Introduction

Freshwater fisheries has a significant contribution to development as an important source of proteins. More than 90% of all freshwater fisheries, i.e. wild capture and aquaculture, occurs in developing countries [1]. Besides providing food and a livelihood for millions of the world’s poorest people, freshwater fisheries contributes to the overall economic income by means of export commodity trade, tourism and recreation [2]. As a consequence, freshwater fisheries has become an important economic activity for both rural and urban populations in Africa and globally. The widening gap between supply and demand for fish products, has further made capture fisheries to be the largest extractive use of wildlife worldwide. Increases in human population, rising incomes and increasing urbanisation coupled with stagnation or decline of supplementary proteins, further exacerbates the situation. As a result most communities globally have responded by venturing into aquaculture to supplement capture fisheries. Despite its progress, African aquaculture needs to address a cascade of challenges including lack of national policies to guide aquaculture development, unfavourable investment policies, the absence of linkages between farmers, lack of research/technology development and extension, and unfavourable investment climates, inadequate quality seed and feed, and above all infectious and parasitic diseases [3,4].

Diplostomum species, especially larval stages namely cercariae and metacercariae are among the main agents of important diseases in fish and aquaculture systems. Diplostomum species are strigeoid digeneans of the family Diplostomidae [5,6]. The family Diplostomidae has four subfamilies: Diplostominae, Crassiphialinae, Alariinae and Codonocephalinae, which are classified on the basis of host specificity and metacercariae types [7]. However, the present review focuses on members of the compound genus Diplostomum [8], that includes three subgenera, often elevated to genera level [9]. They are Diplostomum, Tylodelphys and Dolichorchis. These parasites are ubiquitous in freshwater systems, but the most frequently encountered stages are the metacercariae that reside unencysted in the eyes, cranial and brain cavities of freshwater fish. Diplostomum can be highly pathogenic to fish and thus threaten the natural and aquaculture practices globally. Fingerlings in particular may experience high rates of mass mortalities when heavily infected with metacercariae, or as a result of massive cercarial penetration [5]. The metacercariae can also impair the fish escape response, diminish fish crypsis and thus may increase their vulnerability to predation [10]. In addition, cercarial penetration can increase the chances of bacterial infection in fish [11] and productivity of farms could be severely compromised.

Many studies on Diplostomum species have been reported in the Northern Hemisphere where their life cycles and
general biology have been well investigated. In Africa, on the other hand, a few studies are known and most of them have been restricted to reporting occurrence [12–15]. In general the taxonomy, biology and life cycles of Diplostomum species in Africa remains incomplete. With this regard, the present review focuses on Diplostomum species occurring in the African continent on the aspects of biology, taxonomy and life cycles based on the available literature. I highlighted areas where there is a dearth of information that require further research. This approach is a reflection of my personal view and not an attempt to be exhaustive.

**Life cycle and host specificity**

*Diplostomum* species, like other digenean trematodes, have attracted attention of countless studies due to their complex life-cycles, which involve a series of larval stages coupled with sexual and asexual modes of reproduction [16]. In the process they incorporate parasitic stages (sporocyst, metacercaria and adult) and free-living stages (egg, miracidium and cercaria). For a successful completion, diplostomid life cycle requires three hosts, namely, a mollusc intermediate host, a wide variety of fishes and amphibians as second intermediate host and a bird definitive host [6,10].

Sexually matured adults of *Diplostomum* species are found in the alimentary tract of bird host where they produce eggs that reach the external environment with the host’s faeces. Eggs hatch to release a ciliated free-swimming miracidium that seeks out a molluscan intermediate host. Snail hosts becomes infected after actively being penetrated by the miracidium. Once in the molluscan tissue, the miracidium transforms into a mother sporocyst, which produce asexually embryos that develop into daughter sporocysts generation. Moreover, the second intra-molluscan generations, daughter sporocysts, produce cercariae after another rounds of asexual reproduction [16]. Cercariae are well equipped with features and behaviours geared towards fish-finding, recognition and penetration [6]. After emergence, the cercariae actively infect a wide variety of fishes to become metacercariae. In the host, the metacercariae migrate along specific neural tracts to access sites with relatively minimal immune resistance like visceral organs, optic lobes and the brain so that they remain viable for successful transmission to the next host [16]. Bird hosts normally get infected upon consuming metacercariae in fish hosts [17].

