

**SPATIO-TEMPORAL INFECTION, DIVERSITY AND MORPHOLOGICAL
CHARACTERISATION OF PARASITE FAUNA OF *Xenopus* SPECIES IN
LAKE VICTORIA BASIN, KENYA**

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**A THESIS SUBMITTED TO THE BOARD OF POST-GRADUATE
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THE CONFERMENT OF THE DEGREE OF MASTER OF SCIENCE IN
LIMNOLOGY IN THE SCHOOL OF AGRICULTURE AND NATURAL
RESOURCE MANAGEMENT, KISII UNIVERSITY.**

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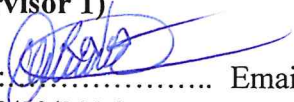
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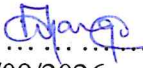
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DEDICATION

This work is lovingly dedicated to my dear parents, Julius Ogumbo and Catherine Onyoyo, whose unwavering love, sacrifices, guidance, and constant encouragement have been the foundation of my academic journey. Your belief in my potential, prayers, and tireless support gave me the strength to persevere through every challenge and to pursue my dreams with determination. This achievement is a reflection of your dedication and the values you instilled in me. May this work stand as a small token of my deepest gratitude and appreciation.

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ABSTRACT

Xenopus frogs (Pipidae) are aquatic anurans widely distributed across Africa, where they play important ecological, biomedical, and cultural roles. However, their populations are increasingly threatened by a range of stressors, including habitat degradation, climate change, and parasitic infections. This study investigated spatio-temporal infection and diversity indices of gastrointestinal helminths in *Xenopus borealis* and *Xenopus victorianus* from the Lake Victoria Basin, Kenya. A total of 480 individuals (240 per species) were collected between June 2024 and January 2025 from selected streams and ponds using baited traps and seine nets. Spatio-temporal variations in environmental conditions were recorded using a YSI multiparameter meter. Standard parasitological techniques were employed for the recovery, fixation, and identification of parasites. Four helminth genera were recorded: the intestinal cestode *Cephalochlamys* sp., and two nematodes, *Contracaecum* sp. (intestinal) and Camallanid spp. (stomach), Polyopisthocotyleans (*Polystoma* sp.) (urinary bladder) and Digenea spp (intestinal). Prevalence ranged from 23% to 90%, and mean intensity from 1.0 to 8.3, depending on parasite taxon and host species. Nematodes were the most prevalent and diverse group across all localities. Diversity metrics, including Shannon-Weiner, Margalef richness, Berger-Parker dominance, and Simpson's index, showed variation between hosts and sampling sites. Analysis of variance (ANOVA) revealed no significant spatial differences in infection indices ($p > 0.05$) except in *Polystomadid* sp in *X. victorianus* ($p < 0.05$), but temporal differences were significant ($p < 0.05$) for *Contracaecum* infections in *X. borealis* and camallanid spp., and *Polystoma* sp. in *X. victorianus*. These findings provide the first baseline data on helminth fauna from *Xenopus* species in Kenya and support the need for advanced morphological and molecular analyses to clarify taxonomy and better understand host parasite environment interactions

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LIST OF ABBREVIATIONS AND ACRONYMS

BS	Busia
GBIF	Global Biodiversity Information Facility
GPS	Global Positioning System
HDMS	Hexamethyldisilazane
ISERC	Institutional Scientific and Ethical Review Committee
IUCN	International Union for Conservation of Nature
KTRH	Kisii Teaching and Referral Hospital
LM	Light Microscopy
MDE	Minimum Detectable Effect
NACOSTI	National Commission for Science, Technology and Innovation
OIE	World Organization for Animal Health
OPD	Observed Prevalence Difference
PAST	Paleontological Statistics Software
SEM	Scanning Electron Microscopy
HB	Homa Bay
KI	Kisii
KK	Kakamega
LVB	Lake Victoria Basin
MG	Migori
SY	Siaya
VH	Vihiga

CHAPTER ONE

1.0 INTRODUCTION

1.1. Background information

Amphibian species, although they often go unrecognized, are globally important in many aspects, such as pest control, food, medicinal values, cultural values, scientific explorations, and indicators of changes in the environment. However, there are concerns about the declining global trends of amphibian species, with over 40% facing threats of extinction (Stuart et al., 2004). Partly, it is the nature of their sensitive bodies that makes amphibians among the most vulnerable to environmental changes, predation, and parasitic diseases. The order Anura is, by far, the most diverse group of amphibians with over 7000 recognized species and distributed in 49 families living in both aquatic and terrestrial habitats (Vitt & Caldwell, 2014; Zweifel & Cash, 2021).

The order has a worldwide distribution pattern, with 80% of the species living in tropical habitats (Zweifel & Cash, 2021). Anuran species are generally considered good environmental indicators of habitat quality and ecosystem status because of their highly permeable bodies that are sensitive to environmental variations (Hocking & Babbitt, 2014; Tagar et al., 2019). Hence, the diversity and geographical distribution patterns of anuran species are significantly influenced by not only the abiotic and biotic factors (Wells, 2007; Oda et al., 2016), but also the evolutionary processes that allow them to adapt to varying habitat conditions (Kabanze et al., 2023).

Of the 49 families, the Pipidae family comprises a group of the most primitive, but highly specialized species of frogs, which are largely aquatic and highly distributed in the

southern hemisphere. In this family, three species of the genus *Xenopus* have been reported in Kenya (Spawls et al., 2006). These include the Northern Clawed frog, *Xenopus borealis* Parker, 1936; Lake Victoria Clawed frog, *Xenopus victorianus* Ahl, 1937; and Muller's Clawed frog *Xenopus muelleri* Peters, 1844 these are widely distributed in water bodies found in the Western, Central, Eastern, and Coastal regions of Kenya. The latter is predominantly found in the coastal areas (Spawls et al., 2006).

The interaction between anurans and their parasites is complex. Parasitic infestations in anurans are as a result of ecological, biological, and environmental factors. These interactions are increasingly becoming the focus of herpetological and parasitological research due to their implications for conservation biology and ecosystem management (Schoeman et al., 2022). Anuran parasites range from ectoparasites to endoparasites. These parasites include nematodes, trematodes, cestodes and copepods. Each of these parasites affects its anuran hosts in unique ways, often causing debilitating diseases that can lead to significant declines in population density, alterations in community composition, and even species extinctions (Alroy, 2015). It can also lead to an adverse impact on ecosystem services, such as alteration of ecosystems and disturbance to aquatic food webs. Parasites infecting anurans are not only a concern for the conservation of amphibian communities, but also pose broader ecological implications for other organisms and humans as well.

Environmental factors, including habitat degradation, land-use change, water quality, and climatic conditions, can influence host-parasite interactions and parasite transmission dynamics. Evidence from East Africa further suggests that anthropogenic land use and eutrophication can shape parasite abundance, diversity, and community composition, demonstrating the importance of environmental conditions in determining parasite

dynamics (Hammoud et al., 2025). Moreover, host population density and genetic diversity play significant roles in parasite transmission and virulence. Studies on *X. laevis* in Europe revealed that denser populations experience higher parasitic loads due to increased contact rates and shared habitats (Hallinger et al., 2020).

Parasitic infections in amphibians are a critical aspect of biodiversity and ecological health. In South Africa it is reported that over 60% of *X. laevis* individuals harboured gastrointestinal helminths, indicating a high prevalence of parasites in similar species (Schoeman et al., 2020). Additionally, research on Anura species in Brazil showed parasitic prevalence rates ranging from 40% to 90% (Benício et al., 2022), demonstrating the widespread occurrence of parasitic infections in amphibians. With the world becoming a global village where cultural interactions are continually dynamic, there is a great potential shift in how humans and future human populations will interact with amphibians, hence, introducing a new dimension of socio-economic aspects of anurans. This will not only place additional pressure on anuran populations but may also contribute to the spread of parasites, both to and from anurans, affecting their viability and that of the ecosystems they inhabit.

With the foregoing background, this study aimed to explore the spatio-temporal infection, diversity indices, to morphologically characterize the parasitic fauna of the Northern Clawed Frog (*X. borealis*) and the Lake Victoria Clawed Frog (*X. victorinus*) within the Lake Victoria Basin of Kenya.

1.2. Statement of the problem

Amphibians play important ecological roles and are sensitive indicators of environmental change. However, their permeable skin and biphasic aquatic–terrestrial life cycle expose

them to a range of environmental stressors and parasitic infections that may affect their health, survival, and population dynamics. Environmental conditions, including variations in water quality and habitat characteristics, may further influence host–parasite interactions and infection patterns.

Despite the ecological importance of amphibians, information on parasite diversity, infection dynamics, and their relationship with environmental conditions remains limited in Eastern Africa. In Kenya, comprehensive studies on parasites infecting anurans are particularly scarce, despite the occurrence of amphibians in diverse aquatic habitats, including wetlands, streams, ponds, and aquaculture systems. Furthermore, the parasite fauna and infection patterns of *X. borealis* and *X. victorinus*, two widely distributed species in the Lake Victoria Basin, remain poorly documented. The lack of information on the diversity, spatial and temporal variation, and environmental determinants of parasitic infections in these species limits understanding of their host–parasite relationships and the ecological factors influencing infection dynamics.

1.3. Justification of the study

Amphibians play vital ecological roles, contributing to ecosystem stability and functioning. They regulate insect populations, control disease vectors, and provide food for predators, thereby influencing ecosystem dynamics. Furthermore, they hold cultural significance in many societies and contribute to scientific research, particularly in fields such as medicine and ecology (Alford et al., 2009). Amphibians have a biphasic lifestyle and serve as a vital link for energy and nutrient flow between terrestrial and aquatic ecosystems (Finlay & Vredenburg, 2007). Many species of amphibians have large populations and fast growth rates, thereby occupying key roles in aquatic food webs as

food sources and predators. Therefore, in various ecosystems, they help in shaping the functions and diversity of their prey communities. Furthermore, they serve as indicators of environmental health, with declines in amphibian populations signalling broader ecosystem disturbances (Stuart et al., 2004). As anurans remain an integral part of the local ecosystems, their global decline among the other amphibian communities has ignited significant concerns among conservationists, ecologists, and researchers because of the critical roles they play in maintaining ecological balances, serving as bioindicators of environmental health, and their position in the food web (Hocking & Babbitt, 2014).

Studying the welfare of anuran species in Kenya is important, not only for their conservation and management but also for understanding the potential disease-related risks that might be attributable to their interactions with cultured fish species, especially the carnivorous North African catfish *Clarias gariepinus* (Burchell, 1822). Moreover, by clearly understanding the interaction between parasitic infestation and host immunity in anurans, fascinating insights into disease resistance and susceptibility could be discerned and inform their conservation and management strategies (Oda et al., 2016).

1.4. Objectives

1.4.1 Main objective

The main objective of this study was to assess the spatio-temporal infection, diversity indices, and morphological characterisation of parasite fauna of *Xenopus* species in Lake Victoria Basin, Kenya.

1.4.2 Specific objectives

- i. To identify and characterise the parasite fauna of *X. borealis* and *X. victorinus* from Lake Victoria Basin morphologically.
- ii. To determine the spatio-temporal patterns of parasite and infection indices (prevalence and mean intensity) and diversity in *X. borealis* and *X. victorinus*.
- iii. To determine the relationship between spatio-temporal variation in water quality parameters and parasite infection and diversity indices in *X. borealis* and *X. victorinus* in the LVB.

1.5 Research questions

1. What types and morphological characteristics of parasite fauna infect *X. borealis* and *X. victorinus* within the Lake Victoria Basin, Kenya?
2. What are the spatio-temporal patterns of parasite infection indices (prevalence, mean intensity, and abundance) and diversity in *X. borealis* and *X. victorinus* from the Lake Victoria Basin, Kenya?
3. What is the relationship between spatio-temporal variation in water quality parameters and parasite infection and diversity indices in *X. borealis* and *X. victorinus* in the LVB?

CHAPTER TWO

2.0 LITERATURE REVIEW

2.1 Anuran classification

The order Anura comprises diverse and largely carnivorous, short-bodied tailless amphibians of the phylum Chordata with over 7,000 described species (Frost, 2021). Anurans, commonly known as frogs and toads, are distributed across diverse habitats worldwide, where they play pivotal roles in maintaining the health and functioning of wetland ecosystems (Frost, 2021). Anuran classification is typically based on morphological features such as skin texture, toe structure, and habitat preference (Fabrezi et al., 2014). Anura includes all modern frogs and any fossil species that fit within the definition. Characteristics of anuran adults include nine or fewer presacral vertebrae, the presence of urostyle formed of fused vertebrae, no tail, a long and forward sloping ilium, shorter forelimbs than hind limbs, fused tibia and fibula, fused ulna and radius, elongated ankle bones, and absence of prefrontal bone (Fabrezi et al., 2017). The anuran larva or tadpole has a single central respiratory spiracle and mouth parts that consist of keratinous beaks and denticles.

One of the most noticeable morphological features of anurans is their skin texture, which varies significantly between frogs and toads. Frogs typically possess smooth, moist skin, aiding in respiration through cutaneous gas exchange (Tattersall, 2007). This adaptation allows frogs to thrive in aquatic and semi-aquatic habitats. In contrast, toads exhibit rough, warty skin, which serves as a defence mechanism against predators (Pough et al., 2016). The presence of parotoid glands in toads secreting toxins further enhances their protection against predators (Vitt & Caldwell, 2014).

Another distinctive morphological feature in anurans is their toe structure, particularly evident in their adaptation to different locomotion modes. Frogs typically possess webbed toes, facilitating efficient swimming and jumping (Liem et al., 2001). The webbing between their toes increases surface area, enabling better propulsion in water. Toads in contrast have relatively shorter and less webbed toes, adapted for walking rather than swimming (Pough et al., 2016). This adaptation reflects the terrestrial lifestyle of toads, which predominantly inhabit drier environments compared to frogs.

The length of limbs is a morphological trait that varies between frogs and toads and is correlated with their habitat preferences and locomotion strategies. Frogs generally have longer hind limbs compared to toads, which contribute to their powerful jumping ability (Tattersall, 2007). This adaptation allows frogs to swiftly evade predators and efficiently capture prey. Toads, on the other hand, have relatively shorter hind limbs adapted for slow, terrestrial locomotion (Vitt & Caldwell, 2014). The shorter limbs of toads are better suited for walking and hopping short distances rather than extensive jumping.

2.2 Anuran distribution and habitat characteristics

They are widely distributed, ranging from the tropics to sub-arctic regions, but tropical rain forests have the greatest concentration of species diversity. Anurans live in varied habitats such as rain forests, rivers, streams, deserts, and alpine environments. Some live entirely in aquatic environments while others entirely on terrestrial habitats with direct development and are independent of a water bodies, relying only upon humid habitats for reproduction (Crump, 2015).

The diversity and distribution of anurans is influenced by regional and local factors such as seasonal distribution of rainfall, moisture, and temperature (Schivo et al., 2019).

Regions with marked seasonality have more species diversity than the non-seasonal areas. Rainfall and the availability of water bodies with high atmospheric humidity are important factors that influence anuran reproduction and distribution. Species with wide distributions in strongly seasonal areas mainly have aquatic ovipositions and are less dependent on humid forests than those that lay their eggs in terrestrial habitats (Buxton & Sperry, 2016).

Anuran species composition often varies among habitats. Lakes and forest habitats have the greatest anuran diversity. Species that survive in forested areas are often influenced by size and degree of isolation, but frequency and intensity of human disturbance and the nature of the surrounding vegetation may play a major role as well (Nessi et al., 2023).

Kenya has a great diversity of anuran species. Some of these species are threatened by ecological disturbances and human influences. Despite this influence, some species have adapted to the changing environmental conditions. Reed frogs, found in marshes, reed beds and other wet areas, tree frogs found in forests especially in the eastern region of Kenya, grass frogs found in grasslands, savannas, and other open areas, are just a few of the many frog species found in Kenya. Their distribution may vary depending on factors such as habitat loss, climate change and other environmental factors.

Xenopus borealis, is primarily found in Kenya and Tanzania, especially in high-altitude areas. This species favours a range of freshwater habitats such as lakes, rivers, and marshes, often at higher elevations, where it occupies both natural and anthropogenically modified water bodies (Frost, 2024). In contrast, *X. victorinus* has a more extensive distribution across East Africa, including Uganda, Rwanda, Burundi, and Tanzania. This species is highly adaptable, often inhabiting eutrophic or standing water, which includes

temporary bodies of water that arise from seasonal rains. *Xenopus victorinus* tends to avoid larger rivers with predatory fish, favouring isolated, less competitive aquatic environments that allow it to thrive in various water bodies throughout the region (Channing & Howell, 2006).

The distribution dynamics of *Xenopus* species are similarly influenced by environmental conditions. Lake Victoria Basin is a dynamic ecosystem, with the existence of diverse wetlands and rivers. The lake basin receives a lot of rainfall, and is often hot and humid all of which supports the reproductive success of anurans as most of them prefer wet and humid climates (Koskei, 2023). The presence of abundant water bodies and diverse wetland systems and forests around the lake offers ideal conditions and provide high humic compounds that *Xenopus* species often prefer in their habitats. Increased nutrient loading from agricultural activities leads to eutrophication, which degrades water quality and creates unfavourable conditions for amphibians while fostering parasite proliferation. Additionally, fluctuating water levels, resulting from climate change and altered rainfall patterns, can affect breeding sites and disrupt the reproductive cycles of Anura species (Koskei, 2023).

According to recent data from the Global Biodiversity Information Facility (GBIF, 2024), *X. borealis* is primarily distributed in the highland areas surrounding Lake Victoria, while *X. victorinus* is more widespread across the lowland regions near the lake's shores. These species thrive in freshwater habitats, but their distribution is threatened by habitat degradation, pollution, and changes in water availability due to human activities.

2.3 Importance of anurans

Frogs and toads are voracious predators of various invertebrates, including insects,

spiders, and worms, thereby helping to control their populations. Conversely, they serve as prey for a wide range of predators, including birds, mammals, and reptiles, contributing to the transfer of energy and nutrients within the food web (Ubiratã Ferreira Souza et al., 2023). Anurans are also sensitive to changes in environmental conditions, making them valuable indicators of wetland health (Blaustein & Kiesecker, 2002). Their population sizes, reproductive success, and overall health can reflect the quality of their habitat, including water quality, pollution levels, and habitat degradation. Through their feeding activities and excretion, they contribute to the decomposition of organic matter and the recycling of nutrients, which are essential for the growth of aquatic plants and algae (Hocking & Babbitt, 2014).

Anuran species richness and diversity contribute significantly to the overall biodiversity of wetland ecosystems (Kabanze et al., 2023). Different species occupy various ecological niches, and their interactions with other organisms help maintain ecosystem resilience and stability. Some anuran species aid in seed dispersal by ingesting seeds and transporting them to new locations through their digestive systems (Willson & Dorcas, 2003). This process facilitates the regeneration of vegetation in wetland habitats and contributes to plant diversity.

The decline of anurans can signal deteriorating environmental conditions such as habitat loss, pollution, climate change, and emerging diseases (Stuart et al., 2004). As amphibians are highly dependent on both terrestrial and aquatic ecosystems, any disruptions in these habitats can have profound effects on their populations. The International Union for Conservation of Nature (IUCN) reported that approximately one-third of anuran species are facing extinction (IUCN Red List, 2014). This alarming statistic highlights the severity of the threats facing amphibians worldwide. Furthermore, according to Wells

(2007), around 122 anuran species are believed to have become extinct since 1980, indicating a rapid loss of biodiversity within a relatively short timeframe.

Several factors contribute to the decline of anurans including parasites and parasitic diseases, chemical contaminants from agriculture, industry, and urban runoff, degrading water quality and impacting the health and survival of amphibians (Blaustein & Kiesecker, 2002). Also, altered precipitation patterns, increased temperatures, and habitat shifts affect the breeding, hibernation, and foraging behaviours of anurans (Parmesan & Yohe, 2003). Furthermore, the destruction of natural habitats due to urbanisation, agriculture, logging, and infrastructure development fragments populations and reduces available breeding sites (Russell & Downs, 2012). Parasites and parasitic diseases affect fecundity, growth, and development and they can also lead to mortalities in severe cases (Preston & Johnson, 2010).

Parasitic infections, such as those caused by helminths and protozoa, are common among amphibians and can lead to detrimental effects on host health. The pathophysiological impacts of parasitic infections, such as those caused by *Rhabdias* species, include lung inflammation and respiratory distress, which can severely impact amphibian populations. The presence of pulmonary nematodes in anurans can interfere with oxygen exchange processes at the diffusion barrier and impair the host's ability to maintain blood oxygenation under conditions of vigorous exercise (Moretti et al., 2017). Some gastrointestinal parasites such as nematodes and trematodes can affect energy balance and reduce the hosts aerobic capacity in turn interfering with processes like calling effort. This can limit the host's chances of reproductive success (Madelaire et al., 2013).

2.4 Description of common anuran parasites

2.4.1 Helminths

Helminths largely include flukes, tapeworms and roundworms. They have a well-defined body with well-developed organ systems such as the reproductive organs. A large proportion of helminths is known to be very active feeders. Flatworms (flukes and tapeworms), from their name, have a flattened body covered by a plasma membrane, which is not the case with roundworms, which are covered by a tough cuticle with their body shape being cylindrical (Williams & Jones, 1994; Cone, 1995; Cribb et al., 2002). With diverse life cycles, helminths are often assessed in terms of how they affect the health of their host and are often evaluated in the ecological aspect. Some of them bear zoonotic potential, and examples include *Spirometra* sp.; *Gnathostoma* sp., *Diphyllbothrium* sp., *Alaria* sp., *Echinostoma* sp. and pentastomids (Pentastomida), most of which are food borne diseases (Hallinger *et al.*, 2020).

2.4.2 Digeneans

They belong to trematodes which constitute about 9000 described species which include orders Digenea and Aspidogastrea. These parasites affect both warm- and cold-blooded animals. Digeneans have a complex life cycle with at least one intermediate host with the adult stage parasitising vertebrates. Their larval stage development is mostly in molluscs (Paperna & Dzikowski, 2006). The intestinal *Hoplobatrachus* sp., *Sclerophrys* sp. and *Ribeiroia* sp. parasites cause significant damage in their anuran hosts (Soubeiga et al., 2020).

Morphologically, they comprise an alimentary canal; mouth, pharynx, oesophagus and

two blind caeca; a ventral and anterior sucker which are used for locomotion and attachment and hermaphroditic reproductive system. With a complex life cycle, the digenean morphological differences exist throughout the stages, especially in the adult and larval stages (Woo, 1995).

2.4.3 Nematodes

Nematodes are characterised by their elongated cylindrical body shape with bilateral symmetry. Nematodes are not segmented; their body composition consists of a complete digestive system; the anterior consists of the oesophagus, the intestine or mid-section, and the posterior with the far end being that of the anus/cloacae (Jadwiga, 1991). Nematodes have over 80000 described species, with over 15000 parasitic species. Because of their diversity, these parasites affect plants and animals, including anurans (O'Rourke & Rosenbaum, 2015). The site of infection is through the alimentary canal with a lifecycle that involves a definitive host and more than one intermediate host. The parasitic nematode has got four larval stages before arriving at maturity.

The body cavity is filled with perivisceral fluid. Male nematodes are generally smaller in body size than their female counterparts. They are oviparous with few species that occur or are viviparous. Because of their lifecycle, nematodes move from one intermediate host to another till maturity and thus in their larval stages can be transmitted to humans by ingesting raw or poorly prepared infected anurans which in turn can cause serious infections (Hallinger et al., 2020).

2.4.4 Cestodes

These are endoparasites that require at least one intermediate host. Their life cycle is

mostly complete in the adult stage specifically in the definitive host. There are over 5000 described species. The cestodes are known to have two life cycle stages whereby adults are found to inhabit the intestines, plerocercoid larvae are found in the viscera and musculature, and proceroids are noted to be in aquatic crustaceans (Woo, 1995).

According to Woo (1995), morphological characteristics of adult cestodes are distinctive with a strongly flattened dorsoventral, descriptions of the body is inclusive of the scolex (head), neck, and strobila (body), the body generally made up of proglottids. The scolex is an organ used to steadily attach the parasite to the host's intestinal mucosa, so it is provided with structures such as suckers and bothria, hooks and bothridia. Cestodes lack an intestine, they gain nourishment through absorption by the tegument covering the surface of the body. Few cestodes are known to be hermaphroditic, whereby each proglottid has its own set of gonads both male and female. The site of infection is majorly in the intestine where Otachi (2009) observed an aggregation of plerocercoids of cestodes in the intestinal wall which was a similar discovery to that of Jadwiga (1991).

2.4.5 Acanthocephalans

Phylum Acanthocephala consists of intestinal parasites which are spiny-headed or thorny-headed with about 1200 described species containing classes Archiacanthocephala, Eoacanthocephala and Palaeacanthocephala. The site of infection is in the intestine and thus is often referred to as vertebrate intestinal parasites. They have diverse host species ranging from birds, mammals, and amphibians to fish. Their morphological appearance is characterised by presence of proboscis at the anterior end they don't have a digestive tract and thus absorb nutrients through their body surface.

They are cylindrical ranging from a few mm to almost 70 cm in length (Jadwiga, 1991). Comparisons between male and female acanthocephalans are considered to be dioecious with males being smaller in size than the females and oviparous. They have an indirect life cycle having crustaceans as their intermediate hosts (Otachi, 2009). Transmission is through ingestion of infected larvae as the larvae stages are encysted in the body cavity and reach maturity when ingested by a definitive host.

Adult worms have been reported in the intestinal tract of anurans in Brazil, Tanzania, China and Madagascar, in the anurans studied in Nigeria, only cystacanths have been found attached to organs of the viscera, including the gastro-intestinal tract, the liver and the urinary bladder (Aisien et al., 2021). The Pathogenic effects of acanthocephalans are distinctive to damage caused, and damage is caused by the proboscis in the intestinal wall comparable to the infection intensity (Mahmoud et al., 2006).

