Letters in Animal Biology 05(1): 30 - 41 *Review Article*

Letters in Animal Biology

Journal homepage: www.liabjournal.com

Prevalence of nematodes in wildlife and livestock animals and their control by medicinal plants

Muhammad Sajid *[1], Arooj Fatima [1], Tayyaba Mehmood [1], Muhammad Mahboob Ahmed Sirajvi [2], Maria Manzoor [2], Muhammad Zohaib [1], Usama Zafar Iqbal [3], Arslan Muhammad Ali Khan [4]

[1] *Department of Zoology Wildlife and Fisheries, University of Agriculture Faisalabad, Pakistan*
[2] *Department of Zoology, The Islamia University of Bahawalpur, Pakistan*
[3] *Department of Botany, University of Agriculture Faisalabad, Pakistan*
[4] *Department of Parasitology, University of Agriculture Faisalabad, Pakistan*

Article info

Received: 16 November 2024
Received in revised form: 13 February 2025
Accepted: 19 February 2025
Published online: 21 February 2025

Keywords
Parasitic nematodes
Prevalence
Wildlife
Livestock
Medicinal plants

* *Corresponding author:*
Muhammad Sajid
Email: m.sajidsahu123@gmail.com

Reviewed by:
Zaib-Un Nisa
Government college university, Faisalabad

Husnain Idrees
Equine Veterinary Hospital and Diagnostic Laboratory Lahore Race Club, Pakistan

Consent to publish the name of third reviewer could not be obtained

Abstract

All animal species are infected with parasitic nematodes, which are extremely widespread and diverse. Their significance in natural ecosystems is becoming increasingly clear. Host movement has long been known to have a significant role in deciding the genetic makeup of parasitic nematode populations. More recently, studies have emphasized the significance of nematode life histories, environmental factors, and other host ecological components. Parasitic infections by nematodes continue to be a major global health concern, particularly in developing nations, despite advancements in contemporary human and veterinary treatment. Research on alternative treatments for nematode infections has increased due to mounting evidence of these nematodes' multidrug resistance and the adverse effects of currently available synthetic pharmaceuticals. In this situation, investigating possible botanical antiparasitics which are widely available and priced might be a practical substitute. In this review the prevalence of nematodes is summarized and how the infection of nematodes can be treated through plant-based drugs instead of conventional approaches.

1. Introduction

Nematodes, cestodes, and trematodes belong to helminths that cause parasitic infections in domestic and wildlife species including small and large ruminants. Among all helminths, nematodes have significant importance due to their host-parasite interaction and their effect on the host immunity (King and Li 2018). Nematodes and other protozoans cause a complex immunological response. The infections due to nematodes are much more severe and most of the time they develop resistance due to their genetic modifications. Nematodes also have zoonotic importance because they cause many human infections but most importantly, they target domestic and wildlife species (Thompson 2023). Several wildlife species are affected by nematode infections which cause huge economic and species losses. In most cases, light infections will drop the growth and development of the animals, and heavy infections cause the death of the animals (Stear et al. 2003). Nematode infections play a significant role in determining wildlife health and biodiversity. These parasitic worms are common in many wildlife species and can affect population dynamics, affecting both individual animals and ecosystem stability. Wildlife infected with nematodes may experience reduced health, reproductive output, and increased vulnerability to predators, which can lead to population declines, particularly in species with already small populations (Stear et al. 2023). Infection with nematodes can result in severe health consequences, including organ damage, nutritional depletion, and compromised immune function. For example, gastrointestinal nematodes in ruminants and lungworms in some wildlife species can lead to malnutrition and respiratory issues, which make animals more susceptible to other diseases and environmental stressors (Pybus et al. 2023). In ecosystems where predator-prey relationships are tightly interwoven, such health impacts can also alter predation patterns by weakening prey species or reducing their populations, which in turn affects predator species and other members of the ecosystem (Postema 2023).

The significance of nematode infections extends beyond individual hosts to influence biodiversity by potentially reducing genetic diversity within affected populations. When infection levels are high, selective pressure can lead to an increased frequency of resistant individuals, potentially limiting genetic variability. Additionally, nematode infections can create competition between host species, as some hosts become reservoirs for these parasites, spreading them to other species, thus altering interspecies dynamics (Elhady et al. 2024).

https://doi.org/10.62310/liab.v5i1.185

Moreover, nematodes can have indirect impacts on biodiversity. Some nematode species are zoonotic, meaning they can be transmitted to humans, especially in regions where wildlife and human populations overlap. The presence of zoonotic nematodes requires careful management to protect public health while preserving biodiversity, which adds complexity to conservation efforts. The dynamic interplay between host species, parasite load, and environmental factors underscores the importance of understanding and managing nematode infections within wildlife populations to safeguard both ecosystem health and biodiversity (Otranto and Deplazes 2019). This review is focused on various nematode species present in different animals in different geographical areas and their transmission to other animals of wildlife importance. The second part of the study described the role of medicinal plants in controlling nematode infections because they are present in every part of the world and more specifically their prevalence is higher in North America, Europe, the Middle East, and some countries of Asia.

