie A.I. & Taege M. (1991). Mortality in hatchery reared fry of the African catfish *Clarias gariepinus* (Burchell) caused by *Gyrodactylus groschafti* Ergens, 1973. *Bull. Eur. Assoc. Fish Pathol.*, 11: 82-85.

Okaeme A.N., Obiekezie A.L., Lehman J. Antai E.E. & Madu C.T. (1988). Parasites and diseases of cultured fish of Lake Kainji area Nigeria. *J. Fish Biol.*, 32: 479-481.

Öktener A., Eğribaş E. & Başusta A. (2008). Preliminary investigation on serious mortalities of fish in Balıklıgöl (Halil-ür Rahman Gölü, Şanlıurfa). *G.U. J. Sc.*, 21, 1: 9-13.

Paperna I. (1964a). Parasitic helminths of inland-water fishes in Israel. *Israel J. Zool.*, 13: 1-26.

Paperna I. (1964b). The metazoan parasite fauna of Israel inland water fishes. *Bamidgeh* (*Bull. Fish Cult. Israel*), 16: 3-66.

Paperna I. (1968). Ectoparasitic infections of fish in Volta Lake, Ghana. *Bull. Widl. Dis. Assoc.*, 4: 135-137.

Paperna I. (1973). Occurrence of Cnidospora infections in freshwater fishes in Africa. *Bull Inst. Fond. Afr. Noir*, 35 (A-3): 509-521.

Paperna I. (1974). Larval *Contracaecum* in the pericardium of fishes from East African lakes. *Proc. Helminthol. Soc. Was.,* 41: 252.

Paperna I. (1991). Diseases caused by parasites in the aquaculture of warm water fish. *Annual Rev. Fish Dis.*, 1: 155-194.

Paperna I. (1995). Digenea (Phylum Platyhelminthes). *In: Woo P.T.K. (Ed). Fish diseases and disorders. Protozoan and Metazoan infections. Vol. 1, CABI International, Wallingford, U.K.*: 329-389.

Paperna I. (1996). Parasites, infections and diseases of fishes in Africa. An update. *FAO CIFA Technical Paper. 31*: 1-220.

Paperna I. & Lengy J. (1963). Notes on a new subspecies of *Bolbophorus confusus* (Krause, 1914) Dubois, 1935 (Trematoda, Diplostomatidae), a fish-transmitted bird parasite. *Israel J. Zool*., 12: 171-182.

Paperna I. & Smirnova M. (1997). *Branchiomyces*-like infection in a cultured tilapia (*Oreochromis* hybrid, Cichlidae). *Dis. Aquat. Org.*, 31: 233-238.

Paperna I. & Thurston J.P. (1968). Report on ectoparasitic infections of freshwater fish in Africa. *Bull. Off. Int. Epizoot*., 69: 1192-1206.

Paperna I. & Thurston J.P. (1969). Monogenetic trematodes collected from cichlid fish in Uganda, including the description of five new species of *Cichlidogyrus*. *Revue de Zool. Botan. Africain.*, 19: 15-33.

Paperna I., Van As J.G. & Basson L. (1984). Review of diseases affecting cultured cichlids. *In: Fishelson L. & Yaron Z. (ed.) International Symposium on Tilapia in Aquaculture, Proceedings. Tel Aviv University Press.*: 174-184.

Pariselle A. & Euzet L. (1995). Gill parasites of the genus *Cichlidogyrus* Paperna, 1960 (Monogenea, Ancyrocephalidae) from *Tilapia guineensis* (Bleeker, 1862), with descriptions of six new species. *Syst. Parasitol.*, 30: 187-198.

Pariselle A. & Euzet L. (2003). Four new species of *Cichlidogyrus* (Monogenea: Ancyrocephalidae), gill parasites of *Tilapia cabrae* (Teleostei: Cichlidae), with discussion of relative length of haptoral sclerites. *Folia Parasitol.*, 50: 195-201.

Prudhoe S. & Hussey C.G. (1977). Some parasitic worms in freshwater fishes and fish predators from Transvaal, South Africa. *Zoologica Africana*, 12: 113-147.

Roberts R.J. & Sommerville C. (1982). Diseases of tilapias. *In: Pullin R.S.V. & Lowe-McConnell R.H. (eds.) The biology and culture of Tilapias. ICLARM Conference Proceedings, 7, Manila, Philippines*: 247-263.

Rohde K. (1993). Ecology of marine parasites. 2nd edn. *Wallingford, CAB International.*

Sandoval-Gío J.J., Rodríguez-Canul R. & Vidal-Martínez V.M. (2008). Humoral antibody response of the Tilapia *Oreochromis niloticus* against *Cichlidogyrus* spp. (Monogenea). *J. Parasitol.*, 94, 2: 404-409.

Scholz T., Bray R.A., Kuchta R. & Řepová R. (2004). Larvae of gryporhynchid cestodes (Cyclophyllidea) from fish: a review. *Folia Parasitol.*, 51: 131-152.

Soleng A., Jansen P.A. & Bakke T.A. (1999). Transmission of the monogenean *Gyrodactylus salaris*. *Folia Parasitol.*, 46: 179-184.

Tarer V., Sakiti G.N., Le Breton A. & Marques A. (1996). Presence de *Myxidium leei* Myxosporidie pathogene chez les sparides en aquaculture en Mediterranée. *Ichtyophysiologica Acta*, 19: 127-139.

Thurston J.P. (1965). The pathogenicity of fish parasites in Uganda. *Proc. East African Academy*, 3: 45-51.

Ukoli F.M.A. (1966). On *Clinostomum tilapiae* n. sp. and *C. phalacrocoracis* Dubois, 1931 from Ghana, and a discussion on the systematics of the genus *Clinostomum* Leidy, 1856. *J. Helminthol.*, 40, 1-2: 187-214.

Van As J.G. & Basson L. (1984). Checklist of freshwater fish parasites from southern Africa. *S. Afr. J. Wildl.*, 14: 49-61.

Vidal-Martinez V.M., Osoriosarabia D. & Overstreet R.M. (1994). Experimental infection of *Contracaecum multipapillatum* (Nematoda: Anisakinae) from Mexico in the domestic cat. *J. Parasitol.,* 80: 576-579.

Williams M.O. & Chaytor D.E.B. (1966). Some helminth parasites of freshwater fish of Freetown Peninsula, Sierra Leone. *Bull. Inst. Fr. Afr. Noir, Ser. A*, 28: 563-575.

Woo P.T.K. (1987). *Cryptobia* and cryptobiosis in fishes. *Advan. Parasitol*., 26: 199-237.

Woo P.T.K. (1995). Fish diseases and disorders. Protozoan and Metazoan infections. Vol. 1. *CABI International, Wallingford, U.K.*

Yanong R.P.E. (2006). Nematode (Roundworm) infections in fish. *Circular 91, Department of Fisheries and Aquatic Sciences, Florida Cooperative Extension Service, Institute of Food and Agricultural Sciences, University of Florida.*

Yekutiel D. (1985). Metacercaria infections of cichlid fry in Lake Kinneret. *MSc Thesis, Hebrew University of Jerusalem (Hebrew text, English summary).*

Yimer E. & Enyew M. (2003). Parasites of fish at Lake Tana, Ethiopia. *Ethiopian J. Scien.,* 26, 1: 31-36.

Web site consulted:

http://www.gyrodb.net
http://www.fao.org/fishery/publications/sofia