2.4.5 Monopisthocotyleans

Monopisthocotyleans are flatworms belonging to the class Monopisthocotyla (formerly known as Monogenea) within the phylum Platyhelminthes. They exhibit a simple body plan characterised by a posterior attachment organ known as the haptor, equipped with hooks, clamps, or both, facilitating their attachment to the host's skin or gills. This unique morphology enables them to thrive as ectoparasites, exploiting the resources provided by their amphibian hosts (Bullard et al., 2003).

The life cycle of Monopisthocotyla typically involves direct transmission between hosts, with free-swimming larvae known as oncomiracidia seeking out suitable hosts for colonization. Upon attachment to the host's integument, the larvae undergo

metamorphosis, developing into adult worms capable of reproduction. Monopisthocotyleans exhibit high host specificity, often showing a preference for particular anatomical sites, such as the skin or gills, where they can efficiently feed and reproduce (Whittington & Horton, 1996).

While monopisthocotylean infestations are generally considered non-lethal to anuran hosts, they can still have significant impacts on host fitness and health. Heavy parasite burdens may lead to skin lesions, impaired respiration, and increased susceptibility to secondary infections, ultimately affecting the survival and reproductive success of infected individuals. Furthermore, in cases of anthropogenic disturbance or environmental stress, parasite-mediated diseases can exacerbate population declines and threaten species conservation (Schloegel et al., 2009).

2.4.7 Polyopisthocotyleans

The majority of polyopisthocotylean parasitize the gills of fish, the Polystomatidae family is unique for infecting freshwater tetrapods, including anuran frogs for example, *Polystoma integerrimum* found in the common frog's bladder a rare freshwater tetrapod niche and they are known as blood-feeders (haematophagous), using a specialized buccal cavity and pharynx to ingest host blood from the bladder wall (Mollaret et al., 1997; Justine, 1998). They also possess a posterior opisthaptor with multiple suckers (usually six) and anchors or hooks to secure themselves to the bladder lining (Theunissen et al., 2014). This group has synchronized life cycle for example *P. integerrimum*, reproduction is tightly synchronized with the frog host's breeding season. Eggs are released into the host's urine when the frog enters water to spawn (Tinsley, 1995). They also exhibit developmental plasticity in some species (e.g., *Polystoma gallieni*): if a tadpole is infected

early, the parasite matures rapidly in the tadpole's gills or bladder; if infected after metamorphosis, it develops slowly in the adult frog's bladder (Badets et al., 2013). They have high host specificity: Each polystomatid species typically infects a single or closely related frog species, reflecting long co-evolutionary history (Tinsley, 1995).

2.4.8 Hirudinea

Leeches are segmented worms belonging to the class Hirudinea, characterised by their flattened bodies, suckers for attachment, and blood-feeding habits. While some leech species are free-living, many are ectoparasites that feed on the blood of vertebrates, including mammals, birds, and amphibians. Within anuran populations, hirudinean leeches are commonly found attached to the skin of individuals (Attademo et al., 2012), particularly in moist habitats such as marshes, ponds, and streams.

They rely on anurans as hosts for blood meals, which provide essential nutrients for their survival and reproduction. Anurans may experience negative effects due to leech infestations, including blood loss, tissue damage, and potential transmission of pathogens. However, the extent of these effects can vary depending on factors such as leech species, host immunity, and environmental conditions (Da Silva et al., 2018).

2.4.9 General effects of anuran parasites

Parasites have severe effects on the host they are parasitizing. These effects can affect the growth rate of anurans, lead to behavioural changes and reproductive characteristics, morbidities and survival of frogs and can also lead to adaptive alterations to the host in extreme cases. Such changes amplified could affect multiple populations, communities, and the ecosystem at large. These effects can be seen physically when parasites are plenty

in a host. Physical signs include extra limbs or digits, missing limbs, or misshapen limbs that are twisted. Infested frogs do not swim jump or forage as well as their normal counterparts and have reduced survival rates in natural populations. To treat, control and prevent infections and re-infections, these parasites must be extensively known.

2.4.10 Parasite prevalence survey and sample sizes

Parasitic prevalence estimates and corresponding sample sizes have been a subject of discussion and a challenge, particularly in host populations of unknown number (Jovan & Tella, 2006). The same authors observe that the threshold for establishing the minimum sample size is quite variable and subjective to researchers' decisions, in which sample sizes from three to eight hosts are considered minimum, 15 to 20 are considered moderate while, 30 to 75 are high. It has been shown that a sample size of about 15 host individuals could be applied as a reasonable trade-off between loss of data from analyses and maintaining the acceptable thresholds of uncertainty, usually about a third of the sampling prevalence (Jovan & Tella, 2006).

Parasitological and epidemiological monitoring of aquatic systems estimating an appropriate sample size for a large, indefinite population of aquatic organisms is always a complex challenge. Therefore, generic sample sizes that range from 30 to 150 host individuals are universally acceptable and frequently applied (Corsin et al., 2009; de Blas et al., 2020). These sample sizes have been confirmed to maintain confidence levels of between 95% and 99%, as outlined by the World Organization for Animal Health (OIE, 2018). The sample ranges were originally calculated using the formula by Cannon and Roe (1982) as follows:

$$n = (1 - \alpha^{1/d}) \cdot (N - \frac{d-1}{2})$$

where:

n is the required sample size

α is the probability (confidence level) of observing at least one infected animal in sample when the disease affects at least d/N in population

d = number of diseased animals in population

N = population size

By using different models, de Blas et al. (2020) was able to confirm that generic sample sizes calculated by the above formula were valid for the detection of parasite infections in aquatic systems where the exact numbers of host individuals are unknown. Consequently, the OIE recommends these generic sample sizes to detect infected aquatic organisms. These values are common in epidemiological studies where up to 150 host individuals are considered a desirable sample size for detection of infection over 2%; 60 individuals for infection over 5%; and 30 individuals for infection over 10%, and are derived directly from the formula. Another study (Shvydka et al., 2017) used analytic and simulation approaches to determine the optimal sample sizes in epidemiological and parasitological surveys for various fish species. Their study concluded that sample sizes with above 25 host individuals yield minimal statistical biases, but those below 10 individuals result in unreliable estimates. The study further found that for parasite datasets with highly aggregated dispersion patterns, sample size of 80 host individuals or more are desirable for statistical accuracy and reliability.

However, because most parasitological and epidemiological surveys often entail sacrificing of the host individual, ethical conventions recommend the use of low (but reliable) sample sizes for studies on threatened, endangered or species of conservation

interests (Shvydka et al., 2017). Nevertheless, the minimum sample sizes required in aquatic parasitological surveys tend to depend largely on the actual status of parasite aggregation in the host population (Wilson et al., 2002; Shvydka et al., 2018). Therefore, the infection index (prevalence) is as well as the desired confidence limits are the key determinant of the minimum sample sizes for parasitological and epidemiological studies (Cannon & Roe, 1982; de Blas et al., 2020).

2.4.11 Sampling power analysis and methods

Accurate estimation of parasite prevalence (proportion of hosts infected) depends strongly on the number of host individuals examined. Smaller sample sizes produce wider confidence intervals and increase uncertainty in prevalence estimates, potentially leading to misleading conclusions about parasite distribution or host–parasite dynamics. This relationship between sample size and prevalence precision is well-documented in parasitology (Sarabeev et al., 2025). It has been documented that proper sample size selection in parasitological surveys is important because it directly affects the accuracy of the results, where insufficient sampling leads to unacceptable precision and large uncertainty in the estimates of infection indices (Sarabeev et al., 2025). A classic parasitology literature (Gregory & Blackburn, 1991) emphasizes that parasite infection indices estimated based on small or poorly designed samples can fail to reflect true host–parasite relationships, simply because prevalence and true infection parameters can vary systematically with sample size.

Accordingly, sampling power analysis plays a crucial role in avoiding scenarios of underpowered sample sizes, thereby improving the scientific validity of results. This is defined as the probability of detecting a true effect if at all one exists. Without adequate

statistical power, studies are prone to type II errors (failing to detect real differences or relationships) and may produce misleading or non-replicable results (Sadiq et al., 2025). The principles of testing the statistical power of a study directly apply to parasitological and ecological studies where effect sizes and variability must be anticipated. Recent methodological reviews in parasite research discuss how sample size and power relate to study design, cost, logistics, and ethical research. These reinforce the fact that underpowered sampling schemes undermine the ability to detect meaningful patterns (Mayor et al., 2023; Biggs et al., 2024).

One of the methods used for study design and power analysis is the Minimum Detectable Effect (MDE), a complementary concept commonly used in study planning that defines the smallest difference in a parameter (e.g., prevalence between two host populations or time points) a study can be able to detect with specified power and confidence. The MDE reflects the smallest effect size (such as a difference in proportions) that a statistical test can reliably distinguish, given the chosen sample sizes and error thresholds; but when baseline prevalence or parameter values are unknown, these must be estimated or assumed from pilot data or related literature (Woodward, 1992; Mair et al., 2020).

Minimum Detectable Effect is mathematically related to sample size and variance, meaning that detecting smaller effects requires larger sample sizes to maintain acceptable power. Explicitly specifying an MDE during design ensures that the research is targeted toward biologically meaningful differences rather than statistically trivial ones, and anchors the sampling plan in the reality of field logistics and statistical sensitivity under varying spatial and temporal conditions.

CHAPTER THREE

3.0 MATERIALS AND METHODS

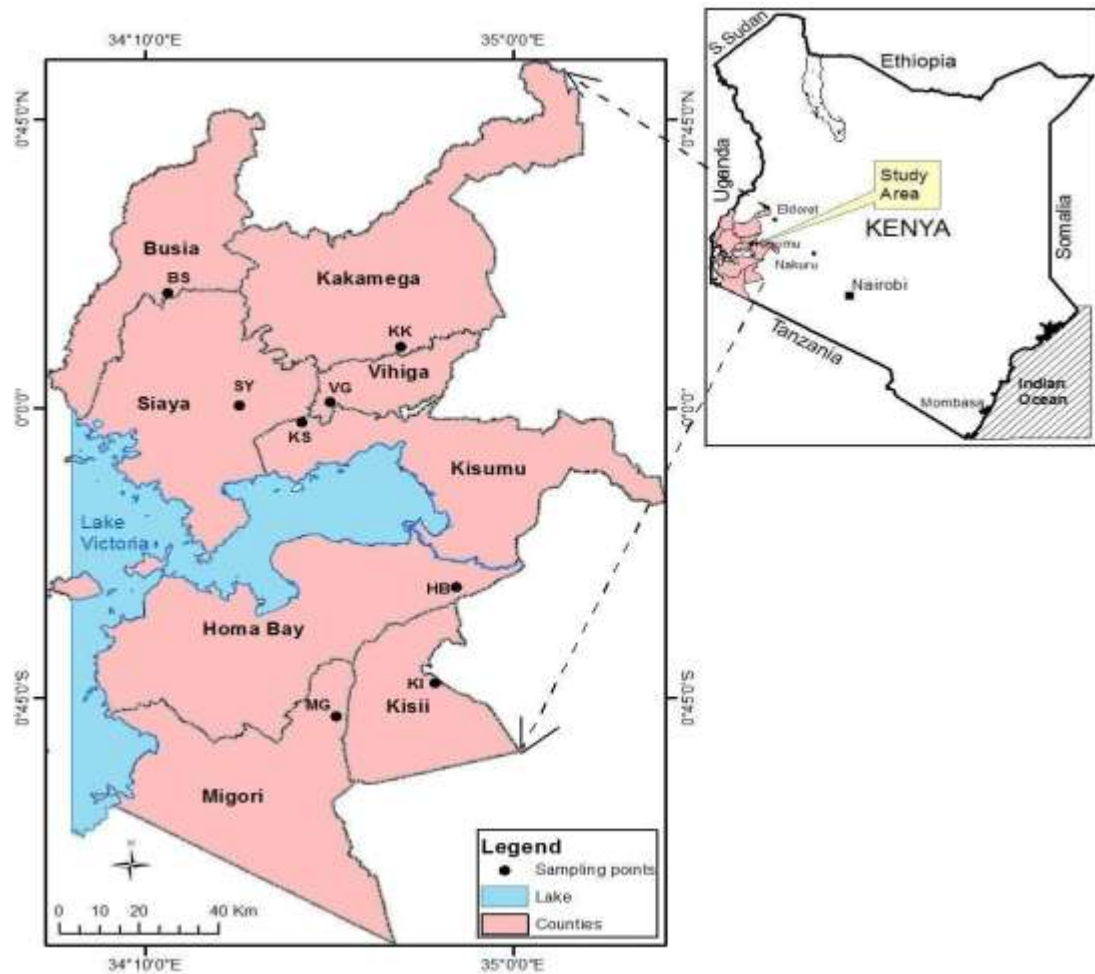
3.1 Study area

This study was conducted in eight counties in the western part of Kenya. The counties comprising Kakamega, Vihiga, Busia, Siaya, Kisumu, Homabay, Kisii, and Migori (Fig. 3.1), are part of the rich riparian ecosystem of Lake Victoria Basin with diverse flora and fauna. The sites were selected because of their historical documentation as harbouring the target species.

Past sightings of the target species in this area not only establish their presence but also confirm their potential habitats, which aided in the systematic collection of data. The proximity of these counties to Lake Victoria was also an important consideration as a hub for amphibian life by providing essential breeding and nursery habitats. It also aligns with the habitat preference of many anuran species and will help us understand the ecological dynamics of these species. The rich biodiversity in western Kenya, highlighted by the presence of Kakamega forest, will offer a wide setting for the study. As a distinct ecosystem, it will house a wide variety of frogs and their potential parasites, presenting an opportunity to explore a multitude of interactions in a single location.

Figure 3.1

Map of Kenya showing the eight Lake Victoria Basin counties and study site locations (generated in ArcGIS, Esri).



3.2 Sampling design

Baseline information on the prevalence of anuran parasites in the proposed study area is generally absent. Therefore, sampling for this research was preceded by a reconnaissance survey conducted in the eight counties (Fig.3.1) to explore and identify water bodies (ponds and wetlands) that might be inhabited by *Xenopus* species. This initial identification of the sites was done by sighting or capture-observe and release of the target anurans. All non-target frogs were released immediately after their capture and identification. *Xenopus* species were identified using their morphological features such

as size, skin texture, and presence of lateral lines (Du Preez & Carruthers, 2009). Sites inhabited by the target anurans were marked, using GPS coordinates, for subsequent definitive parasitological surveys. With each county treated as a unique geographical region, one sampling point was identified from each county, resulting in a total of eight (8) sampling points across the Lake Victoria basin.

Definitive sampling at each of the delineated points was conducted in three sessions comprising the dry (January-March), wet (May-June), and moderate (September - December) weather seasons. Live samples of anurans were sampled using baited traps and seine nets. Any non-target species of anurans caught were released upon immediate identification. The collected specimens were initially examined on-site for ectoparasites before being transported to either Kisii University Biology Laboratory or Kenya Marine and fisheries Research Institute depending on proximity to sampling point ,in aerated tanks containing water from their respective sampling sites. The tanks were covered with fine-mesh nets to prevent the frogs from escaping during transportation.

Water quality parameters were measured at each sampling site during every sampling session using a YSI multiparameter water-quality meter. Measurements were taken directly in the water at the sampling points and recorded for subsequent analysis. Water depth at each sampling site was measured using a handheld sonar depth finder.

3.3 Optimization of host sample size

Estimation of the actual numbers of the targeted species in an infinite population context was a challenge. Since the study sites comprised relatively small ponds and wetlands, a conservative sampling strategy was adopted. Specifically, 30 frogs per site were targeted and distributed evenly across three sampling periods (10 individuals per period). These sample sizes align with the thresholds suggested by Jovani and Tella (2006), who noted that 3–8 hosts may be considered minimal, 15–20 moderate, and 30–75 relatively high, with 15 individuals often serving as a reasonable compromise. This methodological approach, therefore, balanced practicality with statistical rigor while remaining within widely accepted ranges for epidemiological studies. Sample sizes between 30 and 150 individuals are generally sufficient to maintain statistical confidence levels of 95–99% (Corsin et al., 2009; de Blas et al., 2020), and are recognized by the World Organization for Animal Health (de Blas et al., 2020). These thresholds can be derived from the formula proposed by Cannon and Roe (1982), described as follows:

$$n = (1 - \alpha^{1/d}) \cdot (N - \frac{d-1}{2})$$

.....(i)

where: n = required sample size,

α = probability (confidence level) of observing at least one infected animal
in a sample when the disease affects at least d/N in a population,

d = number of diseased animals in production, and

N = population size.

A test run of the Cannon and Roe (1982) formula was first conducted with an assumption

of 1000 hosts as the target population sample frame in the identified ponds and wetlands, at 95% confidence interval ($\alpha = 0.05$). Accordingly, the desired sample sizes for the different levels of infection (prevalence) were respectively calculated (Table 3.1) proving that the targeted sample sizes of 30 host species per sampling site and across the sampling period were optimal for detection of parasite prevalence of as low as 10%.

Table 3. 1

*Test run of the desired sample size of *Xenopus* species, at different levels of prevalence using a hypothetical population size of 1000 anuran per sampling site (formula after Cannon and Roe, 1982).*

No. infected (d)	Parasite prevalence (%)	Desired samples size
100	10	28
200	20	13
300	30	8
400	40	6
500	50	4
600	60	3
700	70	3
800	80	2
900	90	2
1000	100	2
Average		7

3.4 Sampling power analysis

Because baseline data on parasite prevalence were not available before fieldwork, a post-hoc minimum detectable effect (MDE) analysis was conducted to retrospectively assess the study's ability to detect differences in infection metrics among counties. For the two *Xenopus* species, MDEs were calculated for each of four parasite taxa by pairwise

comparison between the counties with the least and highest recorded prevalence. The calculation was done using fixed sample sizes of 30 host specimens and pooled prevalence that were based on the number of hosts infected, assuming a two-tailed significance level (α) of 0.05 and a power of 80%. In this case, the MDE would represent the smallest absolute difference in prevalence among counties that the study was sufficiently powered to detect as statistically significant. Calculations were based on the standard formula for the difference between two proportions (Fleiss et al., 2003), where the pooled prevalence and respective sample sizes were incorporated as follows:

$$\bar{p} = \frac{x_1 + x_2}{n_1 + n_2} \dots\dots\dots (ii)$$

where x_1 and x_2 are the numbers of host infected, n_1 and n_2 the corresponding sample sizes.

Standard error of the difference was calculated as:

$$SE_p = \sqrt{\bar{p}(1 - \bar{p}) \left(\frac{1}{n_1} + \frac{1}{n_2} \right)} \dots\dots\dots (iii)$$

From the above, minimum detectable effect (absolute difference, two-tailed) was calculated as:

$$MDE_p = (Z_{1-\alpha/2} + Z_{Power}) \times SE_p \dots\dots\dots (iv)$$

where $Z_{1-\alpha/2}$ is the critical Z for two-tailed test, and Z_{power} being the normal quantile for the chosen power (0.84 for 80%) in this study.

These calculations were conducted in Microsoft Excel using a customised post-hoc power analysis template developed for multi-site compilation of parasitological field data.

3.5 Sampling power analysis

For this study, the inclusion criteria were the site being either natural or semi natural ponds and wetlands found within the Lake Victoria basin. Exclusion from the study were all the frogs that did not confirm to the description of *X. victorinus* and *X. borealis*. These were released at the site of collection and only the target species retained

3.6 Sampling limitations

Among the limitations of this sampling would be the possible absence of either *X. victorinus* and *X. borealis* in the same habitat. To mitigate against this, alternative sites identified and delineated, were sampled. Another limitation would be the timing for optimal collection of the desired samples. This was mitigated by repeated trapping and hauling of the sites both in the morning and evenings until the sample size was obtained.

3.7 Sample host examinations for parasites

Live anurans sampled in the field were transported in a tank with water from their site of capture to the laboratory at Kisii University (Plate 3.1).

Plate 3.1

Host samples dissections and laboratory examinations at Kisii University Biology Laboratory



In the laboratory, the sex of each frog was determined based on secondary sexual traits. Cloacal folds of females are usually more swollen than the males and the male's palms often darken during breeding season. Females are often larger than males. In cases where external identification was difficult further examination of the reproductive organs was done after dissection to accurately determine the sex. All the anuran specimens were humanely killed by severing the spinal cord key organs including the gastrointestinal tract, liver, lungs, and musculature as described by Duellman and Trueb (1994) were

examined for parasites.

Using parasitological examination procedures, the anuran intestines were examined for the presence of endoparasites. The observed parasites were collected, quantified and fixed in 70% ethanol for morphological analysis. Relevant permits and ethical clearance were obtained from NACOSTI (appendix 2, 3), and KTRH ISERC (appendix 4) before commencement of field work.

3.8 Morphological analyses

3.8.1 Light microscopy (LM)

In the laboratory the parasites were extracted and observed under a stereo light microscope using a Leica EZ4 stereomicroscope (Leica Microsystems GmbH, Wetzlar, Germany). Photomicrographs were taken using an Elikliv EDM11S Electronic Digital Microscope 2000X Biological Microscope 7 Inch IPS LED For Lab (Model Number: Elikliv EDM11S). Morphological identification of parasites photomicrographs was done using various sets of identification keys as provided in Table 3.2.

Table 3. 2

Different categories of expected parasites in target host species and the corresponding references of their identification keys.

Parasite group	Reference(s) of identification keys
Nematodes	Moravec (2019)
Cestodes	Scholz et al. (2021)
Trematodes	Scholz & Horák (2003)
Polystomes	Du Preez et al. (2025)
Acanthocephalans	Amin et al. (1987); Amin (2013); Tkach et al. (2013)
Hirudineans	Ahmed et al. (2015); Phillips et al. (2020)

3.8.2 Scanning electron microscopy (SEM)

Parasites for SEM were preserved in 70% ethanol and dehydrated in a graded series of ethanol concentrations following Rindoria et al. (2023) and dried with hexamethyldisilazane (HDMS) as per Dos Santos & Avenant-Oldewage (2015). Specimens were dried in a portable glass desiccator overnight at room temperature, gold-coated using an Emscope SC500 sputter coater (Quorum Technologies, Newhaven, U.K.). They were then examined using a Zeiss sigma 500vp scanning electron microscope (Jena, Germany) at 4-6kV at the University of Limpopo.

3.8.3 Data analyses

Parasitic fauna of the host species *X. borealis* and *X. victorinus* were identified using morphological techniques. The diversity indices (parasite community structure) were analysed using an online biodiversity calculator (Young, 2017). The standard infection indices (prevalence and mean intensities) were calculated according to the formulae by Bush et al. (1997). All datasets were tested for normality of distribution using the Shapiro-Wilk method on the PAST software. Two -way Analysis of variance (ANOVA) was used to determine the significance of variations on the spatio-temporal scale. Parasite prevalence between the two frog species was compared using the Chi-Square test All the data was compiled and statistical analyses were done using Microsoft Excel and PAST software.

CHAPTER FOUR

4.0 RESULTS AND DISCUSSIONS

4.1 Introduction

This chapter highlights the various results observed from the different methodological approaches used for this study. A total of 240 host specimens of each species (*X. borealis* and *X. victorinus*) were examined. The results of these are presented according to the specific objectives stated in section 1.3.2.

4.2 To identify and characterise the parasite fauna of *X. borealis* and *X. victorinus* from Lake Victoria Basin morphologically

Based on gross morphology and detailed observations under light and scanning electron microscopy (SEM), the parasites recorded were assigned into four major taxonomic groups: Nematoda, Polyopisthocotyleans, Cestoda, and Digenea. Their diagnostic morphological characteristics include the body shape and size, tegmental features, attachment and feeding structures, reproductive organ structure, and the presence or absence of sclerotized or specialized organs peculiar to each group. The morphological descriptions provided here form the basis for taxonomic placement and subsequent comparisons with existing records of amphibian parasites.

4.2.1 Nematoda

Microscopic examination of nematode specimens recovered from examined hosts revealed morphologically consistent features diagnostic of the family Camallanidae (*Paracamallanus* sp., *Procamallanus* (*Spirocallamanus* sp.)) and *Contracaecum* sp. The

feature visible on the whole-mount preparations examined under light microscopy, with emphasis on anterior and posterior structures.

Family: Camallanidae

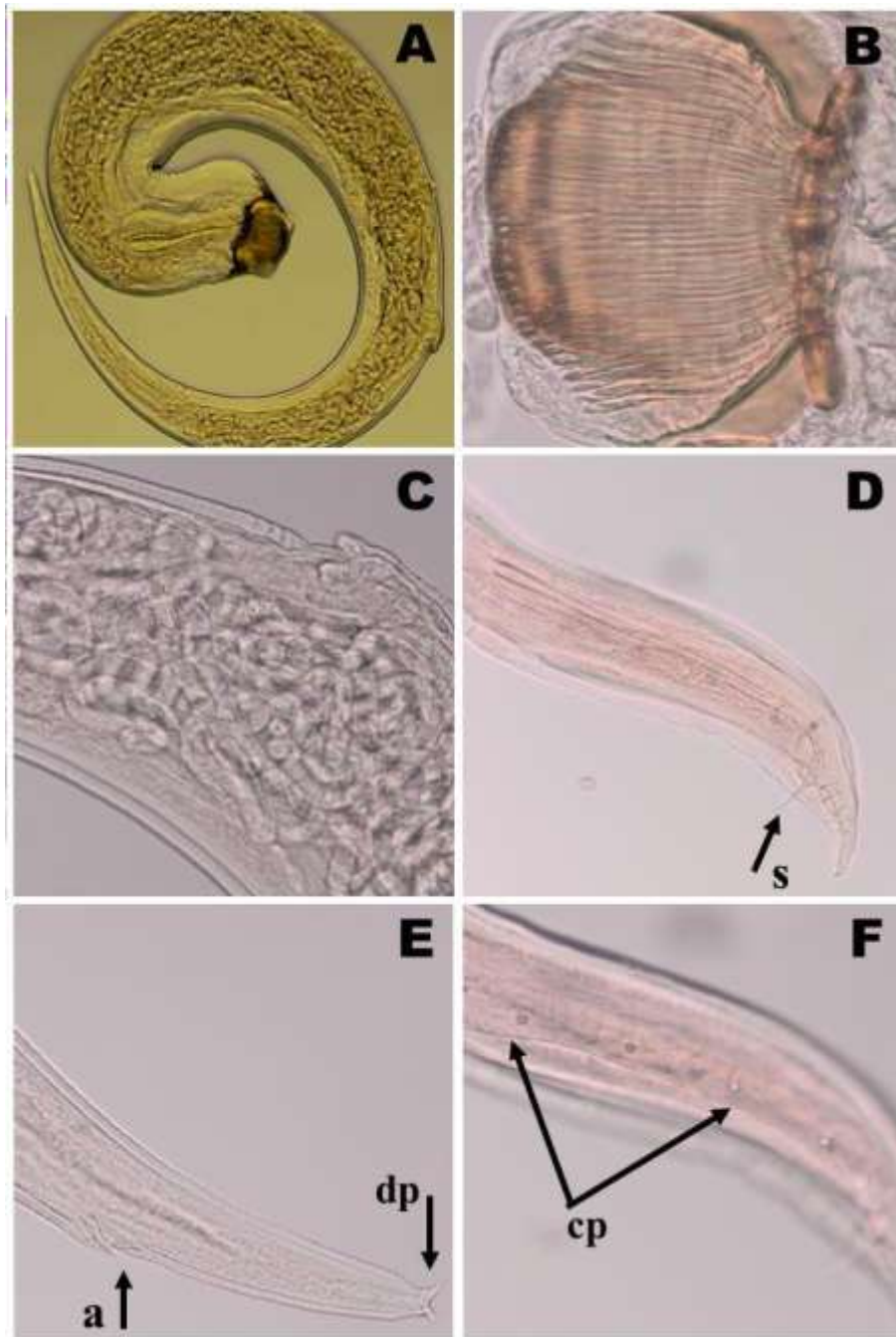
Genus: *Paracamallanus* sp.