2. Prevalence of Eustrongylides nematodes

2.1. North America

USA and Mexico were two countries where firstly the Eustrongylides nematodes were reported.. The prevalence of infection in *Neogobius melanostomus* ranged from 0.5% in Michigan Lake to 95% in *Perca flavescens* in Lake Huron (Muzzall 1999). Among other fishes such as *Fundulus heteroclitus*, Eustrongylidosis was also reported. The intermediate hosts of the infection due to Eustrongylides in the waters of Mexico were reported among fishes such as *Ictalurus balsanus* and *Alloophorus robusts* and the prevalence of the infection ranged from 5.8% to 19% (Quiroz-Martinez and Salgado-Maldonado 2012). The fish-eating birds of the orders Ciconiformes, *Egretta thula, Ardea Herodias,* and *E. caerulae* are the definitive hosts of Eustrongylides (Ziegler et al. 2000). In some snakes *Thamnophis sirtalis Parietalisi* the group of reptiles Eustrongylidosis was also reported. Some investigators described on biting activity of Eustrongylides nematodes in humans in the states of New York, California, New Jersey, and Maryland, but they reported no data regarding the prevalence of infection (Narr et al. 1996).

2.2. Europe

Norway, Bulgaria, the UK, Romania, Italy, Lithuania, Serbia, Poland, Ukraine, Moldova, and Russia were among the most prominent countries in Europe where the prevalence of nematodes was reported. In fish that are intermediate hosts, the prevalence of the infection varied from 0.13% (Sokolov and Moshu 2014) to about 100% in the members of *Esox tunica, Silurus glanis,* and *Perca fluviatilis* (Branciari et al. 2016). In Ukraine (12-100%), Moldova (up to 100%), and Russia (33.1 to 62.2%) highest prevalence of Eustrongylides was recorded in Eastern Europe. In fishes of *Acipenseridae*, *Gobiidae*, *Salmonidae*, *Percidae*, and *Cyprinidae* the Eustrongylides were most commonly found (Sloboda et al. 2010). The statements also stated the incursion of definitive hosts, for instance, Waterfowl (*Phalacrocorax carbo*) with the pervasiveness of disease from 7.14% (Svazas et al. 2011) and wild ducks up to 3.71% (Kavetska et al. 2012). Reptiles and mammals were unintended hosts for Eustrongylides (Sloboda et al. 2010).

2.3. The Middle East

Turkey and Iran were the prominent countries in the Middle East where Eustrongylides was reported. The pervasiveness of disease varies in intermediate hosts such as fish ranging from 3.5% in *Rutilus rutilus* species to near about 100% in some species of the Sander genus such as *Sander lucioperca* (Metin et al. 2014). Among many other fishes such as *Perca fluviatilis Abramis brama,* and *Esox Lucius*, Eustrongylides were found (Fallah et al. 2015). Among many accidental hosts from Amphibian, *Rana ridibunda* and *Pelophylax ridibundus* were reported (Leon Regagnon 2019). Although in the Middle East prevalence of nematodes was high there was no mentioned case of human infection in the study.

2.4. Asia

Japan, China, India, and Java were the lands where *Eustrongylids* were found (Honcharov et al. 2022). The infection rate varies among fishes from intermediate hosts fluctuating from 6.3% in *Hypomesus transpacificus* to 29.3% in *Synbranchus bengalensis* (Subekti et al. 2020). Recently prevalence of *Eustrongylids* nematodes known as *Gnathostome spinigerum* were reported in *Monopterus albus* (Asian swamp eel) (Zhang et al. 2021). Another study revealed that *Eustrongylids* spp. Nematodes are prevalent in various species in China, as evidenced by molecular analysis of their intra-and interspecific evolutionary variations using the COI genes and ITS rDNA regions (Xiong et al. 2013). Similarly, Eustrongylids were collected from the *Channa punctata* found in polluted rivers in India along with other ectoparasites (Biswas et al. 2023). Other than Eustrongylids various gastrointestinal nematodes were detected in wildlife species in various regions of Asia including India, Pakistan, Bangladesh, Sri Lanka, and China (Otranto and Deplazes 2019; Chandrakar et al. 2020; Ahmad et al. 2024).