Nonetheless, *Diplostomum* species life cycles have been well documented in the Palaearctic region, but not fully understood in Africa. Usually, lymnaeid snails (*Lymnaea* and *Radix* spp) and planorbid snails (*Planorbarius corneus*) are the main intermediate hosts [6,18], and in South America the snails of genus *Biomphalaria* such as *B. prona*, *B. straminea* and *B. glabrata* have been reported as the potential first intermediate [19]. In Africa, on the other hand, the range of snail hosts are far from completely understood, despite several experimental attempts undertaken [13,20]. The only known life cycle is that of *Tylolepis xenophis*, in which the freshwater snail *Bulinus tropicus* is the first intermediate host [21]. Beverley-Burton (1963) also tried to infect *Radix natalensis* (*Lymnaeidae*) with *Diplostomum* (*Tylolepis*) *mashonense*, but could not obtain cercariae. Even so, strigeoid cercariae purported to belong to the genus *Diplostomum* have been reported from *Biomphalaria* species from three fish farms in Kibos area within Kisumu, Kenya [22]. Similarly, at Mindu Dam in Tanzania, *Diplostomum*-like furcocercariae had been reported once from the snail *Biomphalaria pfeifferi* [23,24], but since then *B. pfeifferi* have not been spotted in the dam. Thus the consistently high prevalence of the metacercariae of *T. mashonense* in *C. gariepinus* at Mindu Dam [23,25], brought a suspicion that non-lymnaeid snail species could serve as snail hosts, particularly because lymnaeid snails have not been recorded within or around the dam. As such it was hypothesised that other snails besides lymnaeids and *Biomphalaria*, could be responsible for the transmission and *Bulinus* spp were shown to support that hypothesis [25], as shown in figure 1.

In *Diplostomum* species, host specificity is mostly restricted in the first intermediate host while less specific in the second intermediate host and final hosts. For instance in Europe and North America, metacercariae of *Diplostomum* species have been recorded from over 150 species of fish from families Percidae, Salmonidae, Coregonidae, Clupeidae, Gobiidae, to name a few [26,27] and a broad range of piscivorous bird species serving as definitive hosts [10]. Similarly in Africa, *Diplostomum* species have been found in almost every fish family i.e. Characidae, Centrarchidae, Cichlidae, Claridae, Cyprinidae, Hepsetidae, Salmonidae, Schilbeidae to mention but a few [28,29]. However, the catfish *Clarias gariepinus* (family Claridae) is the most examined fish [29] and references therein. Nonetheless the range of fish hosts in Africa is not well known as the level of *Diplostomum* studies in Africa is still at an infant stage.

As far as definitive hosts are concerned, in Africa adult *Diplostomum* have been reported from the Egyptian kite *Milvus migrans aegypticus*, the Egyptian moorhen *Gallinula chloropus chloropus* and the giant heron *Ardea goliath* in Egypt [20,30,31], Pel’s fishing owl *Scotopelia peli* in Ivory Coast [32], the grey heron *Ardea cinerea* in Zimbabwe and Tanzania [13,23,33] the
African Diptostomum species with similar features may follow a similar fate. This idea was supported by Zhokhov et al. [40] and Zhokhov [38] who considered Diplostomum tregenna as Dolichorchis tregenna. At the metacercarial stage the fundamental features that enable distinguishing genera or species are the shape of fore- and hindbodies, presence or lack of additional organs of attachment like pseudosuckers, the structure of the holdfast organ, structure of the reserve bladder, the shape and spread of the calcareous bodies [35]. However these features could only be used on T. mashonense, D. tregenna and other known diplostomid metacercariae, but not on the metacercaria of D. marahouense, which is hitherto unknown. As a result the metacercarial status of D. marahouense is not understood.