The specimens were elongate, cylindrical nematodes with a smooth, thin cuticle and a gently curved to tightly coiled body posture (Figs. 4.1A). Body tapering was evident at both anterior and posterior extremities. The presence of a short, thick-walled buccal capsule with straight internal longitudinal ridges, absence of spiral thickenings, and typical camallanid posterior features supports assignment of these specimens to the genus *Paracamallanus*. The body wall was transparent, allowing visualization of internal organs and pseudocoelomic contents. The internal cavity contained granular material and developing reproductive tissues, consistent with adult camallanid nematodes.

The anterior extremity was characterized by a robust, heavily sclerotized buccal capsule (Fig. 4.1A–B). The buccal capsule was short, thick-walled, and barrel-shaped, with prominent internal longitudinal ridges extending along most of its length (Fig. 4.1B). These ridges were straight, evenly spaced, and sharply defined, producing a distinctly striated appearance. The mouth opening was terminal, and true lips were absent. The morphology of the buccal capsule particularly its compact shape and well-defined internal ridge consistent with diagnostic descriptions of the genus *Paracamallanus*, in which the buccal capsule is strongly sclerotized and ornamented with internal ridges rather than spiral thickenings.

Figure 4.1

Paracamallanus sp. recovered from *X. borealis* and *X. victorinus*: (A) Whole-mount showing strongly curved body and anterior buccal capsule; (B) High magnification of the sclerotized buccal capsule showing straight internal longitudinal ridges; (C) Midbody region showing smooth cuticle and internal organs; (D) Posterior end of male specimen showing spicule(s); (E) Posterior extremity showing anus (a); and digitate processes (dp); (F) Lateral view of midbody region showing paired cervical papillae (cp).



Posterior to the buccal capsule, the oesophagus was visible and appeared divided into an anterior muscular and posterior glandular portion. The cuticle lacked spines or alae. Paired cervical papillae were observed laterally in the anterior-to-midbody region (Fig. 4.1F), consistent with camallanid sensory structures. The posterior extremity was tapered and curved ventrally (Fig. 4.1D, E). In presumed male specimens, the cloacal opening was subterminal. Spicules were visible protruding posteriorly (Fig. 4.1D), slender and lightly sclerotized. A small dorsal papilla was observed near the posterior tip (Fig. 4.1E), and the anus was clearly defined (Fig. 4.1E).

Family: Camallanidae

Genus: *Procamallanus*

Subgenus: *Spirocamallanus* sp.

Specimens assigned to *Procamallanus* (*Spirocamallanus*) sp. were elongate nematodes with a moderately curved body and a more uniform body diameter along much of their length (Fig. 4.2A). The cuticle appeared smooth and flexible, and internal structures were visible through the body wall. The presence of spiral thickenings within the buccal capsule is a defining character of *Procamallanus* (subgenus *Spirocamallanus*). On this basis, the specimen illustrated in Fig. 4.2 A is provisionally assigned to *Procamallanus* (*Spirocamallanus*) sp. The most distinctive feature of these specimens was the buccal capsule bearing conspicuous spiral thickenings (Fig. 4.2B). The capsule was less compact than in *Paracamallanus*, appearing slightly elongated, with spirally arranged ridges or bands encircling the inner surface of the capsule. These spiral structures are diagnostic of the subgenus *Spirocamallanus* within *Procamallanus*. The mouth opening was terminal,

and no lips were observed. The spiral ornamentation of the buccal capsule clearly differentiates these specimens from those assigned to *Paracamallanus*. Posterior to the buccal capsule, the oesophagus was visible and appeared divided into muscular and glandular regions, typical of camallanids. The body cuticle lacked alae or spines, and no external ornamentation was evident along the trunk. The posterior region was tapered and slightly curved (Figs. 4.2C, D). Spicules (Fig. 4.2C) were observed in male specimens; caudal papillae were not clearly resolved in the available images, and the overall tail morphology was consistent with male camallanid nematodes. Digitate processes were observed in female specimens (Fig. 4.2 D).

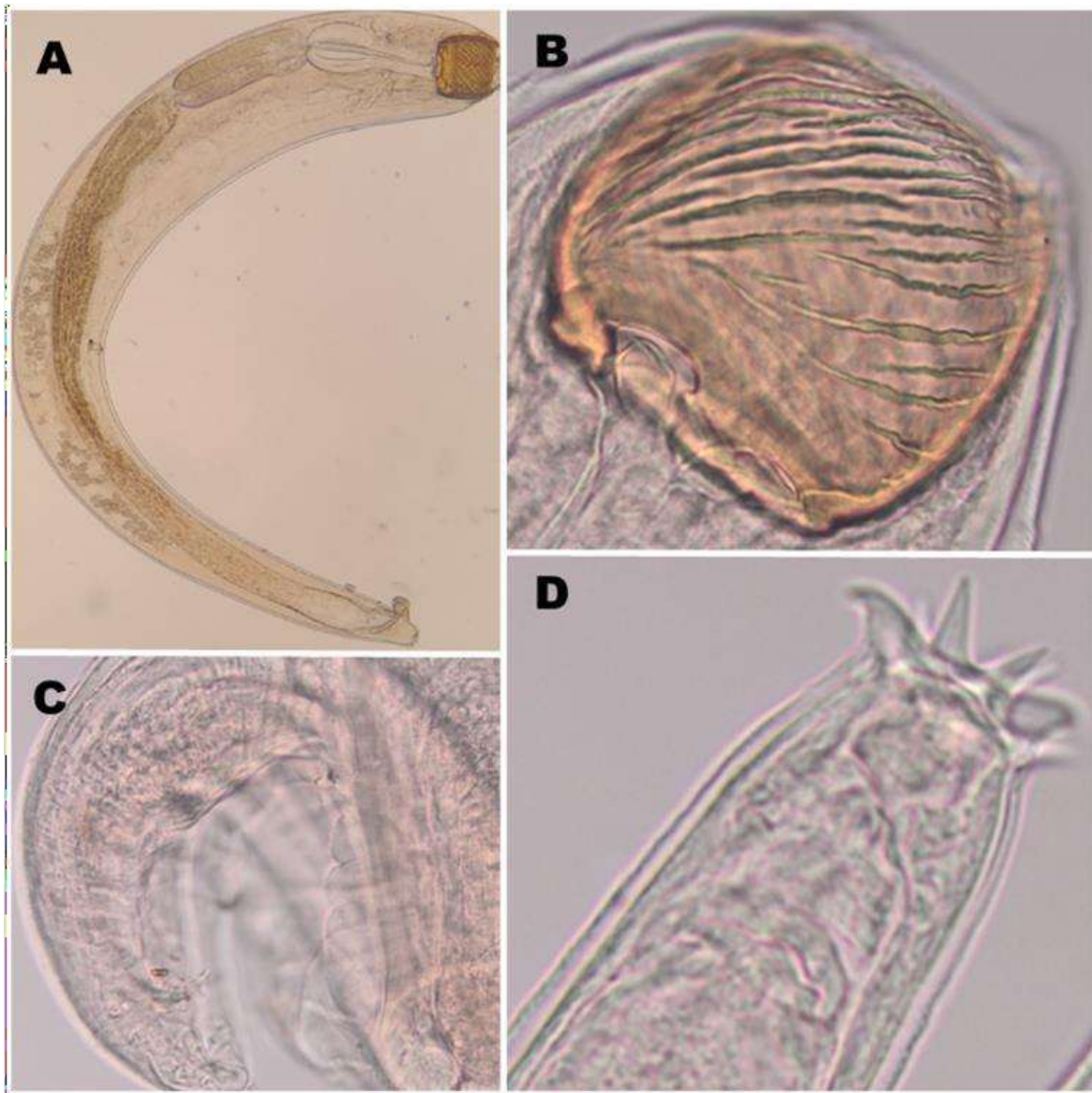
Table 4. 1

Comparative diagnostic characteristics of camallanid genera observed in the present study.

Character	<i>Paracamallanus</i> sp.	<i>Procamaxallanus (Spirocamallanus)</i> sp.
Buccal capsule shape	Short, robust, barrel-shaped	More elongate, cylindrical
Buccal capsule ornamentation	Straight internal longitudinal ridges	Spiral (helical) thickenings
Capsule wall	Thick, heavily sclerotized	Moderately sclerotized
Oral opening	Terminal, lips absent	Terminal, lips absent
Oesophagus	Divided into muscular and glandular portions	Divided into muscular and glandular portions
Cervical papillae	Present	Not clearly resolved
Spicules	Unequal, slender, visible	Not clearly resolved
Tail morphology	Tapered, ventrally curved	Tapered, slightly curved

Figure 4.2

Procamallanus (*Spirocamallanus*) sp. recovered from *X. borealis* and *X. victorianus*. (A) Whole-mount showing elongated body and anterior region. (B) Buccal capsule showing characteristic spiral thickenings diagnostic of *Spirocamallanus*. (C) Posterior body region. (D) Posterior extremity showing tail morphology



Two distinct morphotypes of Camallanid nematodes were identified based on buccal capsule architecture, supporting the presence of multiple Camallanid lineages within the host population. Specimens consistent with *Paracamallanus* exhibited longitudinal ridging, whereas those assignable to *Procamallanus* (subgenus *Spirocamallanus*) displayed characteristic spiral thickenings. These features correspond closely with classical descriptions (Olsen, 1952; Svitin, et al., 2018; Moravec, 2019; Khalifa et al., 2019; Rindoria et al., 2020) and confirm the diagnostic importance of buccal capsule morphology in camallanid taxonomy. The occurrence of these genera in *Xenopus* hosts extends previous reports largely focused on piscine systems (Gibbons, 2010) and may indicate host switching or broader host plasticity within the group (Moravec et al., 2006).

However, while these morphological traits enable reliable generic identification, their usefulness at the species level is more limited. As noted by Moravec (2019) and Svitin et al. (2018), morphological convergence and intraspecific variability can obscure species boundaries within Camallanidae. The distinction of the observed morphotypes as separate species should therefore be interpreted cautiously, particularly in the absence of detailed morphometric data and molecular evidence. This highlights a key limitation of morphology-based taxonomy and underscores the importance of integrative approaches. The co-occurrence of *Paracamallanus* and *Procamallanus* within the same host species further suggests the presence of complex transmission dynamics within the Lake Victoria Basin. Given that Camallanid nematodes utilise aquatic invertebrates as intermediate hosts, their coexistence likely reflects differences in life cycle strategies and host feeding ecology. This finding emphasises the role of *Xenopus* species as reservoirs of diverse nematode fauna and highlights their ecological importance in freshwater parasite transmission networks.

4.2.2 Polypisthocotyleans

Family: Polystomatidae.

Genus: *Polystoma* sp

Species: Unidentified

The microscopic investigations of the recovered flatworms under a compound light microscope and SEM revealed several diagnostic features consistent with members of the family Polystomatidae (Fig. 4.3). The body is long, elongate, and oval; the anterior region tapers slightly and bears a subterminal oral opening, while the posterior expands to form a well-developed haptor.

The opisthaptor is prominent and broadly oval, occupying the posterior extremity of the body. Six muscular suckers are observed on the haptor, arranged symmetrically, consistent with the generic identification of *Polystoma*. Each sucker appears thick-walled and well defined, suggesting strong attachment capability to the host epithelium. A pair of large hamuli is observed embedded within the haptoral tissue. They are robust and strongly sclerotised, sickle-shaped, each consisting of a well-developed handle, guard, and curved hook. The margin of the haptor depicts three pairs of holes that are small uniform in size and evenly distributed corresponding to marginal hooklets consistent with the diagnostic features of the polystomids.

Figure 4. 3

Scanning electron micrograph of *Polystoma* sp. from *X. borealis* and *X. victorianus* showing

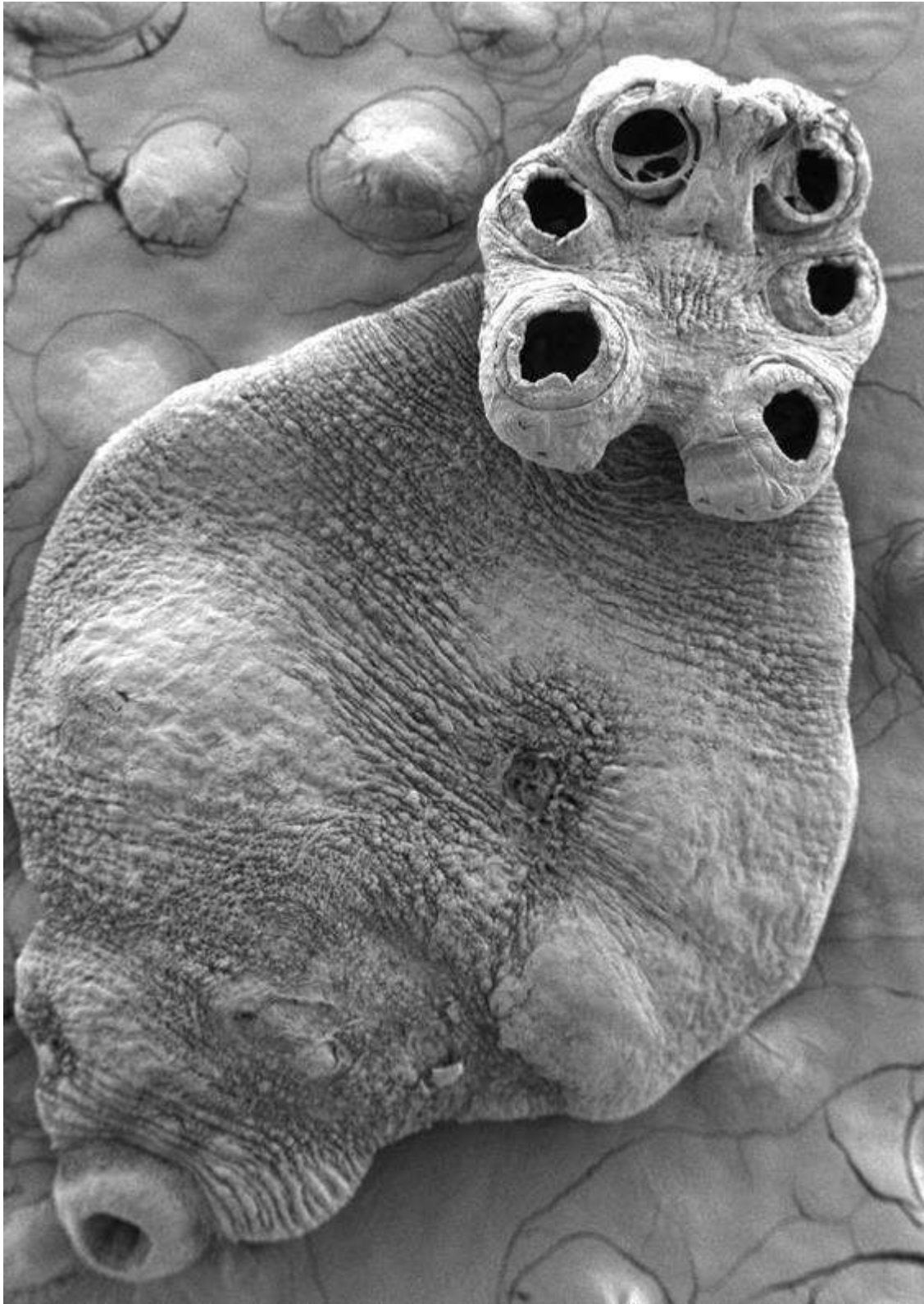
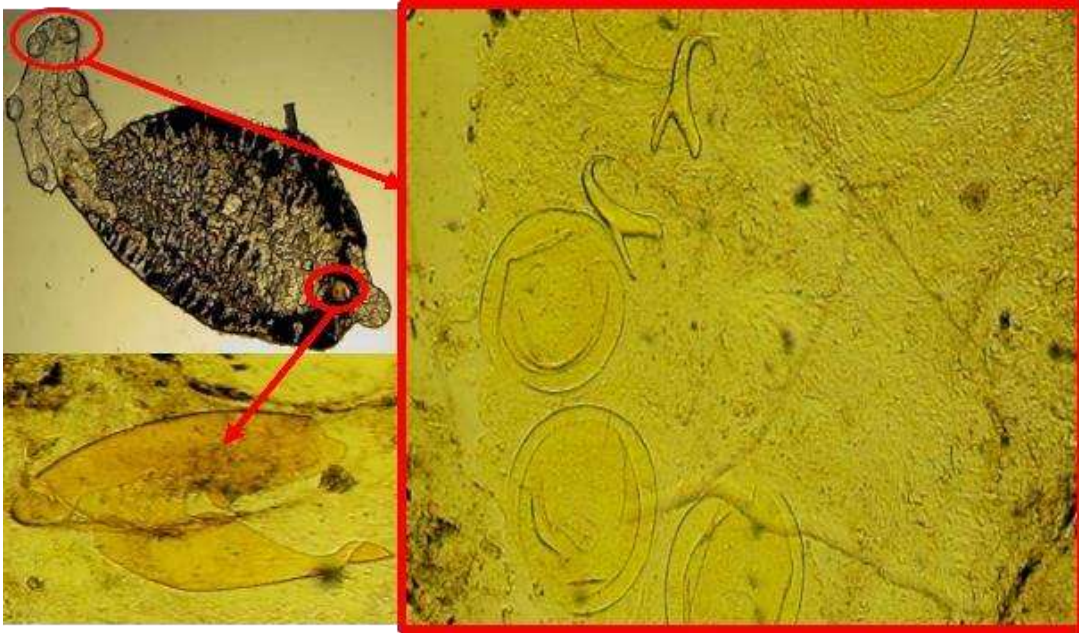


Figure 4. 4

Light microscopy images of Polystoma sp. recovered from X. borealis and X. victorianus in the present study.



The digestive system consists of a single anterior mouth leading to a bifurcated intestine, with two intestinal caeca extending posteriorly without re-joining. These features distinguished polystomatids from cestodes (which lack a digestive tract), and trematodes (which possess different gut patterns). Reproductive structures typical of hermaphroditic monogeneans were evident. A single large oval operculated egg was observed, visible without specialized staining, in mature individuals, indicative of active reproduction. Vitelline follicles were distributed extensively throughout the body except in the region surrounding the oral sucker, ovary, uterus and haptor suckers. The body surface appeared smooth and unarmed, with no spines or scales observed, although slightly granular under SEM.

Polypisthocotyleans including *Polystoma* were also identified, supporting their well-established association with amphibian hosts (Bentz et al., 2001; Verneau et al., 2009). The presence of a prominent opisthaptor bearing six symmetrically arranged muscular suckers is a key diagnostic feature of *Polystoma* spp. (Verneau et al., 2009). The suckers are typically thick-walled and well defined, as also reported in polystomatids from amphibian hosts (Du Preez & Theunissen, 2021). In addition, the occurrence of a pair of large, robust, sclerotised hamuli with distinct handles, guards, and curved hooks is consistent with descriptions of *Polystoma* species in previous studies (Chaabane et al., 2019; Landman et al., 2023; Du Preez & Theunissen, 2021), where such morphology is characteristic and widely applied in taxonomic identification.

The observed attachment structures are indicative of strong host-site specificity, particularly within the urinary bladder, and reflect adaptations for persistent attachment in dynamic internal environments (Du Preez & Theunissen, 2021). The presence of *Polystoma* in *X. borealis* and *X. victorianus* represents a potentially novel host-parasite association in Kenya, contributing to the limited regional data on polystomatid diversity.

4.2.3 Digenea

Scanning electron microscopy examination of trematode specimens recovered from *X. borealis* and *X. victorianus* revealed digenean parasites exhibiting a suite of external morphological characteristics consistent with members of the class Trematoda (Digenea). Owing to the absence of internal anatomical data and molecular sequences, identification is restricted to higher taxonomic levels, and the specimens are herein treated as *Digenea incertae sedis* (undefined specimen)

Unidentified Digenea trematode 1

Class: Trematoda

Subclass: Digenea

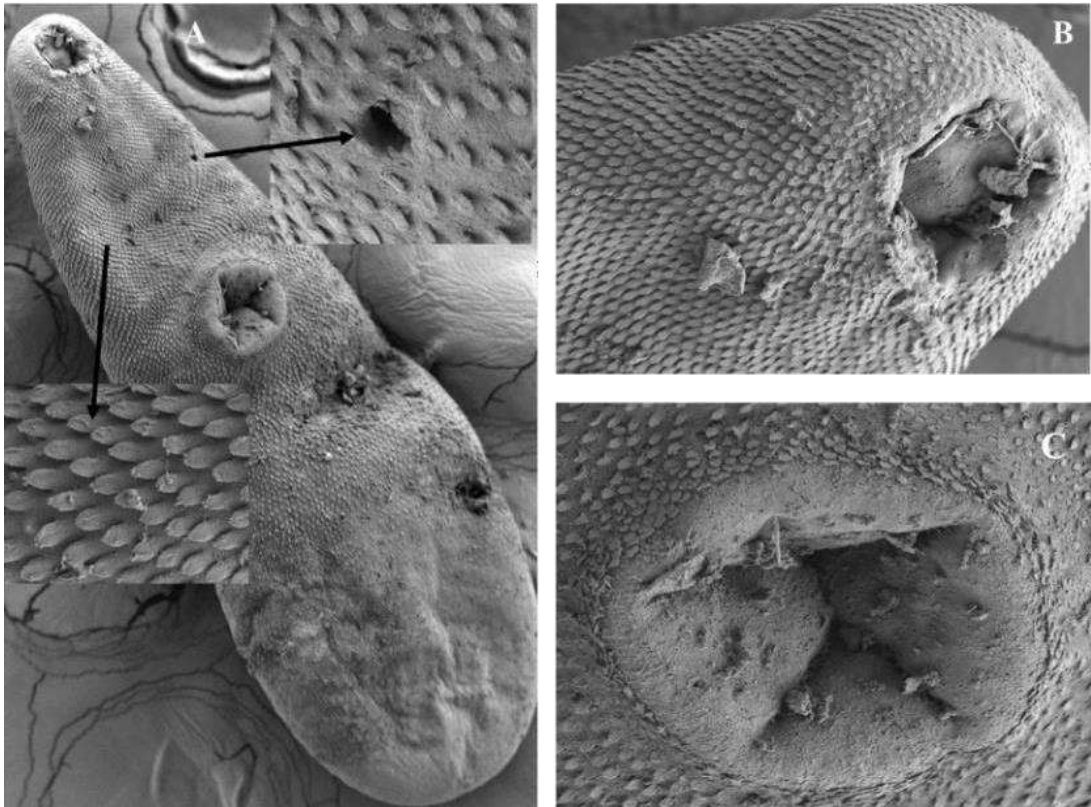
Taxonomic status: Digenea *incertae sedis*

The body was elongate, dorsoventrally flattened, and robust, with a slightly tapered anterior region and a broadly rounded, blunt posterior extremity (Fig. 4.5A). No posterior adhesive organs, caudal appendages, or terminal suckers were observed. The overall body surface was thick and heavily ornamented, suggesting adaptation for firm attachment within host tissues (Fig. 4.5A). The oral sucker was terminal to subterminal, circular, and well developed, with a thickened muscular rim surrounding the oral aperture (Fig. 4.5B). No oral stylet, hooks, or collar spines were observed. The tegument immediately surrounding the oral sucker bore dense surface ornamentation (Fig. 4.5 B). A conspicuous ventral sucker (acetabulum) was present in the anterior half of the body, positioned posterior to the oral sucker (Figs. 4.5A, C). The acetabulum was large, circular, and deeply recessed within the body wall. High-magnification SEM images showed a smooth internal acetabular surface bordered by thick muscular margins, indicating a strong attachment organ (Fig. 4.5C).

The tegument was characterised by dense coverage of flattened, overlapping, scale-like surface elements, arranged in an imbricate pattern across the body surface (Fig. 4.5A). These structures formed a continuous armoured appearance rather than discrete pointed spines. Tegumental ornamentation was most pronounced in the anterior region and gradually decreased in density towards the posterior extremity, though surface structures remained evident throughout.

Figure 4. 5

Scanning electron micrographs of a unidentified digenean trematode 1 recovered from X. borealis and X. victorinus: (4.5A) Whole-body view showing elongate, dorsoventrally flattened body with terminal oral sucker and ventral sucker; (4.5B) anterior extremity illustrating oral sucker and persistent tegumental surface ornamentation: (Fig 4.5C) high-magnification view of the ventral sucker (acetabulum) showing a deeply recessed muscular cavity with smooth internal surface.