3. Nematodes of terrestrial vertebrates of wildlife Animals

3.1. Parasitic nematodes of marsupials

The parasitic nematodes that transmit disease to macropod marsupials are often the topic of population genetic research. A single species of nematodes can infect multiple host species, reflecting its adaptability in host-parasite interactions and its potential impact on biodiversity and disease dynamics (Heppert et al. 2022) of different families. This is due to the variation in the genetic structure of the nematodes that they adapt according to the situation. It is not always true because some studies confirm no genetic variation or variation of some genes that does not express much, but it may be due to a smaller number of studies on parasites and their less interaction with the host. Table 1 shows different species of nematodes and their respective hosts as well as their genetic makeup.

3.2. Nematodes of terrestrial carnivores

There are various nematodes including the genera of *Ancyclostoma, Capillaria, Strongyloides, Toxocara*, and *Trichinella* that affect the carnivorous vertebrates but the most important are *Trichinella* spp. They have wide geographic and host species ranges. Certain population genetic traits, such as differentiation between genetic variations in different populations, are shared by *Trichinella* spp. (La Rosa et al. 2012). For instance, genetic divergence was discovered between populations of *Trichinella pseudospiralis* from Australia, North America, Europe, and Asia, as well as between populations of *Trichinella spiralis* from Asia and Europe. *Trichinella* spp. frequently exhibit different genetic structures in different population that also vary with the change in geographical area (Dunams-Morel et al. 2012). Other species of *Trichinella* such as *T. nelsoni* prevalent in Kenya and Tanzania are different from the species found in South Africa (Di Cesare et al. 2014). Similar to other worms that

Table 1 Marsupial parasitic Strongylid nematodes

Nematode species	Definitive host species	Genetic markers	Reference
Cloacina caenis	*Petrogale assimilis, P. inornate*	ITS1 and ITS2	(Chilton et al. 2017)
C. robertsi	*P. assimilis, P. purpureicollis, P. persephone*	ITS1 & ITS2	(Song et al. 2024)
C. ernabella	*P. purpureicollis*	ITS1 & ITS2	(Peacock et al. 2022)
Globocephaloides trifidospicularis	*Macropus rufogriseus, M. giganteus*	24 isozymes	(Jabbar et al. 2013)
G. macropodis	*M. agilis, M. dorsalis*	ITS1 & ITS2	(Juillard and Ramp 2022)
Labiostrongylus bancrofti	*M. dorsalis, M. parryi*	18 isozymes	(Chilton et al. 2011)
L. unicatus	*M. dorsalis, M. parryi*	17 isozymes	(Palomba et al. 2021)
Macrostrongylus baylisi	*M. giganteus, M. erubescenes, M. robustus robustus, M. parryi*	27 isozymes	(Beveridge and Gasser 2014)
Papillostrongylus labiatus	*M. dorsalis, M. rufus*	ITS2	(Cole and Viney 2018)
Paramacropostrongylus typicus	*M. giganteus, M. fuliginous*	37 isozymes	(Sukee et al. 2018)
Rugopharynx australis	*M. eugenii, Wallabia bicolor, Thylogale billardierii, M. robustus, M. rufus, M. giganteus, M. fuliginosus*	17 isozymes	(Cole and Viney 2018)
R. omega	*T. stigmatica, M rufogriseus*	23 isozymes	(Flores et al. 2019)
R. zeta	*P. assimilis, M. dorsalis*	21 isozymes	(Akhoundi et al. 2017)
Zoniolaimus mawsonae	*M. rufus*	ITS2	(Coley and Viney 2018)

parasitize big carnivorous animals, *Trichinella* spp. appear to lack population genetic heterogeneity within countries and even in continents. This is due to the long-distance dispersion of hosts, which encourages substantial gene flow of parasites. Smaller hosts (like rats and foxes) may also encourage gene flow for *Trichinella* spp., allowing parasites to spread across otherwise dispersed populations of highly mobile hosts (Rothmann and de Waal 2017).