Various literatures regarding Diplostomum species described from Africa reveal a complicated taxonomic situation. In particular, views over the taxonomic position of the named Diplostomum species differ. Dubois [8] considered the genus Diplostomum to be under three subgenera, namely, Diplostomum, Tylodelphys and Dolichorchis (Figure 2). The idea which was supported by Niewiadomska [9], but elevated them to genera. Accordingly, the classification of the three forms was based on the anatomical features of the adults, i.e. the asymmetrical nature of the anterior testis (as in Diplostomum spp.) and the presence of a genital cone (as in Tylodelphys spp.). The genus Dolichorchis is given to materials exhibiting an intermediate position between Diplostomum and Tylodelphys, i.e. asymmetrical anterior testis and the presence of a genital cone. As a result, African material Diplostomum tregenna, D. marahouense and D. mashonense have been assigned within the genus Dolichorchis [9], although they originally belonged to the genus Diplostomum. However, phylogenetic analysis of Diplostomum mashonense by Chibwana et al. [41] suggested a re-allocation to Tylodelphys as previously viewed by Sudarić [42].

Furthermore, records of Tylodelphys species in Africa are generally scarce. The only descriptions of adults Tylodelphys species are those of immature T. clavata from the intestine of a jackal, buzzard Buteo rufofuscus in the Democratic Republic of southern Africa, Anhinga rufa rufa in Ghana [34] and the great white egret Ardea alba in Tanzania [23,33]. Generally, studies of Diplostomum species in bird definitive hosts in the African continent are scarce and limited. This is attributed to (i) low sampling efforts in the tropical countries due to inadequate expertise and resources (ii) difficulty in getting study permits to sacrifice some birds as they are either found in the protected areas (national parks and game reserves) or lack of interests in fish parasitology.

**Taxonomy of diplostomum (Sensu Dubois, 1970) species in africa**

Precise identification of members of the genus Diplostomum is usually difficult because of remarkable morphological similarity within and among species at almost every developmental stage [6]. Also, lack of a well-defined criterion further exacerbates the delineation difficulty within the Diplostomum group [6,35]. Furthermore, the taxonomic problem is aggravated by deformation of the body in the course of fixation, staining and mounting of permanent preparations [6]. In addition, many species have been described on the basis of one or two life cycle stages as a result different stages of the same species have been given different names or different species are known by the same name [6].

The taxonomy of Diplostomum species in Africa in general remains not well understood because the full range of species that may occur in Africa is far from completely known. As a consequence the biology of all reported species such as the range of hosts and life cycle stages (eggs, miracidia, intramolluscan stages, cercariae) is poorly understood. So far in the whole of Africa the adults of only five Diplostomum species have been described; they are D. tregenna [12], D. marahouense [32], D. magnicaudum [30], D. ghanaense [34] and D. ardeae [31]. On the other hand, only four Tylodelphys species adults are known i.e. T. mashonense [13], T. aegyptus [31], T. clavata [36] and T. xenopi [21]. Nevertheless, more than 15 Diplostomum species have been reported as metacercaria in different fish hosts species [29,37,38]. Although some authors tried to name the metacercarial stages to species level, for example D. garrae, D. longicollum, D. montanum and D. tilapia described by Zhokhov [38], have further exacerbated the taxonomic problem. Moreover the taxonomic status of some Diplostomum species that have been recorded from some fish hosts is confusing. For instance, when the genus Clarias was reviewed by [39], several widespread species i.e Clarias ngamensis, C. melandi and C. capensis of southern Africa, C. mossambicus of central Africa and C. lazera of west and north Africa were synonymized under the name Clarias gariepinus. With this regard, the taxonomic status of Diplostomum (Dolichorchis) tregenna recovered from C. lazera and Tylodelphys mashonense found in C. gariepinus is questionable.

In addition Niewiadomska [9] moved D. marahouense Baer 1957 to the genus Dolichorchis due to the presence of a genital cone and asymmetrical anterior testes in the adult form. This taxonomic re-evaluation of D. marahouense implies that other
of Congo (DRC) [36] and T. xenopi from an experimental host, the African darter Anhinga melanogaster in South Africa [21] and T. mashonense from Ardea cinerea and A. alba [13,33]. Other Tylodelphys species have only been described at the metacercarial stages. For instance Tylodelphys (Diplostomulum) victorius was described by Vercammen-Grandjean [43] from the pericardial cavity of Xenopus laevis victorianus in Nyakabere River in DRC. T. grandis was described from C. gariepinus by Zhokov et al. [40] from Lake Tana in Ethiopia. Four other Tylodelphys species, namely, T. claviformis (from Barbus, Labeobarbus, Garra), T. mutic (from Barbus), T. fusiformis (from Oreochromis niloticus) and T. clariae (from C. gariepinus) were also described by Zhokov [44] from Lake Tana in Ethiopia. However, there are several metacercariae of Tylodelphys species from numerous fishes in Africa (Table 1), which have not been described. In addition, the adults of these metacercariae and their life cycles are hitherto unknown, and their classifications only end at the genus level.