No circumoral collar spines, lateral spines, or differentiated tegumental zones were observed. The tegument appeared thick and robust, suggesting a protective and friction-enhancing role during attachment to host tissues. Tegumental spination gradually decreased in density towards the posterior extremity but remained present across the body surface. The posterior end lacked any specialized adhesive structures or terminal sucker and retained the blunt, rounded morphology typical of large-bodied digeneans (Fig 4.5A).

Unidentified Digenea trematode 2

Family: Diplodiscidae

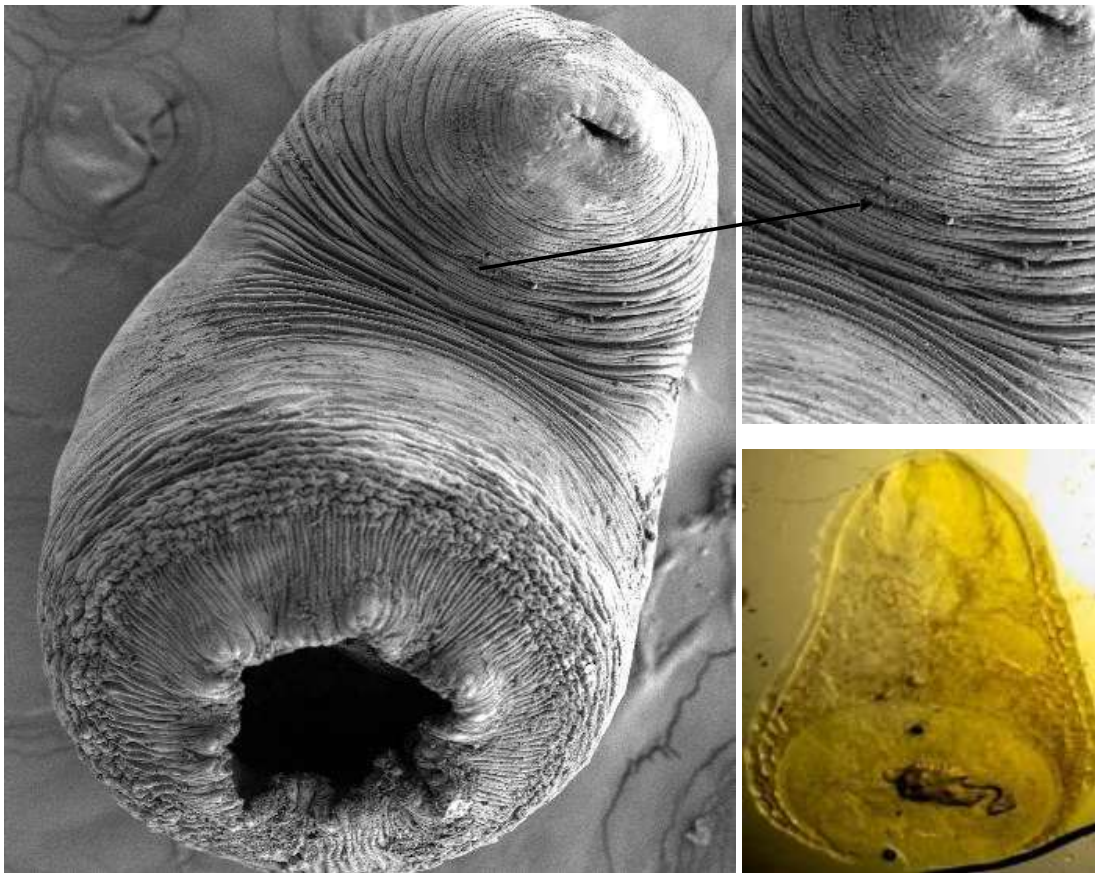
Genus: indeterminate (*incertae sedis*)

Scanning electron microscopy revealed a thick-bodied, compact digenean trematode with a broadly rounded anterior region and a posterior region dominated by a large sucker. The body appeared fleshy, dorsoventrally flattened, and non-segmented, consistent with digenean trematodes. This specimen is best interpreted as an amphistome-type digenean, based on the posteriorly positioned, massively developed ventral sucker, overall body form, and tegumental architecture. A small, terminal oral sucker was present at the anterior extremity. The oral aperture was circular to slit-like and surrounded by a thick muscular rim. SEM images showed distinct concentric ridges and folds around the oral sucker, corresponding to underlying muscular architecture. No oral stylet, hooks, circumoral spines, or cephalic collar structures were observed. The most prominent external feature was a very large, posteriorly positioned ventral sucker (acetabulum). The acetabulum occupied a substantial proportion of the posterior body region and was deeply recessed within the body wall. The sucker opening was broad and circular, bordered by thickened muscular lips. The tegument was smooth, lacking discrete tegmental spines, hooks, or plates. Instead, the body surface was characterised by dense transverse annulations, folds, and wrinkles, giving the tegument a concentrically ridged appearance. These surface features were continuous across the body and more pronounced around both suckers. No differentiated tegmental zones, lateral spines, or collar armature were observed. Light microscopy confirmed a compact, thick-bodied trematode with no evidence of a cephalic collar or external armature. Internal structures were not sufficiently

resolved to allow assessment of reproductive organ arrangement or developmental stage. Egg morphology was not observed. parasite interactions (Smyth & Halton, 1983; Dalton et al., 2004; Brennan et al., 1991; Panyarachun et al., 2013), indicating that these parasites may occupy distinct microhabitats within the host.

Figure 4. 6

*Scanning electron micrographs of an unidentified digenean trematode 2 recovered from *X. borealis* and *X. victorianus* in the present study.*



Digenean trematodes were conservatively assigned to higher taxonomic levels due to the absence of internal anatomical and molecular data, consistent with current systematic practices (Stout et al., 2024; Cribb et al., 2025). The tegumental diversity observed under SEM, including both imbricate ornamentation and annulated folding, is consistent with

similar surface patterning reported in digenean trematodes, where variations such as tegumental spination, scale-like structures, and surface folding have been documented across taxa (Maraganga et al., 2023; Shigoley et al., 2025; Rindoria et al., 2025). Such variation suggests functional differentiation among morphotypes and is associated with protective, absorptive, and adaptive functions in host tissue.

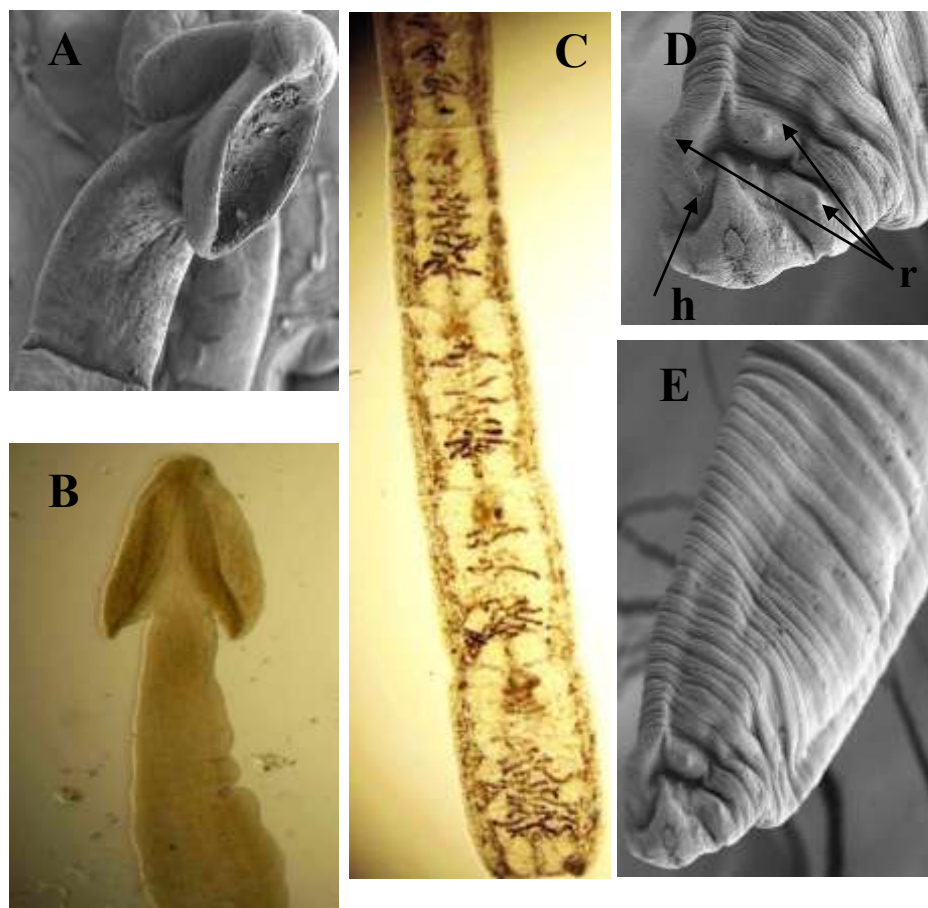
4.2.4 Cestode

Family: Cephalochamydidae

Genus: *Cephalochlamys* sp.

Figure 4. 7

Scanning electron and light micrographs showing morphological features of *Cephalochamydidae* cestode recovered from *X. borealis* and *X. victorianus*. (A) SEM image of the anterior region showing the scolex bearing paired bothria. (B) Light microscopy image of the scolex illustrating overall bothrial configuration and early strobilation. (C) Light micrograph of the strobila showing segmental organization and protoglids. (D) SEM image of the posterior extremity showing terminal proglottids, rp (rounded tegumental protrusions), and h (solitary hook-like structure.) (E) SEM image of the posterior strobila highlighting transverse annulations and longitudinal tegumental ridges



The microscopic investigations of the recovered flatworms under a compound light microscope revealed several diagnostic features consistent with members of the family Cephalochamydidae. The specimen has a distinct neck with the scolex appearing as an arrowhead with a pair of bothria uniting at the apex.

Scanning electron microscopy and light microscopy (Figs. 4.7 A-B) revealed a cestode parasite with a distinctly differentiated scolex, neck, and anterior strobila. The scolex was elongate and laterally compressed, forming a characteristic cephalic collar or hood-like structure partially overlapping the anterior strobila. The anterior region (Figs. 4.7 A and B) was characterised by a distinct scolex bearing a pair of shallow, elongate bothria (dorsal and ventral) that united at the apex. The bothria appeared as longitudinal, deep, muscularised grooves extending along the scolex surface, lacking hooks or an apical rostellum. Light microscopy confirmed the bothrial arrangement. The neck region was short and indistinct, transitioning rapidly into the strobila. The strobila (Fig. 4.7 C) was elongated and composed of numerous proglottids, with segmentation becoming progressively more defined posteriorly.

Scanning electron microscopy observations of the posterior region (Figs. 4.7D, E) showed terminal proglottids with pronounced transverse annulations and well-developed longitudinal tegumental ridges. The posterior extremity was blunt and exhibited surface folding consistent with contraction or pre-detachment of terminal segments. The surface exhibited multiple rounded protrusions distributed irregularly across the tegument. These protrusions varied slightly in size and elevation and appeared embedded within the tegumental surface rather than projecting freely. Notably, a single, small hook-like structure was observed at the posterior extremity (Fig. 4.7D).

This structure was curved, sharply defined, and distinct from surrounding tegumental folds. No paired or repetitive hook arrangements were observed, and the structure did not resemble a typical scolex armature. Its position at the posterior end suggests it may represent a modified tegumental structure, a remnant attachment feature, or a contraction-related projection rather than a true posterior anchoring organ.

Cestodes exhibiting characteristics consistent with cephalochlamyd taxa were also recorded, with scolex morphology indicative of adaptation for attachment via muscular adhesion rather than mechanical anchoring (Yamaguti, 1959; Khalil et al., 1994; Nachtigall, 1974; Mbokane et al., 2020). The presence of tegumental protrusions, potentially functioning as sensory or glandular structures (Casado et al., 1999), further highlights the structural and functional complexity of these parasites, although their precise role remains unclear.

Examination of helminths from *X. borealis* and *X. victorianus* in the Lake Victoria Basin revealed a diverse parasite assemblage. This diversity reflects the ecological complexity of aquatic systems within the Lake Victoria Basin, where the availability of multiple intermediate hosts facilitates the maintenance of complex parasite life cycles. Similar patterns of high helminth diversity have been reported in *Xenopus* species globally (Furman et al., 2015), particularly in *X. laevis* from South Africa, where comparable assemblages have been documented (Mbokane et al., 2020), suggesting that such diversity may be a general characteristic of aquatic *Xenopus* populations.

Overall, the findings of this study demonstrate that morphological approaches, particularly when combined with SEM, provide a robust basis for the identification and characterisation of helminth parasites in *Xenopus* species. However, limitations related to

incomplete anatomical resolution, absence of morphometric data, and the potential for cryptic diversity highlight the need for integrative taxonomic approaches. The incorporation of molecular data is therefore essential to validate morphological identifications and achieve reliable species-level resolution, thereby strengthening our understanding of parasite diversity within the Lake Victoria Basin.

4.3 To determine the spatio-temporal patterns of parasite infection indices (prevalence and mean intensity) and diversity in *X. borealis* and *X. victorianus*.

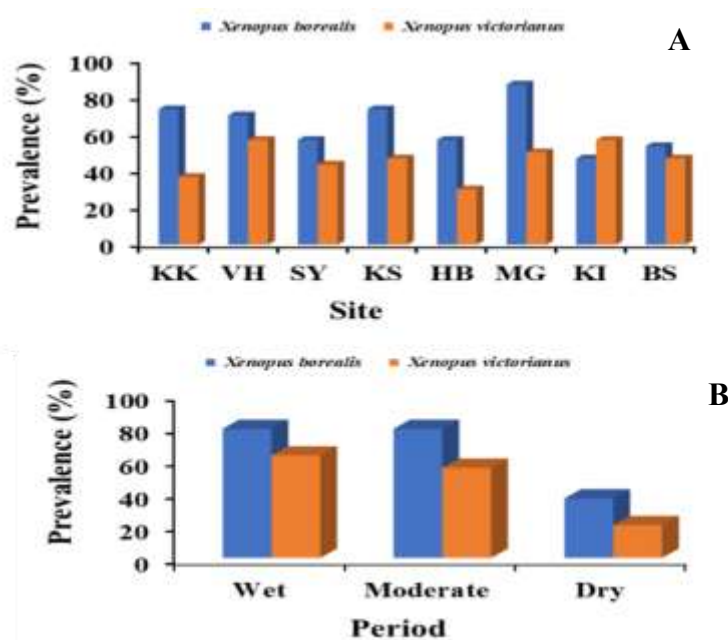
Four taxa of endohelminths were recorded in both *X. borealis* and *X. victorianus*. These were broadly categorized as cestodes (*Cephalochlamys* sp.) and nematodes (*Contracaecum* sp., *Camallanus* spp.) digenea and monopisthocotyleans (*Polystoma* sp.). The prevalence and mean intensities of these parasites varied between the two host species (Figs. 4.8-4.17). *Camallanus* spp. exhibited the highest prevalence and mean intensity in both hosts (Figs. 4A, 5B). Notably, in *X. victorianus*, *Camallanus* spp. had a higher prevalence (85%) and mean intensity (8.3) during the dry season (Figs. 4.16-4.17) compared to *X. borealis*. In *X. borealis*, the prevalence of *Camallanus* spp. remained relatively stable, ranging from 75% to 77.5%, indicating minimal seasonal influence on parasite load in this host. In contrast, *Polystoma* sp. and digenean spp. were less prevalent overall. Temporally, parasite prevalence was highest during the wet season across all taxa, compared to the moderate and dry seasons. Spatially, infestation levels were the highest in Kakamega, Vihiga, Kisii, and Kisumu counties, a pattern observed consistently for both host species.

4.3.1 Contracaecum sp.

Contracaecum sp. infections were widespread and exhibited high prevalence across most sampling sites (Fig.4.8). *Xenopus borealis* showed consistently higher prevalence than *X. victorinus* at all sites, with exceptionally high values at MG and KS, where prevalence exceeded 80%. In *X. victorinus*, prevalence was comparatively lower but still substantial, particularly at VH, KI, and BS. Seasonal patterns showed the highest prevalence of *Contracaecum* sp. during the wet and moderate seasons in both species, with a sharp decline during the dry season (Fig. 4.8). The reduction was more pronounced in *X. victorinus*, where dry-season prevalence dropped to nearly half of wet-season values.¹

Figure 4. 8

Spatial (A) and temporal (B) variations in prevalence for Contracaecum sp in X. borealis and X.victorinus



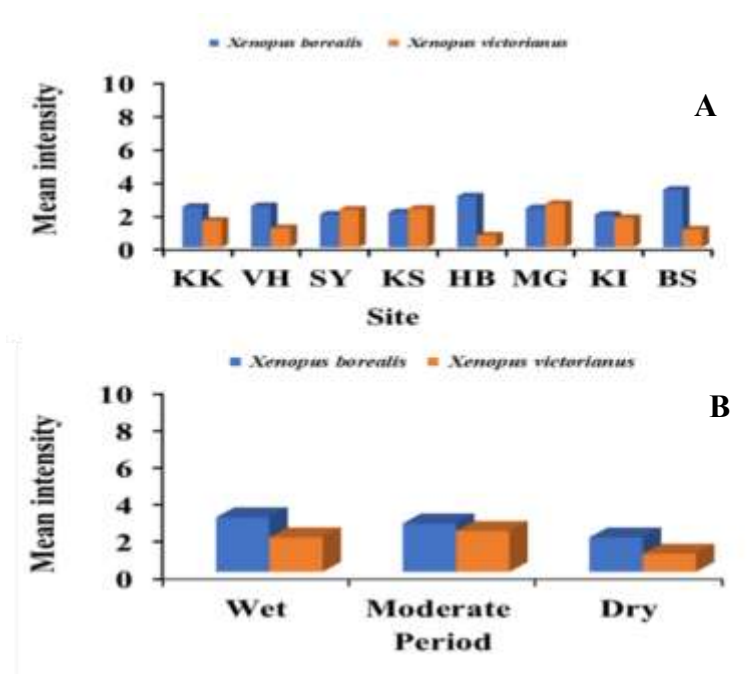
¹ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Mean intensity values for *Contracaecum* sp. were generally higher than those observed for digeneans and cestodes (Figs. 4.9,4.10,4.14). In *X. borealis*, intensity ranged from approximately 2.0 to 3.8 parasites per infected host, with the highest intensities recorded at BS and HB. *X. victorinus* exhibited lower intensities overall, with most sites recording values below 3.0.

Temporal patterns of mean intensity followed a similar trend, peaking during the wet season, remaining moderately high during the moderate season, and declining during the dry season (Fig. 4.10). Across all seasons, *X. borealis* maintained higher intensities than *X. victorinus*.²

Figure 4.9

Spatial (A) and temporal (B) patterns of the mean intensity of infection in X. borealis and X. victorinus for Contracaecum sp. in the present study.



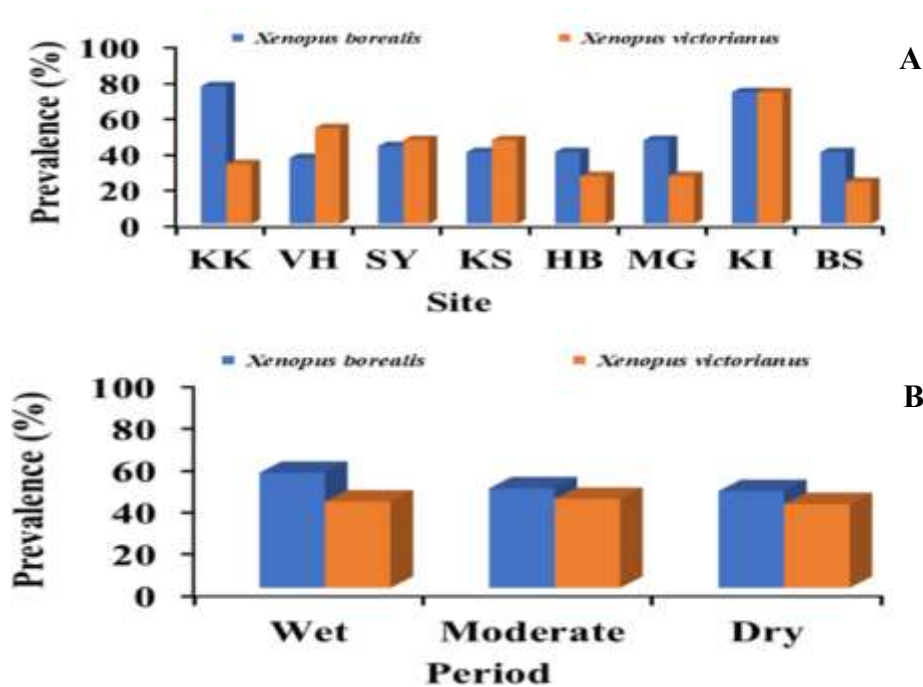
² Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

4.3.2 Cestode (*Cephalochlamys* sp.)

Cestode prevalence demonstrated clear interspecific and spatial differences (Fig. 4.10). *Xenopus borealis* exhibited consistently higher prevalence across most sites, with particularly elevated values at KK and KI, where prevalence approached or exceeded 70%. In contrast, *X. victorinus* showed lower and more variable prevalence, with notable reductions at HB and MG. Seasonal variations in prevalence were relatively stable across wet, moderate, and dry seasons in both host species (Fig. 4.10). *Xenopus borealis* consistently maintained higher prevalence than *X. victorinus* across all seasons, with only minor seasonal fluctuations.³

Figure 4. 10

Spatial (A) and temporal (B) variations in prevalence for Cephalochlamys sp in X. borealis and X. victorinus

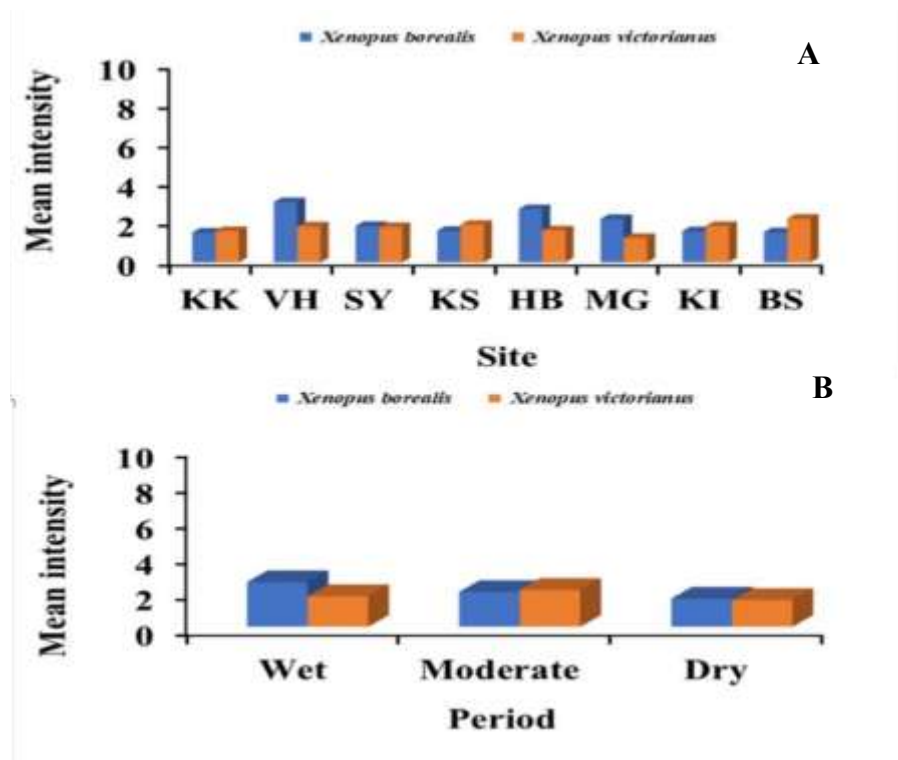


³ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Mean intensity of cestode infections remained low across all sites for both species, generally ranging between 1.5 and 3.0 parasites per infected host (Fig.4.11). *Xenopus borealis* displayed slightly higher intensities at VH and HB, whereas *X. victorinus* showed modestly elevated intensity at BS relative to other sites. Mean intensity showed limited seasonal variation, remaining close to 2–3 parasites per infected host in *X. borealis* and slightly lower in *X. victorinus* (Fig. 4.11). The highest intensities were observed during the wet season, followed by a slight decline during the dry season.⁴

Figure 4. 11

Spatial (A) and temporal (B) variations in mean intensity for Cephalochlamys sp in X. borealis and X. victorinus.



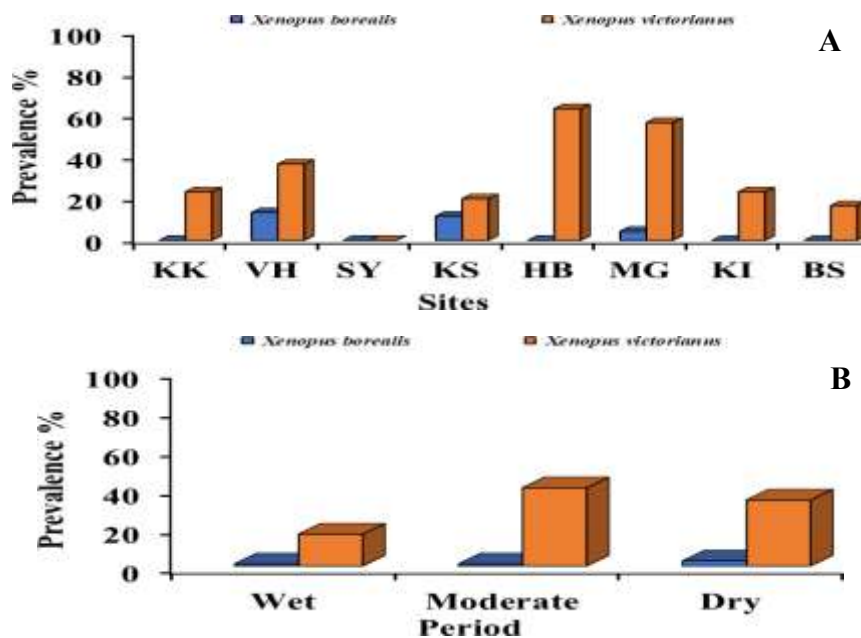
⁴ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

4.3.3 Polystoma sp.

Prevalence varied across sampling sites for both *X. borealis* and *X. victorinus* (Fig. 4.12). Overall, *X. victorinus* exhibited higher prevalence values than *X. borealis* at all sites. In *X. victorinus*, prevalence was highest at HB and MG, exceeding 60% and approximately 55%, respectively. Moderate prevalence levels were recorded at VH and KI, while lower prevalence values were observed at KK, KS, SY, and BS. In contrast, *X. borealis* showed consistently low prevalence across all sites, generally remaining below 15%. The highest prevalence for *X. borealis* was observed at VH and KS, while minimal or near-zero prevalence was recorded at KK, SY, KI, and BS.⁵

Figure 4. 12

Spatial (A) and temporal (B) variations in prevalence for Polystoma sp in X. borealis and X. victorinus.