3.3. Parasitic nematodes of rodents

According to mitochondrial sequence analysis of nematode species in rodents, populations of the parasite *Heligmosomoides polygyrus*, which infects the European wood mouse (*Apodemus sylvaticus*), show significant population genetic structure throughout the host species' range. The mitochondrial mutation rate and generation time of *H. polygyrus* are quicker than those of its host. This suggests that mitochondrial genetic drift occurs more quickly in *H. polygyrus* than in *A. sylvaticus*, which contributes to the relatively stronger population genetic structure of *H. polygyrus* (Nieberding et al. 2008). Both *Trichuris* species are widespread in Europe. *T. muris* infects rats and mice, including *A. sylvaticus*, and *T. arvicolae* infects arvicoline rodents, such as lemmings and voles. Analysis of both nuclear and mitochondrial loci reveals that *T. muris* and *T. arvicolae* exhibit widespread population genetic structure throughout their geographic distribution, similar to *H. polygyrus* (Wasimuddin et al. 2016). *H. polygyrus* and both *Trichuris* species showed mostly identical patterns of population genetic structure, with a distinction between eastern and western populations and more variety in southern populations than in northern ones. The rodent hosts' range extension from southern refugia during the last ice age, at least 12,000 years ago, may be reflected in these patterns (Callejon et al. 2012).

3.4. Amphibian and reptilian parasitic nematodes

Anole lizards (*Anolis spp.*) are infected by *Spauligodon anolis*, whilst a variety of lizards and snakes are infected by the species complex *Parapharyngodon cubensis* (*P. cubensis* A, *P. cubensis* B, and *P. cubensis* C). According to a study on the population genetics of these nematodes, which were obtained from a variety of Caribbean *Anolis spp*. hosts, genetic variation was divided across and between islands (Falk and Perkins 2013). Nevertheless, populations of *S. anolis* exhibited more genetic differentiation than populations of *P. cubensis* A or *P. cubensis* B. This is probably because *S. anolis* has a limited host species range consisting of poor dispersers. Nevertheless, populations of *S. anolis* exhibited more genetic differentiation than populations of *P. cubensis* A or *P. cubensis* B. This is probably due to the fact that *S. anolis* has a limited host species range consisting of poor dispersers (Calsbeek 2009). However, each of the *P. cubensis* complex's species uses a greater variety of hosts, some of which may be more mobile. However, even though their host range varied, cryptic species of *Spauligodon atlanticus*, parasites of *Gallotia* spp. lizards, all displayed robust genetic structures within and across the Canary Isles Islands (Jorge et al. 2011). This may be because the geographical ranges of the host species of *S. atlanticus* do not overlap, precluding nematode gene flow between them.

3.5. Parasitic nematodes of ungulates

Since ungulate (hoofed mammal) individuals travel far longer distances than rodents, their parasitic nematodes may be able to spread their genes more readily. Similar to *Teladorsagia boreoarcticus* in muskoxen (*Ovibos moschatus*), *Ostertagia gruehneri* and *Marshallagia marshalli*, parasites of reindeer (*Rangifer tarandus*), exhibit a lack of population genetic organization (Cole and Viney 2018). On the other hand, populations of the parasite *Mazamastrongylus odocoilei*, which affects white-tailed deer (*Odocoileus virginianus*), exhibited genetic structure spread to almost all continents (Long et al. 2008). A parasite of many deer species (*Cervus* spp. and *Dama* spp.) is *Dictyocaulus eckerti*. While *D. capreolus* displayed relatively lesser genetic diversity and more highly genetically structured populations when sampled sympatrically.

3.6. Parasitic nematodes of marine mammals and birds

Among the enopleane vertebrate nematodes, two families named Trichinelloidea and Dioctophymatoidea are present in coastal birds, while the remaining group of enopleane nematodes is present in aquatic organisms. Nematodes of these two families are mostly present in every organ of the bird but most of the nematodes are found in the gastrointestinal tract, gizzard, and proventiculus of the birds. They may be found in other organs such as the heart and dermal layers but their number is not more than the gastrointestinal parasites. Even every nematode has its predilection site. The species of different systematic groups generally have their specific predilection sites. Prior to reaching the final host, the majority of nematode infections that affect marine mammals and birds spread tropically among intermediate species. Determinate host individuals will sample broadly from the parasite population since hosts of each trophic level are likely to eat several infected hosts at the lower trophic levels, causing hosts to mass parasites from a range of sources. This might result in genetically varied parasite intrapopulation that prevents inbreeding and encourages elevated Ne levels (Cole and Viney 2018). The gene flow of their parasitic nematode populations is predicted to be strong due to the long travel lengths of many marine fish, mammal, and bird hosts; this implies that these worm populations will show minimal genetic structure. A large number of parasitic nematodes that infect marine animals indeed have minimal genetic organization within their populations. A group of cryptic species with distinct geographic and definite host ranges make up the Anisakis simplex complex (Pontes et al. 2005).