Table 1: A list of Diplostomum species (sensu Dubois, 1961) occurring in Africa with their hosts, developmental stage, site of occurrence and freshwater systems they were recovered.

| Parasite | Hosts | Stage        | Site              | Locality/Country | Reference                          |
|----------|-------|--------------|-------------------|------------------|------------------------------------|
| Diplostomum/Dolichorchis tregenna | Milvus migrans aegypticus | Adult | Small intestine | Nile River, Sudan; Osse River, Benin | Khalil, 1963; Okaka & Akhhigge, 1999 (from Lake Tana in Ethiopia) |
| Diplostomum/Dolichorchis tregenna | Clarias lazera/gariepinus Channa obscura | Metacercaria | Brain | Lamingo Dam, Nigeria | Osse River, Benin Lk Tana, Ethiopia (from Lake Tana in Ethiopia) |
| Diplostomum spathaceum | Clarias gariepinus | ? | Intestine/stomach | Lamingo Dam, Nigeria | Osse River, Benin Lk Tana, Ethiopia (from Lake Tana in Ethiopia) |
| Diplostomulium tugrense | Tilapia zilli | ? | Intestine | Lamingo Dam, Nigeria | Osse River, Benin Lk Tana, Ethiopia (from Lake Tana in Ethiopia) |
| Diplostomum sp | Scophetilia peli | Adult | Small intestine | Cote d'Ivoire | Baer 1957 (from Lake Tana in Ethiopia) |
| Diplostomum magnumcaudus | Gallinula chloropus | Adult | Small intestine | Egypt | El-Naffar, 1979 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Anhinga rufa rufa | Adult | Small intestine | Ghana | Ukoli, 1968 (from Lake Tana in Ethiopia) |
| Diplostomum ardea | Ardea goliath | Adult | Small intestine | Egypt | El-Naffar et al., 1980 (from Lake Tana in Ethiopia) |
| Diplostomum commutatum | Cynoglossus senegalensis | Adult | Small intestine | Cross River, Nigeria | Abraham et al., 2004 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Barbus poechii | Metacercaria | Eye | Lake Naivasha, Kenya | Otachi et al., 2015 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Brycinus lateralis | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Heptesodes odoe | Metacercaria | Eye | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Schilbe intermedius | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Oreochromis andersonii | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Oreochromis macrochir | Metacercaria | Eye | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Sargochromis Greenwoodi | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Serranochromis angusticeps | Metacercaria | Eye | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Coptodon rendalli | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Tilapia sparrmanii | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Labeo umbratus | Metacercaria | Eye | Okavango RS, South Africa | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Labeo capensis | Metacercaria | Eye | Okavango RS, South Africa | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Cyprinus carpio | Metacercaria | Eye | Okavango RS, South Africa | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Marcusius macrolepidotus | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Polimyrus castelnaui | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Pterocephalus castostoma | Metacercaria | Eye/brain | Okavango RS, Botswana | Grobelaar et al., 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Labeobarbus marequensis Barbus trimaculatus | Metacercaria | Eyes | Nwanedi-Luphepe dams, South Africa | Mboke et al. 2015 (from Lake Tana in Ethiopia) |
| Diplostomum garae | Garra dembecha | Metacercaria | Eye | Lake Tana, Ethiopia | Zhokkkov, 2014 (from Lake Tana in Ethiopia) |
| Diplostomum longicolium | Barbus humilis | Metacercaria | Eye | Lake Tana, Ethiopia | Zhokkkov, 2014 (from Lake Tana in Ethiopia) |
| Diplostomum montanum | Barbus humilis | Metacercaria | Eye | Lake Tana, Ethiopia | Zhokkkov, 2014 (from Lake Tana in Ethiopia) |
| Diplostomum tilapia | Oreochromis niloticus | Metacercaria | Eye | Lake Tana, Ethiopia | Zhokkkov, 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Tilapia sparrmanii, Pseudocrenilabrus philander | Metacercaria | Eye | Supersand Dam, South Africa | Moena et al., 2013 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Barbus intermedius Clarias gariepinus | Metacercaria | Eye | Koka reservoir, Ethiopia | Zhokkkov, 2014 (from Lake Tana in Ethiopia) |
| Diplostomum sp | Oreochromis niloticus | Metacercaria | Eye | Dams & farms, Eldoret, Kenya | Migiro et al., 2012 (from Lake Tana in Ethiopia) |
| Diplostomum spathaceum | Clarias gariepinus | Intestine/stomach | Lamingo Dam, Nigeria | Osse River, Benin Lk Tana, Ethiopia (from Lake Tana in Ethiopia) |