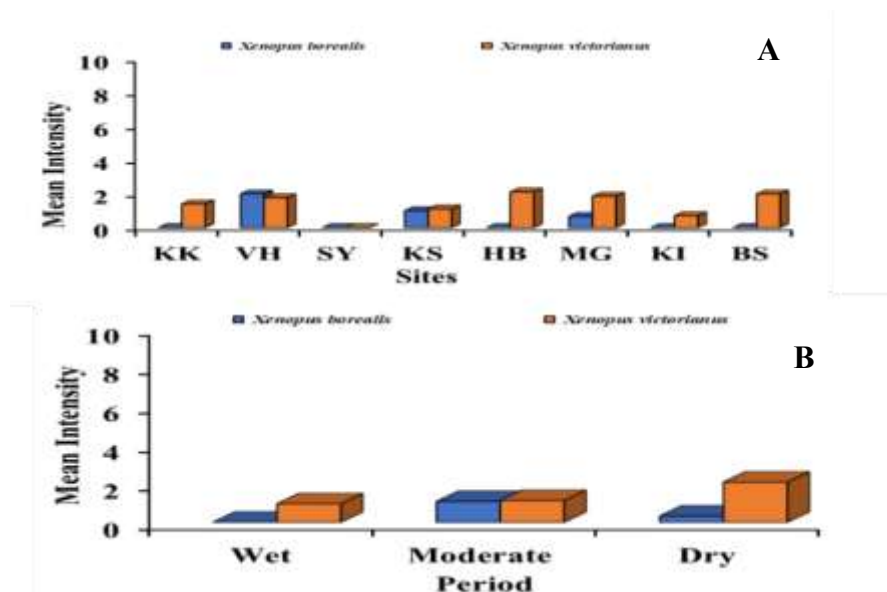


⁵ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Although prevalence differed across sampling periods for both host species, *X. victorinus*, prevalence increased from the wet period (approximately 20–25%) to a peak during the moderate period (approximately 45–50%), followed by a slight decline during the dry period (approximately 40%). In *X. borealis*, prevalence remained low across all periods, with values consistently below 10%. Slight increases were observed during the moderate and dry periods compared to the wet period, but overall variation across periods was minimal relative to *X. victorinus*.⁶

Figure 4. 13

Spatial (A) and temporal (B) variations in mean intensity for Polystoma sp in X. borealis and X. victorinus



⁶ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Mean intensity of infection varied among sampling sites for both *X. borealis* and *X. victorinus* (Fig. 4.13). Overall, *X. victorinus* exhibited higher mean intensity values than *X. borealis* at most sites. In *X. victorinus*, mean intensity was highest at HB and BS, with values exceeding 2 parasites per infected host. Relatively high intensities were also observed at MG and VH, while moderate values were recorded at KK and KS. Lower mean intensity was observed at KI and SY.

In contrast, *X. borealis* showed generally low mean intensity across sites, with values close to zero at KK, SY, HB, KI, and BS. The highest mean intensity for *X. borealis* was recorded at VH (approximately 2 parasites per infected host), followed by KS and MG, which showed moderate values. Overall, spatial variation in mean intensity was more pronounced in *X. victorinus* than in *X. borealis*. Mean intensity also varied across sampling periods (Fig. 4.13). In *X. victorinus*, mean intensity increased progressively from the wet period (approximately 1 parasite per infected host) to the moderate period (approximately 1.2), reaching the highest value during the dry period.

In *X. borealis*, mean intensity remained low across all periods. The lowest values were recorded during the wet period, with a slight increase during the moderate period (approximately 1.1), followed by a decline during the dry period. Compared to *X. victorinus*, temporal variation in mean intensity in *X. borealis* was minimal.

4.3.4 Digenea

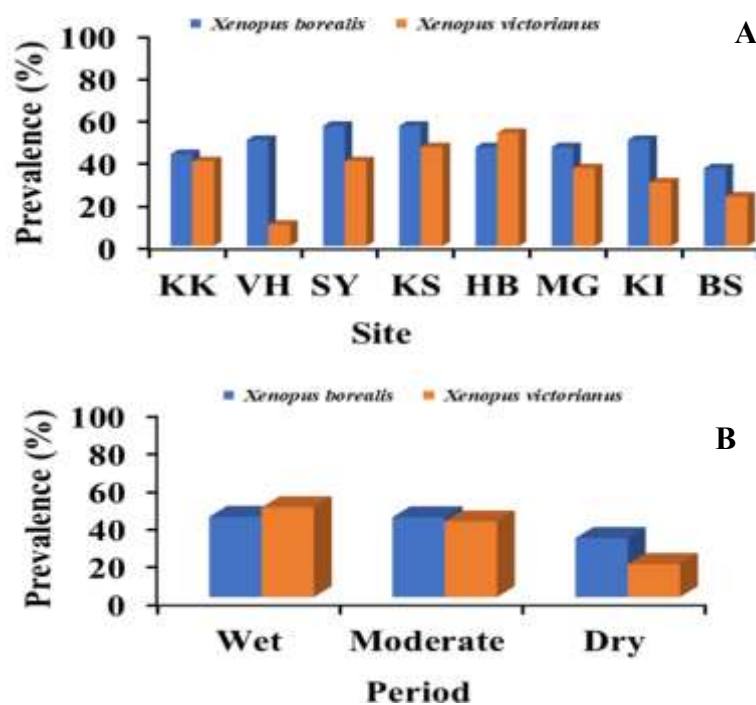
The prevalence of digenean infections varied markedly among sampling sites in both *X. borealis* and *X. victorinus* (Fig. 4.14). Across most sites, *X. borealis* consistently exhibited higher prevalence values than *X. victorinus*. Notably high prevalence in *X. borealis* was recorded at SY, KS, KI, and VH, where values generally exceeded 50%. In

contrast, *X. victorinus* showed comparatively lower prevalence at several sites, with a pronounced reduction at VH, where prevalence dropped to the lowest observed levels among all sites.

Seasonally, digenean prevalence was highest during the wet season in both host species, with *X. victorinus* showing slightly higher prevalence than *X. borealis* during this period. Prevalence declined moderately during the dry season, with a more pronounced reduction observed in *X. victorinus*.⁷

Figure 4. 14

Spatia(A)l and temporal(B) variations in prevalence for Digenea spp in X. borealis and X. victorinus.



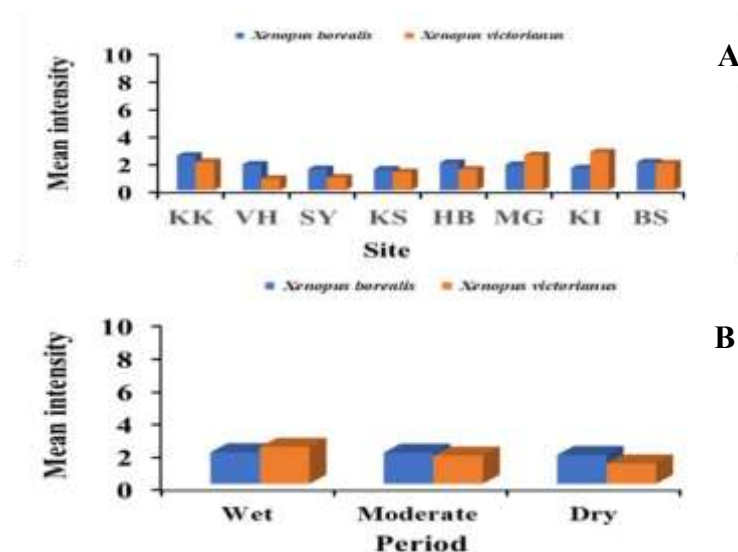
⁷ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Mean intensity of digenean infection showed relatively low but variable values across sites (Fig. 4.15). In *X. borealis*, intensity ranged approximately between 1.5 and 2.7 parasites per infected host, with slightly elevated intensities at KK and HB. *X. victorinus* exhibited comparable but marginally lower intensities overall, although higher values were observed at MG and KI, indicating localized aggregation of infections.

Temporal variations showed a similar seasonal pattern, peaking during the wet season in both species (approximately 2.5–3.0), declining slightly during the moderate season, and reaching the lowest values during the dry season (Fig. 4.15). Across all seasons, *X. borealis* maintained marginally higher mean intensities than *X. victorinus*.⁸

Figure 4. 15

Spatial and temporal variations in mean intensity for Digenea spp in X. borealis and X. victorinus.



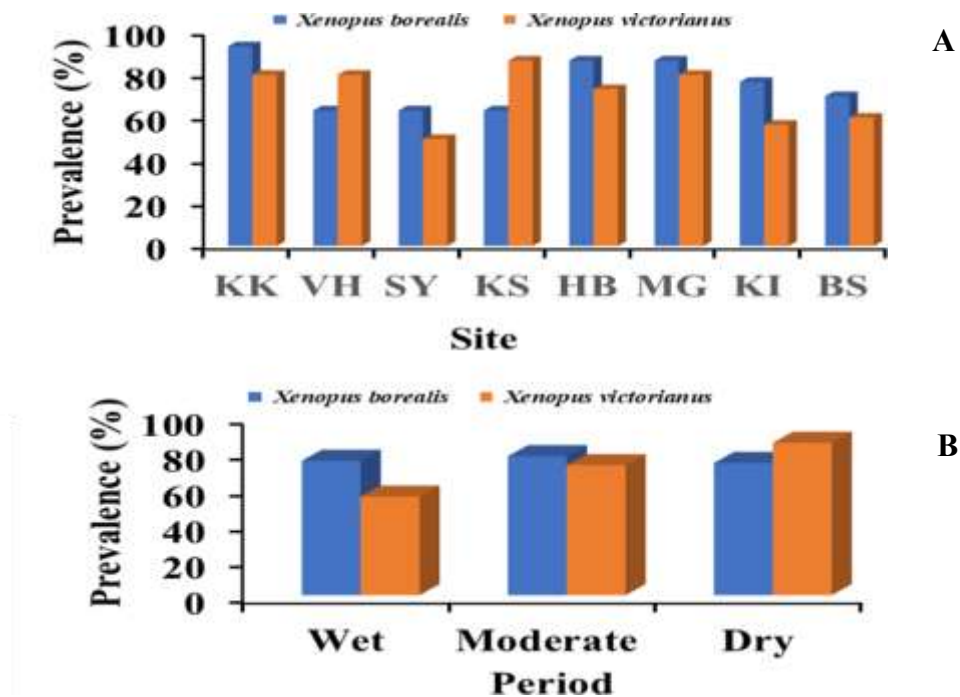
⁸ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

4.3.5 Camallanus spp.

The prevalence and mean intensity of *Camallanus* spp infection varied between *X. borealis* and *X. victorinus* across sampling sites and seasons within the Lake Victoria Basin (Fig. 4.16).⁹

Figure 4.16

Spatial and temporal variations in prevalence for Camallanus spp. in X. borealis and X. victorinus.



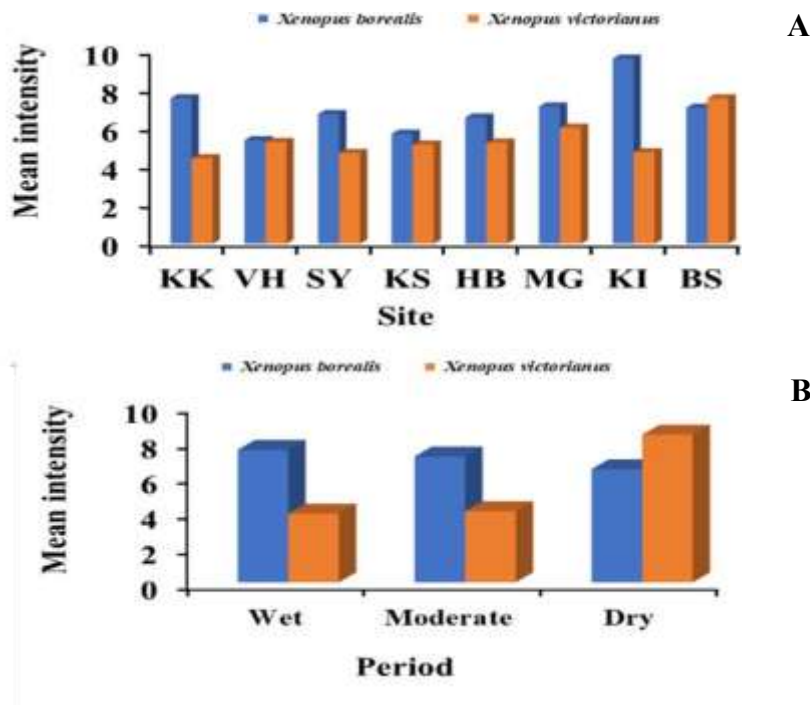
⁹ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Across the sampled sites, *Camallanus* spp. prevalence was consistently high in both host species, although variation was evident among locations (Fig. 4.16). In *X. borealis*, prevalence ranged from approximately 65% to over 90%, with the highest values recorded at KK, HB, and MG, and comparatively lower prevalence observed at VH and SY. In *X. victorinus*, prevalence ranged from about 50% to 90%, with the highest prevalence recorded at KS and MG, and the lowest at SY. At most sites, *X. borealis* exhibited higher prevalence than *X. victorinus*, except at KS and VH, where *X. victorinus* showed relatively higher prevalence.

Seasonal analysis revealed differences in *Camallanus* spp. prevalence between wet, moderate, and dry periods (Fig. 4.17). In *X. borealis*, prevalence remained consistently high across all seasons, ranging from approximately 78% during the wet period to over 85% during the moderate period, with a slight decline observed during the dry period. In contrast, *X. victorinus* showed a marked increase in prevalence from the wet period (approximately 60%) to the dry period, where prevalence exceeded 90%. During the dry period, *X. victorinus* exhibited higher prevalence than *X. borealis*.

Figure 4. 17

Spatial (A) and temporal (B) variations in mean intensity for Camallanus spp. in X. borealis and X. victorinus



Mean intensity of *Camallanid* spp. infection also varied among sites and between host species (Fig. 4.17), top right). In *X. borealis*, mean intensity ranged from approximately 5 to 10 parasites per infected host, with the highest intensity observed at KI and KK. In *X. victorinus*, mean intensity ranged from about 4 to 8 parasites per infected host, with the highest values recorded at BS and MG. Overall, *X. borealis* exhibited higher mean intensity than *X. victorinus* at most sites, although overlapping values were observed at VH and KS.¹⁰

¹⁰ Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS).

Mean intensity of *Camallanus* spp. infection exhibited seasonal variation in both host species (Fig. 4.17). In *X. borealis*, mean intensity was highest during the wet period (approximately 8 parasites per host) and declined slightly during the moderate and dry periods. Conversely, *X. victorinus* showed a progressive increase in mean intensity from the wet period (approximately 4 parasites per host) to the dry period, where mean intensity reached approximately 9 parasites per infected host.

Overall, *Camallanus* spp. infection was characterized by high prevalence and moderate to high mean intensity in both *X. borealis* and *X. victorinus*, with clear spatial and seasonal variation observed across the Lake Victoria Basin.

4.3.6 Spatial variation in parasite taxa prevalence

4.3.6.1 Chi-square (χ^2)

Chi-square (χ^2) tests revealed significant spatial and temporal variation in the prevalence of several parasite taxa across the eight Lake Victoria Basin sampling sites for both *Xenopus* species (Table 4.2). In *X. borealis*, significant spatial differences in prevalence were detected for *Contracaecum* sp., *Camallanus* spp., *Cephalochlamys* sp., and *Polystoma* sp. In contrast, no significant spatial variation was observed for digeneans (*incertae sedis*) in *X. borealis*. For *X. victorinus*, significant spatial differences in prevalence were observed for *Camallanus* spp., *Cephalochlamys* sp., *Polystoma* sp. and digeneans (*incertae sedis*). No significant spatial variation was detected for *Contracaecum* sp. in *X. victorinus*.

Table 4. 2

*Chi-square (χ^2) test results for spatial variation in parasite taxa prevalence in *X. borealis**

*and *X. victorinus* across eight Lake Victoria Basin sites ($df = 7$, $\alpha = 0.05$).*

Host species	Parasite taxon	Chi-Square	<i>p</i> -value	Significance
<i>X. borealis</i>	<i>Contracaecum</i> sp.	16.304	0.022	*
	<i>Camallanus</i> spp.	16.807	0.018	*
	<i>Cephalochlamys</i> sp.	21.451	0.003	*
	<i>Polystoma</i> sp.	19.617	0.006	*
	Digenea (<i>incertae sedis</i>)	3.7375	0.809	
<i>X. victorinus</i>	<i>Contracaecum</i> sp	7.1832	0.410	
	<i>Camallanus</i> spp.	18.213	0.011	*
	<i>Cephalochlamys</i> sp.	25.291	0.001	*
	<i>Polystoma</i> sp.	44.762	0.000	*
	Digenea (<i>incertae sedis</i>)	17.289	0.015	*

* Significant difference.

Chi-square analyses further demonstrated variation in parasite prevalence across the three sampling periods for both host species (Table 4.3). In *X. borealis*, significant temporal differences were observed for *Contracaecum* sp. and digeneans (*incertae sedis*). No significant temporal variation was detected for *Camallanus* spp., *Cephalochlamys* sp. or *Polystoma* sp. In *X. victorinus*, significant temporal variation in prevalence was detected for *Contracaecum* sp., *Camallanus* spp., *Polystoma* sp, and digeneans (*incertae sedis*). Conversely, *Cephalochlamys* sp. did not exhibit significant temporal variation in prevalence.

Table 4. 3

*Chi-square (χ^2) test results for temporal variation in parasite taxa prevalence in *X. borealis**

*and *X. victorinus* across three sampling periods in Lake Victoria Basin sites ($df = 2$, $\alpha = 0.05$).*

Host species	Parasite taxon	Chi-Square	p-value	Significance
<i>X. borealis</i>	<i>Contracaecum</i> sp.	42.116	0.000	*
	<i>Camallanus</i> spp.	0.3146	0.854	
	<i>Cephalochlamys</i> sp.	1.4334	0.488	
	<i>Polystoma</i> sp.	3.339	0.188	
	Digenea (<i>incertae sedis</i>)	27.264	0.000	*
<i>X. victorinus</i>	<i>Contracaecum</i> sp.	33.164	0.000	*
	<i>Camallanus</i> spp.	17.587	0.000	*
	<i>Cephalochlamys</i> sp.	0.1032	0.949	
	<i>Polystoma</i> sp.	11.548	0.003	*
	Digenea (<i>incertae sedis</i>)	17.143	0.000	*

* Significant difference.

4.3.6.2 ANOVA test

There were no spatial differences in mean infection intensity detected, except for *Polysoma* sp. that showed both spatial and temporal variation in the mean infection intensity ($F_{7,14} = 3.093$, $p < 0.05$) and *Contracaecum* sp. in *X. borealis* ($F_{2,14} = 5.563$, $p < 0.05$) and *Camallanus* spp. in *X. victorinus* ($F_{2,14} = 3.799$, $p < 0.05$) and *Camallanus* spp. in *X. victorinus* ($F_{2,14} = 14.859$, $p < 0.001$) that had temporal variations.

4.3.7 Statistical power of infection detection

Standardized sample sizes of 30 individuals per county enabled assessment of the study's ability to detect spatial variation in parasite prevalence for each parasite taxon across the two *Xenopus* species (Table 4.4). Observed prevalence differences exceeded the corresponding MDE thresholds for *Contracaecum* sp., *Camallanus* spp., and *Cephalochlamys* sp., indicating sufficient statistical power to detect spatial differences in infection prevalence for these taxa. In contrast, OPD values for polystomatids in *X. borealis* and digeneans (*incertae sedis*) fell below the estimated MDEs, suggesting limited power to reliably detect differences in prevalence for these parasite groups.

For *X. victorinus*, observed prevalence differences were generally higher. Observed prevalence differences exceeded MDE thresholds for four of the five parasite taxa, including *Contracaecum* sp., *Camallanus* spp., *Cephalochlamys* sp., and *Polystoma* sp. in *X. victorinus*, indicating adequate statistical power for detecting spatial variation in these infections. However, for digeneans (*incertae sedis*), the observed prevalence difference (43.3 %) was marginally below the estimated MDE (44.9 %), indicating insufficient power to conclusively detect spatial differences for this taxon despite relatively large observed variation.

Table 4. 4

Minimum detectable effect (MDE) analyses for three taxa of parasites infecting *X. borealis* and *X. victorinus* in LVB. Sample sizes (n) = 30; $Z_{1-\alpha/2} =$

1.96; $Z_{power} = 0.842$ for the power of 80%.

Host	Parasite taxon	Prevalence range %	Observed Prevalence Difference (OPD) %	Pooled prevalence	Standard Error (SE)	MDE (<i>Post-hoc</i>) %	Detection (OPD > MDE)
<i>X. borealis</i>	<i>Contracaecum</i> sp.	46.7 – 73.3	26.7	0.60	0.092	25.7	Yes
	<i>Camallanus</i> spp.	63.3 – 93.3	30.0	0.78	0.067	18.8	Yes
	<i>Cephalochlamys</i> sp.	36.7 – 76.7	40.0	0.57	0.099	27.9	Yes
	<i>Polystoma</i> sp.	6.7 – 13.3	6.7	0.10	0.142	39.9	No
	Digenea (<i>incertae sedis</i>)	36.7 – 56.7	20	0.46	0.106	29.6	No
<i>X. victorinus</i>	<i>Contracaecum</i> sp.	30.0 – 56.7	26.7	0.433	0.112	31.3	Yes
	<i>Camallanus</i> spp.	50.0 – 86.7	36.7	0.683	0.083	23.1	Yes
	<i>Cephalochlamys</i> sp.	23.3 – 73.3	50.0	0.483	0.119	33.3	Yes
	<i>Polystoma</i> sp.	16.7– 63.3	46.7	0.40	0.135	37.78	Yes
	Digenea (<i>incertae sedis</i>)	10.0 – 53.3	43.3	0.32	0.160	44.91	No

4.3.8 Diversity indices

Diversity indices of helminth taxa collected in hosts from different sampling sites within the LVB (Table 4.5) and across different hydrological periods (Table 4.6) are presented below.

Table 4. 5

Diversity indices of helminth taxa in Xenopus borealis and Xenopus victorianus from the Lake Victoria Basin, Kenya. The different sampling locations/ points are coded as KK (Kakamega), VH (Vihiga), SY (Siaya), KS (Kisumu), HB (Homa Bay), MG (Migori), KI (Kisii), BS (Busia).

	Host	Diversity Parameters	Location							
			KK	VH	SY	KS	HB	MG	KI	BS
<i>X. borealis</i>		Number of frogs (n)	30	30	30	30	30	30	30	30
		Number of parasite taxa	4	4	4	4	4	4	4	4
		Number of parasites	305	188	180	174	265	280	272	234
		Dominant taxon <i>Camallanus</i> spp.	211	113	126	106	175	187	208	162
		Shannon-Weiner index	0.82	0.91	0.81	0.90	0.87	0.84	0.71	0.79
		Margalef richness index	0.35	0.38	0.38	0.39	0.36	0.35	0.36	0.37
		Berger-Parker dominance	0.69	0.60	0.70	0.61	0.66	0.67	0.76	0.70
		Simpson's index	0.53	0.45	0.54	0.45	0.50	0.51	0.61	0.54

<i>X. victorinus</i>	Number of frogs (n)	30	30	30	30	30	30	30	30	
	Number of parasite taxa	4	4	4	4	4	4	4		
	Number of parasites	154	178	120	200	140	194	151	186	
	Dominant taxon	<i>Camallanus</i> spp.	118	129	64	137	118	156	92	149
	Shannon-Weiner index	0.70	0.80	1.01	0.84	0.54	0.62	0.94	0.63	
	Margalef richness index	0.40	0.40	0.41	0.40	0.40	0.40	0.40	0.40	
	Berger-Parker dominance	0.80	0.72	0.53	0.70	0.84	0.80	0.61	0.80	
	Simpson's index	0.61	0.60	0.40	0.52	0.72	0.70	0.44	0.70	

Camallanus spp. dominated the parasite ¹¹communities at all locations and in both host species. The Shannon-Wiener diversity index (H') ranged from 0.71 to 0.91 in *X. borealis* and from 0.62 to 1.01 in *X. victorinus*. The Margalef richness index (R) ranged from 0.35 to 0.41 in both host species, indicating low species richness relative to the total number of parasites observed. Berger-Parker dominance index (BP) ranged from 0.60 to 0.70 in *X. borealis* and from 0.53 to 0.84 in *X. victorinus*, with higher dominance values generally observed in *X. victorinus*. These results confirm that a single parasite taxon dominated the parasitic assemblages. The Simpson's dominance index (D) further supported these findings, with values ranging from 0.45 to 0.61 in *X. borealis* and from 0.40 to 0.72 in *X. victorinus*.