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Techniques and Methods

In order to have a reliable research output, researchers need a good criterion to filter from diversely available techniques and methods. For instance, Chappell [45], reported that methods for identification of Diplostomum species depend upon: (i) site of infection in a host; (ii) metacercarial morphology; (iii) infected host species; (iv) chaetotaxy of cercariae (not reliable unless supported by evidence obtained from experimental infections); (v) experimental establishment of the life cycle and recovery of the adult worm. Of the five methods above only three i.e. site of infection, infected host species and metacercarial morphology are the most common in African studies. In other words, most studies have been dealing with occurrences. Diplostomum metacercariae have been reported in the cranial cavity [13,14,46,47], eyes [29,48] and abdominal cavities [21]. Although other Diplostomum species recorded by some studies occur in unusual sites in fish host or unusual host, they are neither accompanied with morphometrics nor figures. For example metacercariae and adults of Diplostomum commutatum were isolated from the stomachs and intestines of fishes Pseudolitilthus elongatus and Cynoglossus senegalensis, respectively [49]. Generally most of studies on Diplostomum species in Africa are not accompanied with the known taxonomic methods i.e. morphometrics and illustrations. With this regard it is difficult for other researchers to support or refute conclusions.

According to Niewiadomska [7], a precise identification of Diplostomum species requires the a priori knowledge of all the features in the developmental cycle i.e. adult, cercaria and metacercaria. However, a correct description of cercariae and metacercariae relies on traditional and modern methods of morphological research like numerical taxonomy, as well as verification of the correctly identified adult forms obtained experimentally. So far, in Africa, identification of Diplostomum species across the life cycle has not been done due to incomplete information on the cercarial stage [13,20]. In addition, maintaining life cycles using in vivo systems is difficult, laborious and expensive and in vitro cultivation is almost impossible [6]. All these considerations indicate that both the identification of the larval stages and uncovering the diversity of Diplostomum species in Africa is challenging. Furthermore, experimental approaches cannot be applied to a large-scale screening of natural infections in intermediate hosts (fish or snails) due to the impossibility of linking each larval stage with its corresponding adult stage [25]. Although, the application of molecular techniques on Diplostomum has advanced significantly elsewhere [9,50–54], only a few researches in Africa have ventured into these techniques.

As polymerase chain reaction (PCR)-based methods for molecular analysis have started to advance in Africa, and a variety of molecular markers have been applied in different studies. Some of the gene regions that have been used to assess genetic diversity and variability among species and linking of life cycle stages are partial 18S rDNA sequences [23,55], 28S recombinant DNA [47], ITS rDNA (ITS1-5.8S-ITS2 [25,41,48] and the DNA barcode region of cytochrome c oxidase I (COI) [25,56]. T. mashonense is the most analysed diplostomid species in Africa, and almost every molecular marker mentioned above has been tested on it (see above citations). However, studies based on molecular methods on African Diplostomum are few in number, and the majority of them have used different markers, thus making it difficult to compare the findings taxonomically. For instance, it is not clearly understood if the T. mashonense studied by Chibwana et al. [25,41], is similar to that dealt by Moema et al. [47]. In other words, although DNA sequences of those workers’ materials have been deposited in the public nucleotide databases (like GenBank), they cannot be aligned together for identification purposes. As a result, more and more of the so-called Diplostomum species are discovered.
spatially and temporally, but their sequences cannot be used to
harmonise their identities. Increased synonyms in *Diplostomum*
species as shown by Niewiadomska [6] could be the ultimate
outcome.