Temporal patterns mirrored spatial trends, with parasite counts ranging from 326 to 750 across the hydrological periods (Table 4.6). Parasitic communities exhibited low diversity throughout the wet, moderate, and dry seasons. In *X. borealis*, the Shannon-Wiener diversity index (H') remained relatively constant, ranging from 0.67 to 0.87. In contrast,

X. victorinus displayed notable seasonal variation, with H' peaking at 1.02 during the wet season and dropping to 0.41 in the dry season. Correspondingly, both the Berger-Parker dominance index (0.65–0.78 in *X. borealis*; 0.51–0.89 in *X. victorinus*) and the Simpson's dominance index (0.49–0.64 in *X. borealis*; 0.38–0.80 in *X. victorinus*) increased from the wet to the dry season, indicating a rise in dominance by fewer parasite taxa during drier periods.

Table 4. 6

Comparisons of diversity indices for various helminth taxa in Xenopus borealis and Xenopus victorinus across different hydrological periods from counties within the Lake Victoria Basin, Kenya.

	Host	Diversity Parameters	Season		
			Wet	Moderate	Dry
<i>X. borealis</i>		Number of frogs (n)	80	80	80
		Number of parasite taxa	4	4	4
		Number of parasites	750	693	455
		Dominant taxon	Camallanus spp.	447	357
		Shannon-Weiner index	0.87	0.87	0.67
		Margalef richness index	0.30	0.31	0.33
		Berger-Parker dominance	0.65	0.65	0.78
		Simpson's index	0.49	0.49	0.64

<i>X. victorinus</i>	Number of frogs (n)		80	80	80
	Number of parasite taxa		4	4	4
	Number of parasites		326	372	625
	Dominant taxon	Camallanus spp.	165	240	558
	Shannon-Weiner index		1.02	0.90	0.41
	Margalef richness index		0.35	0.34	0.31
	Berger-Parker dominance		0.51	0.65	0.89
	Simpson's index		0.38	0.48	0.80

Adequate sample size is essential for reliable estimation of parasite infection indices. Guidelines from the World Organisation for Animal Health (OIE), consistent with Cannon and Roe (1982), indicate that sample sizes of 150 individuals detect low prevalence (~2%), with smaller samples sufficient at higher prevalence levels. These thresholds are supported by Shvydka et al. (2018) and de Blas et al. (2020), who showed that samples exceeding 25 hosts substantially reduce bias. In this study, 240 individuals per host species provided robust estimates of prevalence and mean intensity. Despite ethical constraints, post-hoc minimum detectable effect (MDE) analysis confirmed sufficient statistical power ($\alpha = 0.05$; $1 - \beta = 0.80$) to detect meaningful spatial differences.

Overall, no major spatial or temporal differences in parasite infection indices were observed between *X. borealis* and *X. victorinus*, although some taxa showed localized variation. In *X. borealis*, observed prevalence differences for polystomatids and digeneans (*incertae sedis*) fell below MDE thresholds, likely reflecting low prevalence and aggregated distributions (Gourbière et al., 2015; Morrill et al., 2022). In contrast, polystomatids in *X. victorinus* exhibited stronger spatial structuring, suggesting host-specific differences in parasite establishment and transmission.

Parasite assemblages in both hosts were dominated by camallanid nematodes, which showed higher prevalence and mean intensity than *Contracaecum* sp. and *Cephalochlamys* sp., consistent with findings in related pipid hosts (Mbokane et al., 2020). This dominance indicates high transmission efficiency and ecological tolerance. Notably, camallanid prevalence increased markedly in *X. victorinus* during the dry season, while remaining stable in *X. borealis*. This pattern likely reflects increased host aggregation in shrinking water bodies and possible seasonal variation in host immunity.

In contrast, the lower prevalence of *Contracaecum* sp. is consistent with amphibians acting as paratenic hosts, limiting parasite development.

Spatial clustering of higher infection levels in Kakamega, Kisumu, and Kisii suggests that habitat heterogeneity and biodiversity influence transmission dynamics (Fearon et al., 2023; Solórzano-García et al., 2023). Anthropogenic factors, including agricultural expansion and pollution, may further modify these patterns by affecting environmental conditions and intermediate host availability. Environmental variation across the Lake Victoria Basin likely underlies much of the observed parasite distribution. The presence of digeneans and cestodes indicates active trophic transmission involving intermediate hosts, with higher prevalence in sites that favour invertebrate abundance (Cribb et al., 2003; Jackson & Tinsley, 2001). In contrast, nematodes with direct life cycles are more strongly influenced by host behaviour and environmental exposure, including substrate use, host density, and moisture conditions (Anderson, 2000).

Host-specific differences further structure parasite communities. In *X. borealis*, parasite diversity remained relatively stable, whereas *X. victorinus* showed reduced diversity and increased camallanid dominance during the dry season, suggesting decreased community evenness and possible competitive exclusion. Polyopisthocotyleans, particularly polystomatids, exhibited patterns consistent with strong host specificity and co-evolutionary relationships (Poulin, 2007).

Although temporal effects were less pronounced, seasonal changes in rainfall, temperature, and host reproduction likely interact with spatial factors to influence parasite transmission. Overall, the parasite assemblage comprising nematodes, polyopisthocotylean, cestodes, and digeneans reflects a typical, ecologically structured

helminth community in freshwater amphibians (Smyth & Halton, 1983). While broad spatial and temporal differences were limited, variation among taxa and host species underscores the combined influence of host ecology, environmental heterogeneity, and parasite life-history strategies. Importantly, MDE analysis confirms that these patterns are robust.

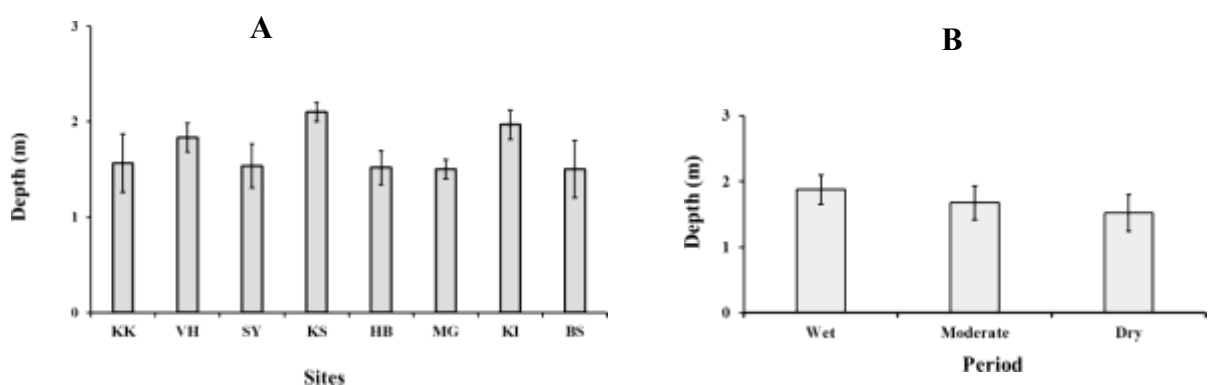
4.4 To assess the spatio-temporal variation in water quality of aquatic habitats of *X. borealis* and *X. victorianus* and its influence on parasite infection and diversity indices in the Lake Victoria Basin, Kenya

4.4.1 Physico-chemical water quality parameters of host habitats

The study areas were distinctively characterized by remarkable variations in the physico-chemical parameters investigated. In the sampled ponds and wetlands, water depths (Figs. 4.18 A, B) ranging from 1.3 m to 2.2 m, with an overall mean of 1.69 ± 0.29 m. Sampling points in Kisumu, Vihiga, and Kisii counties had relatively deeper water bodies, sustained by prolonged wet spells. These counties received comparatively high rainfall, influenced by their altitudes, sparsely distributed naturalized forestland, and the water retention capacity of local soils. During the dry period, all sampling sites experienced a notable decline in water levels. Spatial and temporal variations in water depth across the sites and sampling periods were statistically significant ($F_{7, 14} = 15.451$, $p = 0.001$; $F_{2, 14} = 22.586$, $p = 0.001$).

Figure 4. 18

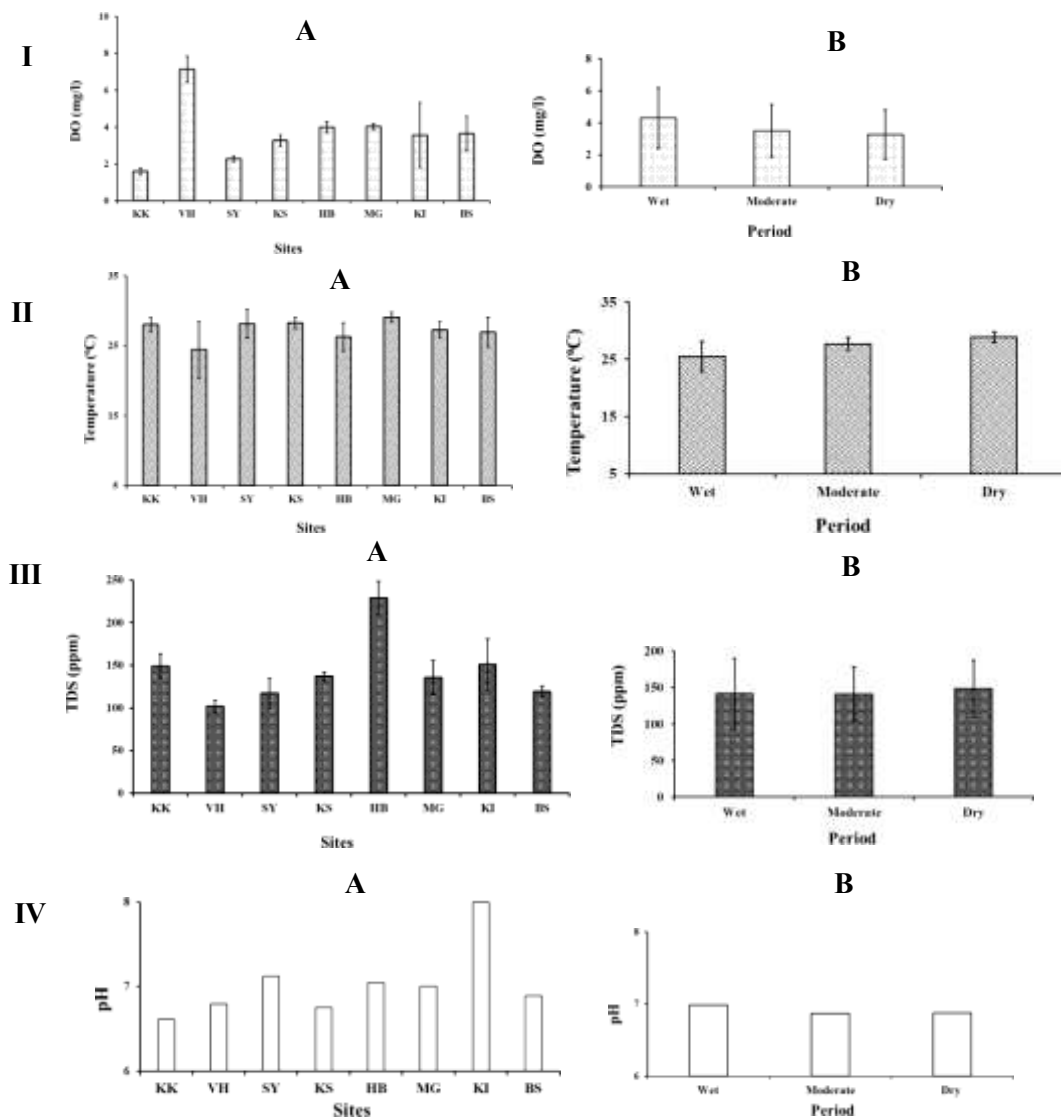
Variation in water depths (mean $\pm$ SE) across (A) sampling sites ($n = 8$) and (B) hydrological periods ($n = 3$) in eight counties within Lake Victoria Basin, Kenya.



The four physico-chemical parameters, DO, temperature, TDS, and pH, varied across sampling sites and over the sampling period (Fig.4.19). Dissolved oxygen concentrations ranged from 1.4 to 7.9 mg/L.¹²

Figure 4. 19

Variation in physico-chemical parameters (mean $\pm$ SE) across sampling sites (n= 8) and hydrological periods (n=3) in eight counties within Lake Victoria Basin, Kenya



¹² Kakamega (KK), Vihiga (VH), Siaya (SY), Kisumu (KS), Homa Bay (HB), Migori (MG), Kisii (KI), Busia (BS)

Two-way ANOVA revealed statistically significant spatial and temporal differences in DO levels, with a significant effect of location ($F_{7, 14} = 22.805$, $p < 0.001$) and sampling period ($F_{2, 14} = 6.515$, $p < 0.001$). Water temperature ranged from 19.8 to 29.1 °C and was significantly influenced by both location ($F_{7, 14} = 4.770$, $p < 0.001$) and sampling period ($F_{2, 14} = 17.447$, $p < 0.001$). pH values ranged from 6.36 to 8.05, showing a significant spatial variation across sites ($F_{7, 14} = 12.596$, $p < 0.001$) but no significant change over time. Similarly, TDS values ranged from 96.5 to 251.6 ppm, with significant differences observed among sites, but not across sampling periods.

4.4.2 Association of water quality parameters with parasite infection indices

Spearman's correlation analysis (Table 4.7) revealed a significant negative relationship between DO levels and the prevalence of two parasite taxa (*Contracaecum* sp. and *Camallanus* spp.). In addition, *Camallanus* spp. showed a significant negative correlation with pH variation across the sampled sites. By contrast, the prevalence of cestodes (*Cephalochlamys* sp.) showed no significant correlation with any water quality parameter.

Table 4. 7

*Spearman's correlation between physicochemical parameters and parasite taxa prevalence in *X. borealis* and *X. victorianus* across eight Lake Victoria Basin sites; significant correlations in bold; n =16)*

Parasite taxon	Spearman's rho (ρ) and <i>p</i> -values (in parenthesis)				
	Depth	Temperature	DO	TDS	pH
<i>Contracaecum</i> sp.	0.158(0.559)	0.353(0.179)	-0.748(0.001)	0.311(0.241)	0.242(0.366)
<i>Camallanus</i> spp.	0.360(0.170)	-0.113(0.675)	-0.525(0.036)	0.035(0.895)	-0.507(0.045)
<i>Cephalochlamys</i> sp.	- 0.271(0.309)	0.327(0.215)	0.137(0.613)	0.393(0.132)	0.012(0.965)
<i>Polystoma</i> sp.	0.288(0.280)	0.067(0.806)	0.286(0.283)	-0.614(0.011)	- 0.313(0.238)
Digenea (<i>incertae sedis</i>)	- 0.052(0.847)	-0.524(0.037)	0.036(0.896)	0.161(0.552)	0.354(0.178)

Variation in water quality parameters played a significant role in shaping parasite infection patterns. Among the variables measured, dissolved oxygen (DO), pH, total dissolved solids (TDS), and temperature were most strongly associated with parasite prevalence.

The observed negative relationship between DO and parasite prevalence suggests that low oxygen conditions may increase host susceptibility to infection. This is further supported by Ageng'o et al. (2024) who found that dissolved oxygen levels could interfere directly with host immune function, increasing vulnerability to infection. This may be explained by increased physiological stress and energy allocation to respiration, reducing immune efficiency (De Padova et al., 2024). Conversely, higher DO levels may limit parasite establishment, consistent with previous findings (Korma, 2022). The

patterns observed in *Xenopus* across the Lake Victoria Basin echo these findings and suggest that pH-mediated immune modulation may be a cross-taxon phenomenon in East African freshwater ecosystems.

Temperature, although a key driver of parasite development and transmission (Schampera et al., 2022), showed limited variation across sites and was only significantly associated with digenean infections. This is consistent with the temperature-dependent life cycles of digeneans, where warmer conditions accelerate development and transmission (McCarthy, 1999). However, the relatively narrow temperature range observed in this study may have constrained its overall influence.

Although water depth did not show a direct correlation with infection levels, it likely exerts indirect effects by influencing other environmental parameters such as oxygen availability, light penetration, and nutrient dynamics (Budria, 2017). Seasonal reductions in water levels may also promote host aggregation, thereby increasing transmission opportunities.

It is important to note that while significant associations between water quality parameters and parasite prevalence were observed, the role of host-specific factors, including immune responses, behaviour, and physiological compatibility, was not explicitly tested. These factors are known to influence parasite distribution and may interact with environmental variables to shape infection patterns (Poulin, 2007).

CHAPTER FIVE

5.0 CONCLUSIONS AND RECOMMENDATIONS

5.1 Conclusions

From this study, the following conclusions were made:

1. A diverse assemblage of helminth parasites, including nematodes, polyopisthocotyleans, cestodes, and digeneans, was recorded. This shows the ecological complexity of the system and the presence of both direct and indirect transmission pathways. The coexistence of host-specific taxa, polyopisthocotyleans, alongside generalist parasites such as camallanid nematodes highlights the diversity of parasite life-history strategies within these amphibian hosts.
2. Parasite infection patterns were generally stable across space and time, with no pronounced large-scale variation in prevalence and mean intensity. However, finer-scale differences were evident among parasite taxa and between host species. Camallanid nematodes dominated parasite assemblages in both hosts, with a marked seasonal increase in *X. victorinus* during the dry period. In contrast, taxa such as *Contracaecum* spp. and polyopisthocotyleans exhibited lower infection levels, likely reflecting life cycle constraints. These patterns emphasise the role of host ecology, behaviour, and compatibility in structuring parasite communities.
3. Environmental conditions played a key role in shaping infection dynamics. Variation in dissolved oxygen, temperature, pH, and total

dissolved solids was associated with differences in parasite prevalence across sites. Although water depth did not directly influence infection indices, it likely acted indirectly by affecting environmental conditions and host aggregation during periods of reduced water availability. The negative relationship between dissolved oxygen and parasite prevalence suggests that environmental stress may increase host susceptibility, while the temperature sensitivity observed in digenean infections highlights the importance of thermal conditions in parasite development and transmission.

4. Overall, parasite distribution in *Xenopus* species within the Lake Victoria Basin is shaped by a complex interaction of parasite life-history traits, host-specific factors, and environmental variability. While broad-scale patterns appear relatively stable, fine-scale spatial and temporal processes are critical in determining infection dynamics. This study provides important baseline data on amphibian parasite ecology in Kenya and contributes to a broader understanding of host–parasite interactions in freshwater ecosystems. Future research should further investigate intermediate hosts, seasonal dynamics, and environmental change to better understand the mechanisms driving parasite transmission and diversity.

5.2 Recommendations

Based on the findings of this study, several recommendations are proposed to advance research, conservation, and management of amphibian parasite systems within the Lake Victoria Basin. These are as follows:

5. There is a need to explicitly investigate the role of intermediate hosts in parasite transmission. The presence of digeneans and cestodes indicates active trophic pathways; however, the specific intermediate host communities and their distribution remain poorly understood. Targeted studies on aquatic invertebrates, particularly molluscs and arthropods, would provide critical insights into parasite life cycles and transmission pathways.
6. Future studies should incorporate long-term and high-resolution temporal sampling to better capture seasonal dynamics of parasite transmission. Although this study identified limited broad-scale temporal variation, the observed seasonal increase in camallanid infections suggests that finer-scale temporal processes, particularly those linked to rainfall patterns and hydroperiod changes, play an important role in shaping infection dynamics.
7. Third, future research should integrate host-specific factors, including immune responses, behaviour, and habitat use, to better understand differences in parasite assemblages between *X. borealis* and *X. victorinus*. Experimental and comparative studies examining host and parasite compatibility would help clarify the mechanisms underlying observed differences in infection patterns.
8. Regular monitoring of physico-chemical parameters should be conducted in aquatic habitats within the Lake Victoria Basin, as these parameters were significantly associated with parasite prevalence. Such monitoring will help detect

environmental changes that may influence parasite transmission and anuran health.

9. The baseline data generated in this study should be expanded through broader geographic and taxonomic surveys, including additional amphibian species and habitats within and beyond the Lake Victoria Basin. Such efforts would enhance regional parasite inventories and contribute to a more comprehensive understanding of amphibian health and biodiversity.

REFERENCES

- Ageng'o, F. O., Waruiru, R. M., Wanja, D. W., Nyaga, P. N., Hamisi, M. M., Khasake, C. N., Wainaina, J. M., & Munde, B. M. (2024). Relationship between water quality parameters and parasite infestation in farmed *Oreochromis niloticus* in selected Rift Valley counties, Kenya. *Aquaculture Research*, Article 6139798. <https://doi.org/10.1155/2024/6139798>
- Ahmed, B. R., Romdhane, Y., & Tekaya, S. (2015). Checklist and distribution of marine and freshwater leeches (Annelida, Clitellata, Hirudinea) in Tunisia with identification key. *Ecologica Montenegrina*, 2(1), 3–19. <https://doi.org/10.37828/em.2015.2.1>
- Aisien, M. S. O., Edo-Taiwo, O., & Imasuen, A. A. (2021). Ecological scenarios and parasite diversity in anurans of West Africa: A review. *Diversity*, 13(6), 1–19. <https://doi.org/10.3390/d13060223>
- Alford, R. A., Cohen, M. P., & Crossland, M. R. (2009). Global amphibian declines: A problem in applied ecology. *Annual Review of Ecology, Evolution, and Systematics*, 40, 133–165. <https://doi.org/10.1146/annurev.ecolsys.110308.120235>
- Alroy, J. (2015). Current extinction rates of reptiles and amphibians. *Proceedings of the National Academy of Sciences*, 112(42), 13003–13008. <https://doi.org/10.1073/pnas.1508681112>
- Amin, O. M. (1987). Key to the families and subfamilies of Acanthocephala, with the erection of a new class (Polyacanthocephala) and a new order (Polyacanthorhynchida). *Journal of Parasitology*, 73, 1216–1219.
- Amin, O. M. (2013). Classification of the Acanthocephala. *Folia Parasitologica*, 60, 273–305. <https://doi.org/10.14411/fp.2013.031>
- Anderson, R. C. (2000). *Nematode parasites of vertebrates: Their development and transmission*

(2nd ed., pp. 1–16). CABI. <https://doi.org/10.1079/9780851994215.0001>

Attademo, A. M., Peltzer, P. M., Lajmanovich, R. C., Junges, C., Bassó, A., & Cabagna-Zenklusen, M. (2012). Trombiculid mites (*Hannemania* sp.) in *Leptodactylus chaquensis* (Amphibia: Anura) inhabiting selected soybean and rice agroecosystems of Argentina. *Journal of Zoo and Wildlife Medicine*, 43(3), 579–584.

Badets, M., Whittington, I. D., Lalubin, F., Allienne, J. F., Miquel, J., & Verneau, O. (2013). Alternative development in *Polystoma gallieni* (Platyhelminthes, Monogenea) and life cycle evolution. *Experimental Parasitology*, 135(3), 537–546.

<https://doi.org/10.1016/j.exppara.2013.09.002>

Benício, R. A., dos Santos, R. S., Freire, S. M., Ávila, R. W., Da Silva, R. J., & Fonseca, M. G. (2022). Diversity of helminth parasites in amphibians from northeastern Brazil. *Biologia*, 77, 1–9. <https://doi.org/10.1007/s11756-022-01132-5>

Bentz, S., Leroy, S., du Preez, L., Mariaux, J., Vaucher, C., & Verneau, O. (2001). Origin and evolution of African *Polystoma* (Monogenea: Polystomatidae) assessed by molecular methods. *International Journal for Parasitology*, 31(7), 697–705. [https://doi.org/10.1016/S0020-7519\(01\)00179-5](https://doi.org/10.1016/S0020-7519(01)00179-5)

Biggs, J., Challenger, J. D., Hellewell, J., Churcher, T. S., & Cook, J. (2024). A systematic review of sample size estimation accuracy on power in malaria cluster randomized trials measuring epidemiological outcomes. *BMC Medical Research Methodology*, 24, 238. <https://doi.org/10.1186/s12874-024-02361-9>

Blaustein, A. R., & Kiesecker, J. M. (2002). Complexity in conservation: Lessons from the global decline of amphibian populations. *Ecology Letters*, 5(4), 597–608. <https://doi.org/10.1046/j.1461-0248.2002.00352.x>

- Brennan, G. P., Hanna, R. E. B., & Nizami, W. A. (1991). Ultrastructural and histochemical observations on the tegument of *Gastrodiscoides hominis* (Paramphistoma: Digenea). *International Journal for Parasitology*, 21(8), 897–905. [https://doi.org/10.1016/0020-7519\(91\)90164-3](https://doi.org/10.1016/0020-7519(91)90164-3)
- Budria, A. (2017). Beyond troubled waters: The influence of eutrophication on host–parasite interactions. *Functional Ecology*, 31(7), 1348–1358. <https://doi.org/10.1111/1365-2435.12880>
- Bullard, S. A., Overstreet, R. M., & Carlson, J. S. (2003). Host specificity of monogeneans (Platyhelminthes) parasitizing sciaenid fishes in the eastern Gulf of Mexico. *Journal of Parasitology*, 89(2), 184–197.
- Bush, A. O., Lafferty, K. D., Lotz, J. M., & Shostak, A. W. (1997). Parasitology meets ecology on its own terms: Margolis et al. revisited. *Journal of Parasitology*, 83, 575–583. <https://doi.org/10.2307/3284225>
- Buxton, V., & Sperry, J. (2016). Reproductive decisions in anurans: A review of how predation and competition affects the deposition of eggs and tadpoles. *BioScience*, 67. <https://doi.org/10.1093/biosci/biw149>
- Cannon, R. M., & Roe, R. T. (1982). *Livestock disease surveys: A field manual for veterinarians*. Australian Government Publishing Service.
- Casado, N., Moreno, M. J., Urrea-París, M. A., & Rodriguez-Caabeiro, F. (1999). Ultrastructural study of the papillae and presumed sensory receptors in the scolex of the *Gymnorhynchus gigas* plerocercoid (Cestoda: Trypanorhyncha). *Parasitology Research*, 85(12), 964–973. <https://doi.org/10.1007/s004360050667>
- Chaabane, A., Verneau, O., & Du Preez, L. (2019). *Indopolystoma* n. gen. (Monogenea, Polystomatidae) with the description of three new species and reassignment of eight known

Polystoma species from Asian frogs (Anura, Rhacophoridae). *Parasite*, 26, 67.

<https://doi.org/10.1051/parasite/2019067>

Channing, A., & Howell, K. M. (2006). *Amphibians of East Africa*. Cornell University Press.

Cone, D. K. (1995). Monogenea (phylum Platyhelminthes). In P. T. K. Woo (Ed.), *Diseases and disorders of finfish in cage culture: Protozoan and metazoan infections* (Vol. 1, pp. 289–327). CABI International Publishing.

Corsin, F., Georgiadis, K., Hammell, L., & Hill, B. (2009). *Guide for aquatic animal health surveillance* (113 pp.). World Animal Health Organisation (OIE).

Cribb, T. H., Barton, D. P., Blair, D., Bott, N. J., Bray, R. A., Corner, R. D., Cutmore, S. C., De Silva, M. L. I., Duong, B., Faltýnková, A., Gonchar, A., Hechinger, R. F., Herrmann, K. K., Huston, D. C., Johnson, P. T. J., Kremnev, G., Kuchta, R., Louvard, C., Luus-Powell, W. J., ... Chisholm, L. A. (2002). Diversity in the monogenean and digenea: Does lifestyle matter? *International Journal of Parasitology*, 32, 321–328.

Cribb, T. H., Barton, D. P., Blair, D., Bott, N. J., Bray, R. A., Corner, D., Cutmore, S. C., De Silva, M., Duong, B., Faltynkova, A., Gonchar, A., Hechinger, R. F., Herrmann, K. K., Huston, D. C., Johnson, P. T. J., Kremnev, G., Kuchta, R., Louvard, C., Luus-Powell, W. J., & Achatz, T. J. (2025). Challenges in the recognition of trematode species: Consideration of hypotheses in an inexact science. *Journal of Helminthology*, 99, e54. <https://doi.org/10.1017/S0022149X25000367>

Cribb, T. H., Bray, R. A., Olson, P. D., & Littlewood, D. T. (2003). Life cycle evolution in the digenea: A new perspective from phylogeny. *Advances in Parasitology*, 54, 197–254. [https://doi.org/10.1016/s0065-308x\(03\)54004-0](https://doi.org/10.1016/s0065-308x(03)54004-0)

Crump, M. L. (2015). Anuran reproductive modes: Evolving perspectives. *Journal of Herpetology*,

49, 1–16. <https://doi.org/10.1670/14-097>

Da Silva, I. C. O., Pacheco, E. O., Da Silva, L. A., Carvalho, P. S., Santana, D. J., & Tavares, L. E. R. (2018). Metazoan parasites of *Odontophrynus americanus* (Anura: Odontophrynidae) from the Serra da Bodoquena Mountain range, Mato Grosso do Sul, Brazil. *Herpetology Notes*, 11, 343–347.

Dalton, J., Skelly, P., & Halton, D. (2004). Role of the tegument and gut in nutrient uptake by parasitic platyhelminths. *Canadian Journal of Zoology*, 82(2), 211–232. <https://doi.org/10.1139/z03-213>

De Blas, I., Muniesa, A., Vallejo, I. A., & Ruiz-Zarzuela, I. (2020). Assessment of sample size calculations used in aquaculture by simulation techniques. *Frontiers in Veterinary Science*, 7, 253. <https://doi.org/10.3389/fvets.2020.00253>

De Padova, J., Anderson, N. K., Halbauer, R., & Fuxjager, M. J. (2024). Acute hypoxia exposure rapidly triggers behavioral changes linked to cutaneous gas exchange in Lake Titicaca frogs. *Behavioural Processes*, 219, 105047. <https://doi.org/10.1016/j.beproc.2024.105047>

Dos Santos, Q. M., & Avenant-Oldewage, A. (2015). Soft tissue digestion of *Paradiplozoon vaalense* for SEM of sclerites and simultaneous molecular analysis. *Journal of Parasitology*, 101, 94–97.

Du Preez, L. H., & Carruthers, V. C. (2009). *A complete guide to the frogs of southern Africa*. Struik Nature.

Du Preez, L. H., & Theunissen, M. (2021). A sucker for the job: Morphology and functioning of suckers of polystomatid monogeneans. *Folia Parasitologica*, 68, 006. <https://doi.org/10.14411/fp.2021.006>

Du Preez, L. H., Landman, W. J., & Verneau, O. (2025). *Polystomatid flatworms: State of knowledge and future trends*. Springer.

- Duellman, W. E., & Trueb, L. (1994). *Biology of amphibians*. Johns Hopkins University Press.
- Fabrezi, M., Goldberg, J., & Chuliver Pereyra, M. (2017). Morphological variation in anuran limbs: Constraints and novelties. *Journal of Experimental Zoology Part B: Molecular and Developmental Evolution*, 328(6), 546–574. <https://doi.org/10.1002/jez.b.22753>
- Fabrezi, M., Manzano, A. S., Abdala, V., & Lobo, F. (2014). Anuran locomotion: Ontogeny and morphological variation of a distinctive set of muscles. *Evolutionary Biology*, 41(2), 308–326. <https://doi.org/10.1007/s11692-014-9270-y>
- Fearon, M. L., Wood, C. L., & Tibbetts, E. A. (2023). Habitat quality influences pollinator pathogen prevalence through both habitat–disease and biodiversity–disease pathways. *Ecology*, 104(2), e3933. <https://doi.org/10.1002/ecy.3933>
- Finlay, J. C., & Vredenburg, V. T. (2007). Introduced trout sever trophic connections in watersheds: Consequences for a declining amphibian. *Ecology*, 88(9), 2187–2198. <https://doi.org/10.1890/06-0344.1>
- Fleiss, J. L., Levin, B., & Paik, M. C. (2003). *Statistical methods for rates and proportions* (3rd ed., p. 800). John Wiley & Sons.
- Frost, D. R. (2021). *Amphibian species of the world: An online reference* (Version 6.1). American Museum of Natural History. <http://research.amnh.org/herpetology/amphibia/index.html>
- Frost, D. R. (2024). *Amphibian species of the world: An online reference* (Version 6.2). American Museum of Natural History. <https://amphibiansoftheworld.amnh.org/index.php>
- Furman, B. L., Bewick, A. J., Harrison, T. L., Greenbaum, E., Gvoždík, V., Kusamba, C., & Evans, B. J. (2015). Pan-African phylogeography of a model organism, the African clawed frog *Xenopus laevis*. *Molecular Ecology*, 24(5), 909–925.

- GBIF. (2024). *GBIF home page*. Retrieved September 10, 2024, from <https://www.gbif.org>
- Gibbons, L. M. (2009). Dioctophymatoidea. In *CABI* (pp. 3–34). CABI.
<https://doi.org/10.1079/9781845935719.0003>
- Gourbière, S., Morand, S., & Waxman, D. (2015). Fundamental factors determining the nature of parasite aggregation in hosts. *PLOS ONE*, *10*(2), e0116893.
- Gregory, R. D., & Blackburn, T. M. (1991). Parasite prevalence and host sample size. *Parasitology Today*, *7*(11), 316–318. [https://doi.org/10.1016/0169-4758\(91\)90269-T](https://doi.org/10.1016/0169-4758(91)90269-T)
- Hallinger, M. J., Taubert, A., & Hermosilla, C. (2020). Endoparasites infecting exotic captive amphibian pet and zoo animals (Anura, Caudata) in Germany. *Parasitology Research*, *119*(11), 3659–3673. <https://doi.org/10.1007/s00436-020-06876-0>
- Hammoud, C., Van Bocxlaer, B., Verschuren, D., Tumusiime, J., Albrecht, C., De Crop, W., Umba Tolo, C., & Huyse, T. (2025). Agricultural land use and ensuing eutrophication both shape parasitic trematode communities in rural African lakes. *Proceedings of the Royal Society B: Biological Sciences*, *292*(2048), 20250070. <https://doi.org/10.1098/rspb.2025.0070>
- Hocking, D. J., & Babbitt, K. J. (2014). Amphibian contributions to ecosystem services. *Herpetological Conservation and Biology*, *9*(1), 1–17.
- Jackson, J. A., & Tinsley, R. C. (2001). *Protopolystoma xenopodis* (Monogenea) primary and secondary infections in *Xenopus laevis*. *Parasitology*, *123*(5), 455–463.
- Jadwiga, G. (1991). *Marine fish pathology: An outline*. Cambridge University Press.
- Jovani, R., & Tella, J. L. (2006). Parasite prevalence and sample size: Misconceptions and solutions. *Trends in Parasitology*, *22*(5), 214–218. <https://doi.org/10.1016/j.pt.2006.02.011>

- Justine, J.-L. (1998). Non-monophyly of the monogeneans? *International Journal for Parasitology*, 28(10), 1653–1657.
- Kabanze, J., Kimanzi, J., Malonza, P., & Rutina, L. (2023). Anthropogenic effects of habitat modification on anuran species diversity in a swamp forest area, Kenya. *African Journal of Ecology*, 62, 1–20. <https://doi.org/10.1111/aje.13245>
- Khalifa, R. M. A., Mohamadain, H. S., & Karrar, Y. F. M. (2019). Redescription and systematic studies on *Procamallanus (Procamallanus) elatensis* Fusco & Overstreet, 199 from three Red Sea *Siganus* sp. fishes. *Journal of the Egyptian Society of Parasitology*, 49, 81–90.
- Khalil, L. F., Jones, A., & Bray, R. A. (1994). Order Cyclophyllidea (diagnosis and key to families). In L. F. Khalil, A. Jones, & R. A. Bray (Eds.), *Keys to the cestode parasites of vertebrates* (pp. 305–307). CAB International.
- Korma, E. W. (2022). *Impacts of environmental parameters on the infectivity of freshwater snail*. IntechOpen. <https://doi.org/10.5772/intechopen.99829>
- Koskei, A. (2023). *Breeding ecology and population status of Kenyan amphibians*. IntechOpen. <https://doi.org/10.5772/intechopen.109825>
- Landman, W., Verneau, O., Vences, M., & du Preez, L. (2023). *Metapolystoma ohlerianum* n. sp. (Monogenea: Polystomatidae) from *Aglyptodactylus madagascariensis* (Anura: Mantellidae). *Acta Parasitologica*, 68(2), 344–358. <https://doi.org/10.1007/s11686-023-00668-z>
- Liem, K. F., Bemis, W. E., Walker, W. F., & Grande, L. (2001). *Functional anatomy of the vertebrates: An evolutionary perspective*. Brooks/Cole.
- Madelaire, C. B., da Silva, R. J., & Gomes, F. R. (2013). Calling behavior and parasite intensity in treefrogs, *Hypsiboas prasinus*. *Journal of Herpetology*, 47(3), 450–455. <https://doi.org/10.1670/12->

- Mahmoud, E. B., Osman, K. A. E., & Hussein, A. M. O. (2006). Site adaptations of *Acanthogyrus (Acanthosentis) tilapiae*: Observations through light and scanning electron microscopy. *Journal of Veterinary Science*, 7(4), 339–342. <https://doi.org/10.4142/jvs.2006.7.4.339>
- Mair, M. M., Kattwinkel, M., Jakoby, O., & Hartig, F. (2020). The minimum detectable difference (MDD) concept for establishing trust in nonsignificant results: A critical review. *Environmental Toxicology and Chemistry*, 39(11), 2109–2123. <https://doi.org/10.1002/etc.4847>
- Maraganga, J. M., Rindoria, N. M., Morara, G. M., Kimani, V. N., van Wyk, C., Dumbo, J. C., Smit, N. J., & Luus-Powell, W. J. (2023). Tegumental topography and molecular characterisation of two trematodes (Platyhelminthes: Digenea) from *Clarias gariepinus* in Kenya. *International Journal for Parasitology: Parasites and Wildlife*, 23, 100897.
- Mayor, A., Ishengoma, D. S., Proctor, J. L., & Verity, R. (2023). Sampling for malaria molecular surveillance. *Trends in Parasitology*, 39(11), 954–968.
- Mbokane, E. M., Theron, J., & Luus-Powell, W. J. (2020). Diversity of parasites from *Xenopus laevis* (Amphibia: Pipidae) and their seasonal rate of infection in selected habitats in the Limpopo Province, South Africa. *Helminthologia*, 57(3), 252–267. <https://doi.org/10.2478/helm-2020-0027>
- McCarthy, A. M. (1999). The influence of temperature on the survival and infectivity of the cercariae of *Echinoparyphium recurvatum* (Digenea: Echinostomatidae). *Parasitology*, 118(4), 383–388. <https://doi.org/10.1017/S003118209900400X>
- Mollaret, I., Jamieson, B. G. M., Adlard, R. D., Hugall, A., Lecointre, G., Chombard, C., & Justine, J.-L. (1997). Phylogenetic analysis of the Monogenea and their relationships with Digenea and Eucestoda inferred from 28S rDNA sequences. *Molecular and Biochemical Parasitology*, 90(2), 433–

Moravec, F. (2019). *Parasitic nematodes of freshwater fishes of Africa*. Academia Praha.

Moravec, F., Justine, J.-L., & Rigby, M. C. (2006). Some camallanid nematodes from marine perciform fishes off New Caledonia. *Folia Parasitologica*, 53(3), 223–239.

<https://doi.org/10.14411/fp.2006.029>

Morrill, A., Nielsen, Ó. K., Skírnisson, K., & Forbes, M. R. (2022). Identifying sources of variation in parasite aggregation. *PeerJ*, 10, e13763.

Nachtigall, W. (1974). *Biological mechanisms of attachment: The comparative morphology and bioengineering of organs for linkage, suction, and adhesion*. Springer. <https://doi.org/10.1007/978-3-642-85775-1>

Nessi, A., Cioccarelli, S., Tremolada, P., Gariano, P., Grandinetti, M., Balestrieri, A., & Manenti, R. (2023). Environmental factors affecting amphibian communities in river basins of the southern Apennines. *Diversity*, 15(5), 625. <https://doi.org/10.3390/d15050625>

O'Rourke, D. P., & Rosenbaum, M. D. (2015). Biology and diseases of amphibians. In J. G. Fox, L. C. Anderson, G. M. Otto, K. R. Pritchett-Corning, & M. T. Whary (Eds.), *Laboratory animal medicine* (3rd ed., pp. 931–965). Academic Press. <https://doi.org/10.1016/B978-0-12-409527-4.00018-3>

Oda, F. H., Batista, V. G., Gambale, P. G., Mise, F. T., de Souza, F., Bellay, S., Ortega, J. C. G., & Takemoto, R. M. (2016). Anuran species richness, composition, and breeding habitat preferences: A comparison between forest remnants and agricultural landscapes in southern Brazil. *Zoological Studies*, 55, 34. <https://doi.org/10.6620/ZS.2016.55-34>

Olsen, L. S. (1952). Some nematodes parasitic in marine fishes. *Publications of the Institute of*

Marine Science, University of Texas, 11(2), 173–215.

Otachi, E. O. (2009). *Studies on occurrence of protozoan and helminth parasites in Nile Tilapia (Oreochromis niloticus L.) from Central and Eastern provinces, Kenya* [Master's thesis, Egerton University].

Panyarachun, B., Ngamniyom, A., Sobhon, P., & Anuracpreeda, P. (2013). Morphology and histology of the adult *Paramphistomum gracile* Fischöder, 1901. *Journal of Veterinary Science*, 14(4), 425–432. <https://doi.org/10.4142/jvs.2013.14.4.425>

Paperna, I., & Dzikowski, R. (2006). Digenea (Phylum Platyhelminthes). In *Fish diseases and disorders* (p. 345).

Parmesan, C., & Yohe, G. (2003). A globally coherent fingerprint of climate change impacts across natural systems. *Nature*, 421(6918), 37–42. <https://doi.org/10.1038/nature01286>

Phillips, A. J., Govedich, F. R., & Moser, W. E. (2020). Leeches in the extreme: Morphological, physiological, and behavioral adaptations to inhospitable habitats. *International Journal for Parasitology: Parasites and Wildlife*, 12, 318–325. <https://doi.org/10.1016/j.ijppaw.2020.09.003>

Pough, F. H., Janis, C. M., & Heiser, J. B. (2016). *Vertebrate life*. Pearson.

Poulin, R. (2007). *Evolutionary ecology of parasites* (2nd ed., p. 343). Princeton University Press. <https://doi.org/10.1515/9781400840809>

Preston, D., & Johnson, P. (2010). Ecological consequences of parasitism. *Nature Education Knowledge*, 3(10), 47.

Rindoria, N. M., Dos Santos, Q. M., & Avenant-Oldewage, A. (2020). Additional morphological features and molecular data of *Paracamallanus cyathopharynx* (Nematoda: Camallanidae) infecting *Clarias gariepinus* (Actinopterygii: Clariidae) in Kenya. *Journal of Parasitology*, 106(1), 157–166.

<https://doi.org/10.1645/19-124>

Rindoria, N. M., Dumbo, J. C., Olonde, F. O., Přikrylová, I., Smit, W. J., & Luus-Powell, W. J. (2025). Morphological and molecular characterisation of an intestinal trematode from the marbled lungfish *Protopterus aethiopicus* in Kenya reveals a new species of *Heterorchis* (Digenea: incertae sedis). *International Journal for Parasitology: Parasites and Wildlife*, 29, 101182.

<https://doi.org/10.1016/j.ijppaw.2025.101182>

Rindoria, N. M., Gichana, Z., Morara, G. N., van Wyk, C., Smit, W. J., Smit, N. J., & Luus-Powell, W. J. (2023). Scanning electron microscopy and first molecular data of two species of *Lamproglena* (Copepoda: Lernaeidae) from *Labeo victorianus* (Cyprinidae) and *Clarias gariepinus* (Clariidae) in Kenya. *Pathogens*, 12, 980.

Russell, C., & Downs, C. (2012). Effect of land use on anuran species composition in north-eastern KwaZulu-Natal, South Africa. *Applied Geography*, 35, 247–256.

<https://doi.org/10.1016/j.apgeog.2012.07.003>

Sarabeev, V., Shvydka, S., Lisitsyna, O., Oros, M., Miterpáková, M., & Ždímalová, M. (2025). The sample size matters: Evaluating minimum and reasonable values in prevalence studies. *International Journal for Parasitology*, 55(13), 683–693. <https://doi.org/10.1016/j.ijpara.2025.05.003>

Schampera, C., Agha, R., Manzi, F., & Wolinska, J. (2022). Parasites do not adapt to elevated temperature, as evidenced from experimental evolution of a phytoplankton–fungus system. *Biology Letters*, 18, Article 20210560. <https://doi.org/10.1098/rsbl.2021.0560>

Schell, S. C. (1970). *How to know the trematodes*. Wm. C. Brown Company.

Schivo, F., Bauni, V., Krug, P., & Quintana, R. (2019). Distribution and richness of amphibians under different climate change scenarios in a subtropical region of South America. *Applied Geography*,

103, 70–89. <https://doi.org/10.1016/j.apgeog.2019.01.003>

Schloegel, L. M., Picco, A. M., Kilpatrick, A. M., Davies, A. J., Hyatt, A. D., & Daszak, P. (2009). Magnitude of the US trade in amphibians and presence of *Batrachochytrium dendrobatidis* and ranavirus infection in imported North American bullfrogs (*Rana catesbeiana*). *Biological Conservation*, 142(7), 1420–1426. <https://doi.org/10.1016/j.biocon.2009.02.003>

Schoeman, A. L., Du Preez, L. H., Kmentová, N., & Vanhove, M. P. M. (2022). A monogenean parasite reveals the widespread translocation of the African clawed frog in its native range. *Journal of Applied Ecology*, 59(11), 2670–2687. <https://doi.org/10.1111/1365-2664.14271>

Schoeman, A. L., Joubert, T.-L., Du Preez, L. H., & Svitin, R. (2020). *Xenopus laevis* as UberXL for nematodes. *African Zoology*, 55, 1–18. <https://doi.org/10.1080/15627020.2019.1681295>

Scholz, T., & Horák, P. (2003). [Review of the book *Keys to the Trematoda, Volume I*, by D. I. Gibson, A. Jones, & R. A. Bray]. *Folia Parasitologica*, 50, 56. <https://doi.org/10.14411/fp.2003.010>

Scholz, T., Waeschenbach, A., Oros, M., Brabec, J., & Littlewood, D. T. J. (2021). Phylogenetic reconstruction of early diverging tapeworms (Cestoda: Caryophyllidea) reveals ancient radiations in vertebrate hosts and biogeographic regions. *International Journal for Parasitology*, 51, 263–277. <https://doi.org/10.1016/j.ijpara.2020.09.009>

Shigoley, M. I., Kmentová, N., Ndegwa, D. M., Topić, M., Thys, K. J. M., & Vanhove, M. P. M. (2025). Scanning electron microscopy reveals new ultrastructural features in metacercariae of *Clinostomum cutaneum*. *Pathogens*, 14(3), 249.

Shvydka, S., Sarabeev, V., Estruch, V. D., & Cadarso-Suárez, C. (2018). Optimum sample size to estimate mean parasite abundance in fish parasite surveys. *Helminthologia*, 55(1), 1–9. <https://doi.org/10.1515/helm-2017-0054>

- Smyth, J., & Halton, D. W. (1983). *The physiology of trematodes*. Cambridge University Press.
- Solórzano-García, B., White, J. M., & Shedden, A. (2023). Parasitism in heterogeneous landscapes: Association between conserved habitats and gastrointestinal parasites in populations of wild mammals. *Acta Tropica*, 237, 106751. <https://doi.org/10.1016/j.actatropica.2022.106751>
- Soubeiga, P., Bounboua, M., Sinaré, Y., Ayoro, J. H., & Kabre, G. B. (2020). Trematodes of the genus *Mesocoelium*, parasites of anurans in the Ganzourgou Province, Burkina Faso. *European Scientific Journal ESJ*, 16(18), 347. <https://doi.org/10.19044/esj.2020.v16n18p347>
- Spawls, S., Howell, K., & Drewes, R. (2006). *A pocket guide to reptiles and amphibians of East Africa*. A & C Black Publishers.
- Stout, L., Daffe, G., Chambouvet, A., Correia, S., Culloty, S., Freitas, R., Iglesias, D., Jensen, K. T., Joaquim, S., Lynch, S., Magalhães, L., Mahony, K., Malham, S. K., Matias, D., Rocroy, M., Thieltges, D. W., & de Montaudouin, X. (2024). Morphological vs. molecular identification of trematode species infecting the edible cockle *Cerastoderma edule* across Europe. *International Journal for Parasitology: Parasites and Wildlife*, 25, 101019. <https://doi.org/10.1016/j.ijppaw.2024.101019>
- Stuart, S. N., Chanson, J. S., Cox, N. A., Young, B. E., Rodrigues, A. S., Fischman, D. L., & Waller, R. W. (2004). Status and trends of amphibian declines and extinctions worldwide. *Science*, 306(5702), 1783–1786. <https://doi.org/10.1126/science.1103538>
- Svitin, R., Schoeman, A. L., & du Preez, L. H. (2018). New information on morphology and molecular data of camallanid nematodes parasitising *Xenopus laevis* (Anura: Pipidae) in South Africa. *Folia Parasitologica*, 65, Article 2018.003. <https://doi.org/10.14411/fp.2018.003>
- Tagar, I., Shaikh, K., Gachal, G. S., Arain, I., Arain, M., & Sarhindi, B. A. (2019). Assessment of

anurans (frogs and toads) inhabiting district Naushahro Feroze, Sindh, Pakistan. *Journal of Entomology and Zoology Studies*, 7(1), 1419–1422.

Tattersall, G. (2007). Skin breathing in amphibians. In W. Aird (Ed.), *Endothelial biomedicine: A comprehensive reference* (pp. 85–91). Cambridge University Press.

Theunissen, M., Tiedt, L., & du Preez, L. H. (2014). The morphology and attachment of *Protopolystoma xenopodis* (Monogenea: Polystomatidae) infecting the African clawed frog *Xenopus laevis*. *Parasitology Research*, 113(6), 2191–2200. <https://doi.org/10.1007/s00436-014-3872-1>

Tinsley, R. C. (1995). Parasitic adaptation in the Polystomatidae (Monogenea). *Parasitology Today*, 11(8), 302–306. [https://doi.org/10.1016/0169-4758\(95\)80041-9](https://doi.org/10.1016/0169-4758(95)80041-9)

Tkach, V. V., Lisitsyna, O. I., Crossley, J. L., Thi Binh, T., & Bush, S. E. (2013). Morphological and molecular differentiation of two new species of *Pseudoacanthocephalus* Petrochenko, 1958 (Acanthocephala: Echinorhynchidae) from amphibians and reptiles in the Philippines, with identification key for the genus. *Systematic Parasitology*, 85, 11–26. <https://doi.org/10.1007/s11230-013-9414-8>

Ubiratã Ferreira Souza, L. R., Mendonça, L. R., Ceron, K., Meneses, A. S. O., Moura, G. J. B., & Dubeux, M. J. M. (2023). Frog eat frogs: The relationship among the Neotropical frogs of the genus *Leptodactylus* and their anuran prey. *Food Webs*, 37, e00326. <https://doi.org/10.1016/j.fooweb.2023.e00326>

Verneau, O., Du Preez, L., & Badets, M. (2009). Lessons from parasitic flatworms about evolution and historical biogeography of their vertebrate hosts. *Comptes Rendus Biologies*, 332(2–3), 149–158. <https://doi.org/10.1016/j.crv.2008.08.019>

Vitt, L. J., & Caldwell, J. P. (2014). *Herpetology: An introductory biology of amphibians and*

reptiles. Academic Press.

Wells, K. D. (2007). *The ecology and behavior of amphibians*. University of Chicago Press.

Whittington, I. D., & Horton, M. A. (1996). A revision of *Neoheterocotyle* (Monogenea: Monocotylidae) with descriptions of *Neoheterocotyle rhynchobatis* sp. nov. from the gills of *Rhynchobatus djiddensis* (Forsskål) (Rhynchobatidae) and *Neoheterocotyle rhinobatidis* sp. nov. from the gills of *Rhinobatos typus* (Smith) (Rhinobatidae) from Western Australia. *Systematic Parasitology*, 33(3), 211–231. <https://doi.org/10.1007/BF00010136>

Williams, H., & Jones, A. (1994). *Parasitic worms of fishes*. Taylor and Francis.