**Future prospects**

Occurrence of *Diplostomum* species in their hosts across the
life cycle, i.e. in snails, fish or birds, is one of the first steps that
need to be performed for the subsequent analyses. However, a
correct identification of hosts is one of the most challenging
endeavour in Africa, which requires an involvement of experts
in specific fields viz. ornithologist, malacologist and fish biologist.
Understanding the biology of *Diplostomum* species and their
hosts would help fisheries managers and extension workers to
deal with the threat of the disease that might be posed by these
parasites. As a consequence, it will raise the economic returns
of fish farms as correct identification of species is critical in
making correct decisions for disease control. Such information
would also be invaluable for planning, implementation and
assessment of control strategies for these parasites in fish
farms. An experimental establishment of the life cycle of
diplostomid species is needed in order to study the biology of
the partially studied developmental stages like eggs, miracidia,
intramolluscan stages, cercariae and metacercariae. In other
words, studies on infectivity, pathology and migration through
the fish host of the parasite would only be possible if a life
cycle is maintained in the laboratory. Since *Diplostomum* species
can easily be cultivated in the laboratory [6], identification and
naming of species should be based on adult stages.

An alternative to completing the life cycle experimentally,
which is tedious and laborious, would be DNA sequencing.
Unfortunately, in Africa, DNA sequencing has rarely been
employed not only in the assessment of *Diplostomum*, but also
other animal groups. In cases where DNA sequencing was
applied, there has been little coordination (the sequences of
various studies cannot be compared because they targeted
exclusive DNA regions) of the four genes (i.e. 18S, 28S, ITS and
mtDNA) frequently used with great success in these trematode
studies. Consequently, the many sequences available on
public databases such as GenBank could not be assembled and
analysed together because they do not represent homologous
gene regions. To alleviate this difficulty, in future studies,
sequencing of all four genes would be an ideal solution in a quest
to identify more species. Alternatively, DNA barcoding efforts
for trematode species across Africa would be recommended.

DNA barcoding, the use of single locus cytochrome c
oxidase subunit 1 (COI) of mitochondrial DNA, has already
been popular in revealing illegal imports of bushmeats (Eaton
et al., 2009), labelling requirements violations in marketed
fish (Wong and Hanner, 2008; Lowenstein et al., 2009) and
accurate identification of potentially toxic tuna (Lowenstein et
al., 2010). The uniqueness of the locus COI across species is
purported to allow rapid and accurate identification of almost
all animal species (Hebert et al., 2003). Although this technique
has been widely adopted by other biological fields, the
parasitological community has barely used it. In parasitology
and *Diplostomum* in particular, DNA barcoding has been able
to reveal diversity and specificity of metacercariae in hosts
[52], disentangle cryptic species [53] and linking life cycle
developmental stages [25]. As already shown by Besansky et
al. (2003), DNA barcoding is potentially a great tool to improve
the rate of discovery of parasites’ species diversity and life
cycles. The author of this review, therefore recommends the
increased use of this genetic region in African diplodistomids
in order to fill the taxonomic gaps prevailing in *Diplostomum*
species emanating from various morphological challenges.
It will also enable quick identification of diplodistomid species
irrespective of their developmental stage once their sequences
are deposited in the public nucleotide databases.

**Conclusion**

Since the first *Diplostomum* species in Africa was described
in the intestines of Egyptian kite *Milvus migrans aegypticus*
in Sudan by Nazmi (1932), many other *Diplostomum* have been
found in other countries and hosts albeit most studies have
been conducted in the sub-Saharan Africa. However, a majority
of studies have been reporting their occurrences in fish either in
natural waters or fish farms. The influence of most *Diplostomum*
in the fish populations is not well understood. Although, interest
on *Diplostomum* studies has increased, most aspects like their
biology, epidemiology, taxonomy and immunology are well
known. The traditional approach to uncover these aspects would
be to complete the life cycles in the laboratory, which would
lead to morphological characterisation of all life cycle stages.
However, establishing and maintaining the life cycles of these
*Diplostomum* species using in vivo systems is difficult, laborious
and expensive. Although, molecular methods have been proven
to provide an alternative solution, are uncommon in African
studies due to lack of equipment, resources and expertise.
However, improvement of some weaknesses for some studies
like providing pictures or diagrams of the *Diplostomum* species
found. For laboratories or researchers that have the capacity
to do molecular analysis, the use of a molecular marker that
is commonly used like barcode region COI and ITS could be a
prospective development in future.

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