Willson, J. D., & Dorcas, M. E. (2003). Effects of seed ingestion on the movement and germination of seeds of the bullfrog, *Rana catesbeiana*. *Oecologia*, 134(4), 568–576.
<https://doi.org/10.1007/s00442-002-1163-1>

Wilson, K., Bjørnstad, O. N., Dobson, A. P., Merler, S., Poglayen, G., Randolph, S. E., Read, A. F., & Skorping, A. (2002). Heterogeneities in macroparasite infections: Patterns and processes. In P. J. Hudson, A. Rizzoli, B. T. Grenfell, H. Heesterbeek, & A. P. Dobson (Eds.), *The ecology of wildlife diseases* (pp. 6–44). Oxford University Press.

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

Woodward, M. (1992). Formulae for sample size, power and minimum detectable relative risk in medical studies. *Journal of the Royal Statistical Society: Series D (The Statistician)*, 41(2), 185–196.
<https://doi.org/10.2307/2348392>

Yamaguti, S. (1959). *Systema helminthum: Vol. II. The cestodes of vertebrates*. Keigaku Publishing House.

Young, T. M. (2017). *Biodiversity calculator for the Simpson and Shannon indexes*.

https://www.alyoung.com/labs/biodiversity_calculator.html

Zweifel, R. G., & Cash, B. W. (2021). Anura. In *Access Science*. <https://doi.org/10.1036/1097-8542.042200>

LIST OF APPENDICES

Appendix 1: Journal Publication

Parasitology Research (2025) 124:163
https://doi.org/10.1007/s00436-025-08609-7

RESEARCH



Spatio-temporal infection and diversity indices of gastrointestinal helminths in *Xenopus* species from Lake Victoria Basin, Kenya

Kiki M. A. Ochieng¹ · Nehemiah M. Rindoria^{2,3} · George N. Morara⁴ · Wilmien J. Luus-Powell³ · Willem J. Smit³

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Abstract

Xenopus frogs (Pipidae) are aquatic anurans widely distributed across Africa, where they play important ecological, biomedical, and cultural roles. However, populations are increasingly threatened by a range of stressors, including habitat degradation, climate change, and parasitic infections. This study investigated spatio-temporal infection and diversity indices of gastrointestinal helminths in *Xenopus borealis* and *X. victorinus* from the Lake Victoria Basin, Kenya. A total of 480 individuals (240 per species) were collected between June 2024 and January 2025 from selected streams and ponds using baited traps and seine nets. Spatio-temporal variations in environmental conditions were recorded using a YSI multiparameter meter. Standard parasitological techniques were employed for the recovery, fixation, and identification of parasites. Three helminth genera were recorded: the intestinal cestode *Cephalochlamys* sp., and two nematodes, *Contracaecum* sp. (intestinal) and *Paracramallanus* sp. (stomach). Prevalence ranged from 23% to 90%, and mean intensity from 1.0 to 8.3, depending on parasite taxon and host species. Nematodes were the most prevalent and diverse group across all localities. Diversity metrics, including Shannon-Wiener, Margalef richness, Berger-Parker dominance, and Simpson's index, showed variation between hosts and sampling sites. Analysis of variance (ANOVA) revealed no significant spatial differences in infection indices ($p > 0.05$), but temporal differences were significant ($p < 0.05$) for *Contracaecum* infections in *X. borealis* and *Paracramallanus* in *X. victorinus*. These findings provide the first baseline data on helminth fauna from *Xenopus* species in Kenya and support the need for molecular analyses to clarify taxonomy and better understand host-parasite-environment interactions.

Keywords Anura · Pipidae · Diversity indices · Helminths · Infection indices

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Appendix 2: Kisii University introduction letter for acquisition of the NACOSTI research permit


KISII UNIVERSITY

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**OFFICE OF THE REGISTRAR RESEARCH, INNOVATION, EXTENSION,
AND RESOURCE MOBILIZATION**

REF: KSU/REIRM/140 **DATE:** 28th August, 2024

The Head, Research Coordination
National Council for Science, Technology, and Innovation (NACOSTI)
Utalii House, 8th Floor, Uhuru Highway
P. O. Box 30623- 00100
NAIROBI - KENYA

Dear Sir/Madam,

RE: KIKI MARIA ATIENO OCHIENG: MAN19/00001/23

The above-mentioned is a student of Kisii University currently pursuing a Degree of Master of Science in Limnology in the School of Agriculture and Natural Resource Management. The topic of her research is, **"Diversity, Morphological and Molecular Characterisation of Parasites of Xenopus Borealis Parker, 1936 And Xenopus Victorianus AHL, 1924 in Lake Victoria Basin Kenya."**

We are kindly requesting assistance in acquiring a research permit to enable the student to carry out the research, upon getting ethical clearance by an accredited Scientific and Ethics Review Committee.

Thank you.


Prof. Christopher Ngacho, PhD
AG. REGISTRAR, (REIRM)
CN/bm


Kisii University is ISO 9001:2015 certified

Appendix 3: NACOSTI research permit

 REPUBLIC OF KENYA	 NATIONAL COMMISSION FOR SCIENCE, TECHNOLOGY & INNOVATION
Ref No: 415801	Date of Issue: 26/September/2024
RESEARCH LICENSE	
	
This is to Certify that Miss. Kiki Maria Arieno ocheng of Kisii University, has been licensed to conduct research as per the provision of the Science, Technology and Innovation Act, 2013 (Rev.2014) in Busia, Homabay, Kisii, Kisumu, Migori, Siaya, Vihiga on the topic: DIVERSITY, MORPHOLOGICAL AND MOLECULAR CHARACTERISATION OF PARASITES OF <i>Xenopus borealis</i> PARKER, 1936 AND <i>Xenopus victorinus</i> AHL, 1924 IN LAKE VICTORIA BASIN, KENYA for the period ending : 26/September/2025.	
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Appendix 4: Ethical clearance for research work from KTRH-ISERC



KISII COUNTY GOVERNMENT DEPARTMENT OF HEALTH

Telegramme "Medical"
Telephone: (058) 31310 Kisii
E-Mail: ktrh.erc@yahoo.com
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KTRH ISERC
KISII TEACHING & REFERRAL HOSPITAL
P.O Box 92 – 40200,
KISII

Date: 28th January 2025

REF: ISERC/KTRH/058/24

Ms. Maria Kiki Atieno
Principal Investigator,
Kisii University,
Kisii.

Dear Ms. Maria,

RE: AUTHORIZATION TO CONDUCT A RESEARCH STUDY TITLED 'DIVERSITY, MORPHOLOGICAL AND MOLECULAR CHARACTERIZATION OF PARASITES *Xenopus borealis parker* 1936 AND *Xenopus victoriaris* AHL 1924 IN LAKE VICTORIA BASIN (ISERC/KTRH/058/24)

This is to inform you that **KTRH ISERC** has reviewed and approved your above research proposal. Your application approval number is **ISERC/KTRH/058/24**. The approval period is **28th January 2025 – 28th January 2026**.

This approval is subject to compliance with the following requirements:

- i. Only approved documents including (informed consents, study instruments, MTA) will be used.
- ii. All changes including (amendments, deviations, and violations) are submitted for review and approval by **KTRH ISERC**.
- iii. Death and life-threatening problems and serious adverse events or unexpected adverse events whether related or unrelated to the study must be reported to **KTRH ISERC** within 72 hours of notification.
- iv. Any changes, anticipated or otherwise that may increase the risks or affected safety or welfare of study participants and others or affect the integrity of the research must be reported to **KTRH ISERC** within 72 hours.
- v. Clearance for export of biological specimens must be obtained from relevant institutions.
- vi. Submission of a request for renewal of approval at least 60 days prior to expiry of the approval period. Attach a comprehensive progress report to support the renewal.

- vii. Submission of an executive summary report within 90 days upon completion of the study to **KTRH ISERC**.

Prior to commencing your study, you will be expected to obtain a research license from National Commission for Science, Technology and Innovation (NACOSTI) <https://research-portal.nacosti.go.ke> and also obtain other clearances needed.

Yours sincerely



Florence Ogero.

Chair, KTRH ISERC

Appendix 5: Statistical analysis report

Two-way ANOVA output for variation in physico-chemical parameters among study sites and seasons

					Dry	6.87/b2
	s1	s2	s3			
KK	6.72	6.36	6.97			
VH	6.7	7	6.74			
SY	7.39	7.24	6.89			
KS	6.89	6.89	6.56			
HB	7.1	6.89	7.19			
MG	6.94	7.03	7.04			
KI	8.05	8.01	7.93			
BS	7.189999991	6.9	6.7			
	Anova: Two-Factor Without Replication					
	SUMMARY	Count	Sum	Average	Variance	
	Row 1	3	20.05	6.68333	0.094033333	
	Row 2	3	20.44	6.81333	0.026533333	
	Row 3	3	21.52	7.17333	0.065833333	
	Row 4	3	20.34	6.78	0.0363	
	Row 5	3	21.18	7.06	0.0237	
	Row 6	3	21.01	7.00333	0.003033333	
	Row 7	3	23.99	7.99667	0.003733333	
	Row 8	3	20.78999999	6.93	0.060699998	
	Column 1	8	56.97999999	7.1225	0.194964286	
	Column 2	8	56.32	7.04	0.215657143	
	Column 3	8	56.02	7.0025	0.180964286	
	ANOVA					
	Source of Variati	SS	df	MS	F	P-value F crit
	Rows	3.573666669	7	0.51052	12.59589975	4.5E-05 2.764199
	Columns	0.060299999	2	0.03015	0.743875922	0.49315 3.738892
	Error	0.56743333	14	0.04053		
	Total	4.201399998	23			

temp						
anova: Two-Factor Without Replication						
MMAP	Count	Sum	Average	Variance		
ow 1	3	84.2	28.067	0.9233		
ow 2	3	73.3	24.433	16.303		
ow 3	3	84.5	28.167	4.1033		
ow 4	3	84.8	28.267	0.7233		
ow 5	3	78.8	26.267	4.2433		
ow 6	3	87.3	29.1	0.49		
ow 7	3	81.9	27.3	1.33		
ow 8	3	80.9	26.967	4.6233		
olumn 1	8	203.7	25.463	7.1598		
olumn 2	8	221.4	27.675	1.165		
olumn 3	8	230.6	28.825	0.7421		
NOVA						
Source of Variation	SS	df	MS	F	P-value	F crit
ows	44.72	7	6.3885	4.7703	0.0063	2.7642
olumns	46.731	2	23.365	17.447	0.0002	3.7389
error	18.749	14	1.3392			
Total	110.2	23				

dissolved oxygen

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	4.8	1.6	0.04
Row 2	3	21.4	7.1333	0.5033
Row 3	3	6.8	2.2667	0.0233
Row 4	3	9.8	3.2667	0.0933
Row 5	3	12	4	0.09
Row 6	3	12.1	4.0333	0.0233
Row 7	3	10.7	3.5667	3.1033
Row 8	3	10.9	3.6333	0.8633
Column 1	8	34.3	4.2875	3.607
Column 2	8	28.1	3.5125	2.7184
Column 3	8	26.1	3.2625	2.3741

ANOVA

Source of Vari	SS	df	MS	F	P-value	Fcrit
Rows	55.986	7	7.998	22.805	1E-06	2.7642
Columns	4.57	2	2.285	6.5153	0.01	3.7389
Error	4.91	14	0.3507			
Total	65.466	23				

tds

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	447.2	149.07	204.57
Row 2	3	305.67	101.89	49.509
Row 3	3	351.73	117.24	292.79
Row 4	3	410.77	136.92	27.451
Row 5	3	687.26	229.09	380.69
Row 6	3	406.73	135.58	408.35
Row 7	3	452.82	150.94	901.42
Row 8	3	355.26	118.42	21869
Column 1	8	1133	141.62	2338.4
Column 2	8	1129.3	141.17	1410.6
Column 3	8	1155.2	144.4	1422

ANOVA

Source of Vari	SS	df	MS	F	P-value	Fcrit
Rows	31672	7	4524.6	14.001	2E-05	2.7642
Columns	48.964	2	24.482	0.0758	0.9274	3.7389
Error	4524.3	14	323.17			
Total	36246	23				

depth

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	4.7	1.5667	0.0933
Row 2	3	5.5	1.8333	0.0233
Row 3	3	4.6	1.5333	0.0533
Row 4	3	6.3	2.1	0.01
Row 5	3	4.55	1.5167	0.0308
Row 6	3	4.5	1.5	0.01
Row 7	3	5.9	1.9667	0.0233
Row 8	3	4.5	1.5	0.09
Column 1	8	15	1.875	0.0507
Column 2	8	13.4	1.675	0.0679
Column 3	8	12.15	1.5188	0.0785

ANOVA

Source of Vari	SS	df	MS	F	P-value	Fcrit
Rows	1.2216	7	0.1745	15.451	1E-05	2.7642
Columns	0.5102	2	0.2551	22.586	4E-05	3.7389
Error	0.1581	14	0.0113			
Total	1.8899	23				

Two-way ANOVA output for variation in mean intensity across sites and seasons

	contraceacum		mi			
	wet	mod	dry			
Kakameg polystomaria	1.77778	3	0			
Kakameg	0	1.2	3			
Vihiga	2	1.33333	2			
Siaya	0	0	0			
Kisumu	0	2	1.33333			
Homabay	1.57143	1.44444	3.33333			
Migori	2	1.42857	2.16667			
Kisii	0	0	2.14286			
Busia	2	1.5	2.5			

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	4.2	1.4	2.28
Row 2	3	5.33333	1.77778	0.14815
Row 3	3	0	0	0
Row 4	3	3.33333	1.11111	1.03704
Row 5	3	6.34921	2.1164	1.11472
Row 6	3	5.59524	1.86508	0.14985
Row 7	3	2.14286	0.71429	1.53061
Row 8	3	6	2	0.25
Column 1	8	7.57143	0.94643	1.04337
Column 2	8	8.90635	1.11329	0.52566
Column 3	8	16.4762	2.05952	1.07029

ANOVA

Source of Var	SS	df	MS	F	P-value	Fcrit
Rows	11.2203	7	1.6029	3.09316	0.03429	2.7642
Columns	5.7658	2	2.8829	5.5632	0.01667	3.73889
Error	7.25493	14	0.51821			
Total	24.2411	23				

	camallanidae					
	wet	mod	dry			
Kakameg	6	10.6667	6.11111	Kakameg		
Vihiga	3.25	7.44444	5.5	Vihiga		
Siaya	7	7	6.3	Siaya		
Kisumu	6.5	4.71429	6	Kisumu		
Homabay	8.55556	6.5	4.71429	Homabay		
Migori	8.3	7.28571	5.88889	Migori		
Kisii	10.5	6.8	11.6667	Kisii		
Busia	9.8	6.5	5	Busia		

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	22.7778	7.59259	7.09053
Row 2	3	16.1944	5.39815	4.40612
Row 3	3	20.3	6.76667	0.16333
Row 4	3	17.2143	5.7381	0.84864
Row 5	3	19.7698	6.58995	3.69491
Row 6	3	21.4746	7.1582	1.46556
Row 7	3	28.9667	9.65556	6.45593
Row 8	3	21.3	7.1	6.03
Column 1	8	59.9056	7.48819	5.37223
Column 2	8	56.9111	7.11389	2.76919
Column 3	8	51.181	6.39762	4.83772

ANOVA

Source of Var	SS	df	MS	F	P-value	Fcrit
Rows	35.4573	7	5.06532	1.28012	0.32771	2.7642
Columns	4.91334	2	2.45667	0.62086	0.55165	3.73889
Error	55.3967	14	3.95691			
Total	95.7673	23				

	camallanidae					
	wet	mod	dry			
Kakameg	2.6	2.77778	8	Kakameg		
Vihiga	4.3	3.5	8.125	Vihiga		
Siaya	mean intensity	3.5	8.125	Siaya		
Kakameg	3	3	1.25	Kakameg		
Vihiga	3	2.88889	1.5	Vihiga		
Siaya	2.33333	1.875	1.66667	Siaya		
Kisumu	2.4	2.375	1.5	Kisumu		
Homabay	3.8	2.33333	3	Homabay		
Migori	1.44444	4	1.57143	Migori		
Kisii	2.5	2	1.33333	Kisii		
Busia	5	2.33333	3	Busia		

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	7.25	2.41667	1.02083
Row 2	3	7.38889	2.46296	0.69856
Row 3	3	5.875	1.95833	0.11632
Row 4	3	6.275	2.09167	0.26271
Row 5	3	9.13333	3.04444	0.53926
Row 6	3	7.01587	2.33862	2.07416
Row 7	3	5.83333	1.94444	0.34259
Row 8	3	10.3333	3.44444	1.92593
Column 1	8	23.4778	2.93472	1.1543
Column 2	8	20.8056	2.60069	0.46885
Column 3	8	14.8214	1.85268	0.5183

ANOVA

Source of Var	SS	df	MS	F	P-value	Fcrit
Rows	5.94121	7	0.84874	1.31313	0.31379	2.7642
Columns	4.91179	2	2.45589	3.79963	0.04806	3.73889
Error	9.04892	14	0.64635			

Kakameg			
Vihiga			
Siaya			
Kisumu	2	2.25	1.5
Homabay	2	1.4	1.5
Migori	0	2.33333	1.4
Total	19.9019	23	

	cephalochlamys					
	wet	mod	dry			
Kakameg	1.66667	1.6	1.28571	Kakameg		
Vihiga	6	1.8	1.4	Vihiga		
Siaya	1.22222	2.33333	2	Siaya		
Kisumu	1.75	1.66667	1.4	Kisumu		
Homabay	3.28571	2.33333	2.5	Homabay		
Migori	3	2.625	1	Migori		
Kisii	1.66667	1.75	1.33333	Kisii		
Busia	1.42857	1.5	1.66667	Busia		

Anova: Two-Factor Without Replication

SUMMARY	Count	Sum	Average	Variance
Row 1	3	4.55738	1.51913	0.04109
Row 2	3	9.2	3.06667	6.49333
Row 3	3	5.55556	1.85185	0.13251
Row 4	3	4.8867	1.62889	0.03343
Row 5	3	8.11905	2.70635	0.25969
Row 6	3	6.625	2.20833	1.13521
Row 7	3	4.75	1.58333	0.04861
Row 8	3	4.59524	1.53175	0.0493
Column 1	8	26.0798	3.25998	2.54881
Column 2	8	25.6083	3.20104	0.1738
Column 3	8	12.5857	1.57321	0.22953

ANOVA

Source of Var	SS	df	MS	F	P-value	Fcrit
Rows	7.44878	7	1.06403	1.12676	0.39924	2.7642
Columns	3.49433	2	1.74716	1.85347	0.1935	3.73889
Error	13.1871	14	0.94205			
Total	24.1396	23				

Total 9.15777 23

Test for normality Shapiro-wilks w

Tests for normal distribution			
	All		
N	8		
Shapiro-Wilk W	0.7923		
p(normal)	0.02364		
Anderson-Darling A	0.7835		
p(normal)	0.02372		
p(Monte Carlo)	0.0252		
Lilliefors L	0.3198		
p(normal)	0.01678		
p(Monte Carlo)	0.0144		
Jarque-Bera JB	1.167		
p(normal)	0.5579		
p(Monte Carlo)	0.1429		

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Monte Carlo N: 9999

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Recompute

Tests for normal distribution			
	All		
N	8		
Shapiro-Wilk W	0.8208		
p(normal)	0.0476		
Anderson-Darling A	0.6504		
p(normal)	0.0557		
p(Monte Carlo)	0.0557		
Lilliefors L	0.2888		
p(normal)	0.04701		
p(Monte Carlo)	0.0488		
Jarque-Bera JB	3.388		
p(normal)	0.1838		
p(Monte Carlo)	0.015		

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	All
N	8
Shapiro-Wilk W	0.7943
p(normal)	0.02484
Anderson-Darling A	0.7282
p(normal)	0.03382
p(Monte Carlo)	0.0354
Lilliefors L	0.2858
p(normal)	0.05171
p(Monte Carlo)	0.054
Jarque-Bera JB	4.216
p(normal)	0.1215
p(Monte Carlo)	0.0093

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Tests for normal distribution

	All
N	8
Shapiro-Wilk W	0.9232
p(normal)	0.4563
Anderson-Darling A	0.3369
p(normal)	0.4028
p(Monte Carlo)	0.4361
Lilliefors L	0.1954
p(normal)	0.4869
p(Monte Carlo)	0.4987
Jarque-Bera JB	1.02
p(normal)	0.6005
p(Monte Carlo)	0.18

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Tests for normal distribution
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	All
N	8
Shapiro-Wilk W	0.8741
p(normal)	0.1654
Anderson-Darling A	0.5309
p(normal)	0.1174
p(Monte Carlo)	0.1257
Lilliefors L	0.2911
p(normal)	0.04365
p(Monte Carlo)	0.0437
Jarque-Bera JB	1.572
p(normal)	0.4556
p(Monte Carlo)	0.0871

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Spearman's rank

Correlation

Table Plot

	1	2
1		0.89553
2	0.035714	

Correlation statistic

☐ Linear r (Pearson)
☐ Spearman's D
☒ Spearman's rs
☐ Kendall's tau
☐ Polyserial rho
☐ Partial linear

Table format

☒ Statistic \ p(uncorr)
☐ Statistic
☐ p(uncorr)
☐ Permutation p
☐ Bonferroni correction

Close Copy Print Help

Digenean	DO
45.83333333	5.6
46	5.55555555
48	5.22222222
50	5.22222222
50.00000000	4
48	4.88888888
54.55555555	5.55555555
46.66666667	5.55555555
46.66666667	5.6
55.55555555	5.55555555
46.66666667	5.22222222
50.00000000	5.22222222
50	4
48	4.88888888
56.66666667	5.55555555
55.55555555	5.88888888

36.66666667	150.94	36.66666667	7.993764257
23.33333333	119.25	23.33333333	6.886074246

Correlation

Table Plot

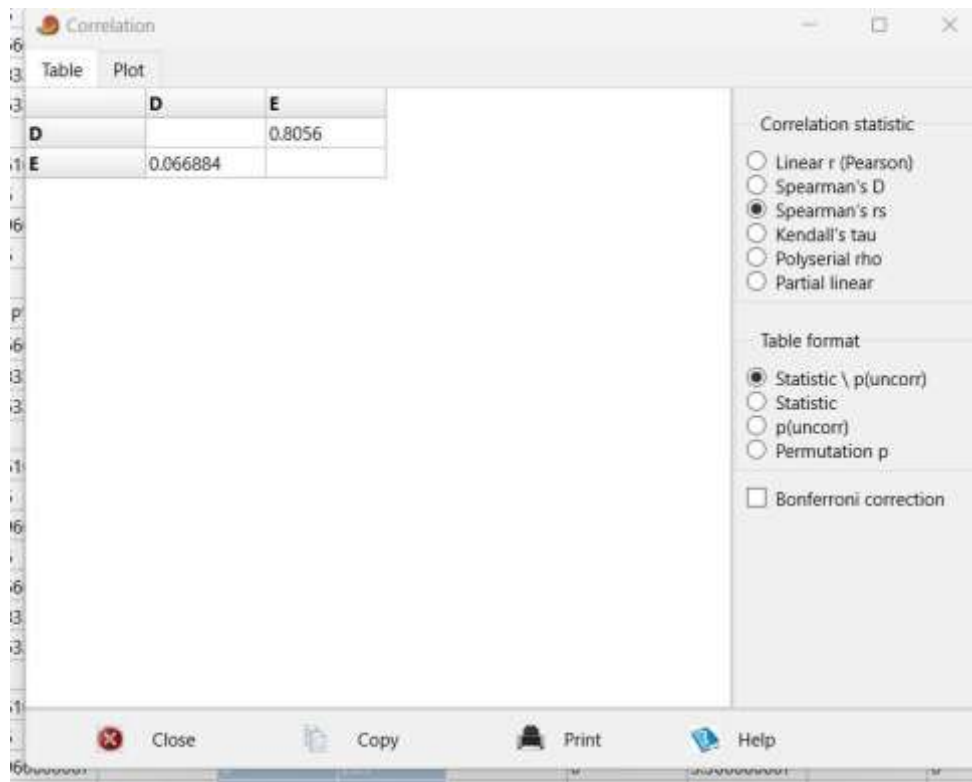
	A	B
A		0.84718
B	-0.052396	

Correlation statistic

☐ Linear r (Pearson)
☐ Spearman's D
☒ Spearman's rs
☐ Kendall's tau
☐ Polyserial rho
☐ Partial linear

Table format

☒ Statistic \ p(uncorr)
☐ Statistic
☐ p(uncorr)
☐ Permutation p
☐ Bonferroni correction



Chi square for spatial and temporal variation

23	7
11	19
13	17
12	18
12	18
14	16
22	8
12	18

Contingency table		—	□	×
Tests	Residuals			
Chi squared				
Rows, columns:	8, 2	Degrees freedom:	7	
Chi ² :	21.451	<i>p</i> (no assoc.):	0.0031563	
Monte Carlo <i>p</i> :	0.003			
Fisher's exact				
Not available				
Other statistics				
Cramer's <i>V</i> :	0.29897	Contingency <i>C</i> :	0.28644	
☐ Sample vs. expected	Permutation N:	9999	Recompute	
	Close		Copy	Print
			Help	

10	20
16	14
14	16
14	16
8	22
8	22
22	8
7	23

Contingency table		—	□	×
Tests	Residuals			
Chi squared				
Rows, columns:	8, 2	Degrees freedom:	7	
Chi ² :	25.291	<i>p</i> (no assoc.):	0.00067366	
Monte Carlo <i>p</i> :	0.0003			
Fisher's exact				
Not available				
Other statistics				
Cramer's <i>V</i> :	0.32462	Contingency <i>C</i> :	0.30876	
☐ Sample vs. expected	Permutation N:	9999	Recompute	

Appendix 6: Plagiarism report



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