4. Impact of nematodes on wildlife animals

In parasite ecology, one of the most common features is the aggregation of parasites within the groups of the host and a number of parasites are found in a small percentage of hosts. Individual hosts often differ in their visible levels of infection in natural populations. A wide range of variables, both internal to the host (host immune system or metabolic activities) and related to host-specific "intrinsic" (some characters that become specie specific due to acquired or hereditary factors) characteristics, contribute to this host heterogeneity in infection. The literature study revealed that different parasitic infections, particularly nematode infections, have a relationship with the age, sex, and reproduction of host animals. These have been studied in various wildlife species of deer, sheep, primates, rodents, and birds (Sol et al. 2003). It has been seen that nematodes mostly affect females compared to males but in most of the studies, it was investigated that both have equal chances of parasitic susceptibility (Lynsdale et al. 2020). The chances of nematode infections in females are more due to their estrous cycle phase, during pregnancy when immunity is decreased, and due to the lactation phase, when females bear a deficiency of micro and macronutrients. The parasitic burden of nematodes affects the overall growth and development of the host, hence leading to diseases. similarly, young animals are more prone to infections as compared to adults. In young animals, the level of immunity is not much to combat parasitic infections and if there is any bacterial or viral infection then there will be chances of mortality of the host. During heavy infections, various chemical drugs have been used but due to their frequent use nematodes have developed resistance and their control is very necessary. Various control strategies have been used for this purpose and some of these are discussed below.

5. Conventional strategies to control parasitic nematodes of wildlife

5.1. Environmental and habitat management

Managing habitats to reduce nematode exposure is the main goal of environmental management strategies. Changing these factors (habitat characteristics, wildlife source, climatic conditions) can interfere with the growth and transmission of many wildlife nematodes because they have intricate life cycles that call for intermediate hosts or certain environmental conditions. For instance, depending on the species, nematode larvae might live in plants, water, or soil. Rotational grazing is one practice that helps polluted land recover and prevents excessive parasite burdens. As many nematode larvae prefer damp conditions, eliminating feces and controlling soil moisture levels can work well (Bricarello et al. 2023). According to a recent study, wildlife managers can lower nematode infection rates by making changes to the environments where species exist (Mukherjee et al. 2023). For instance, exposure to certain nematodes is reduced when possible intermediate hosts, such rodents, are removed from grazing grounds. Certain wildlife populations' parasite burden can also be decreased in wooded regions by habitat changes like vegetation removal or controlled burning (Cable et al. 2017). According to studies, environmental management practices reduce the chances of nematodal infection that lessens the need for chemical treatments and promotes ecological balance (Bouchtaoui et al. 2024).

5.2. Chemical anthelmintic treatments

One of the main strategies for controlling nematode infections in wildlife is still chemical management. Adult and larval nematodes are the targets of anthelmintics, which include ivermectin and benzimidazoles. For instance, in controlled environments, these compounds are often used in feeding or through baiting programs for wild ungulates. However, there are drawbacks to using these medications, most notably the possibility that misuse or incorrect dosage can cause parasite populations to become resistant (Gianechini 2024). Anthelmintic resistance is a growing problem in wildlife parasitology, according to scientific investigations due to accumulation of chemicals in the body and body become immune to it. To minimize misuse of these treatments and to create resistance profiles for various nematode species, checking systems have been put in place in a number of national parks (Kapinder and Verma 2023). Based on diagnostic evaluations, researchers support selective therapy strategies in which only animals with severe infections are treated. This keeps the animal population at a low infection threshold while lowering the chance of resistance (Mukherjee et al. 2023).

5.3. Biological control using predatory fungi

Natural enemies of nematodes, such nematophagous fungus, are used in biological control techniques to capture and introduce worm larvae in the soil. After adhering to and penetrating the nematode's cuticle, the fungus release enzymes that break down the nematode's body. This method has proven effective in controlled settings, especially with cattle, and new studies are looking into how it may be used to manage wildlife (Berhanu et al. 2024). According to field research, adding these fungus spores to soil or water sources that animals use will lessen nematode infestations. Researchers are looking at using the fungus in natural wildlife habitats since it has been effective in lowering nematode numbers in livestock fields (Gianechini 2024). Nevertheless, there are still difficulties in using these fungi extensively in natural settings, and further research is required to comprehend the ecological effects on creatures that are not the intended targets (Panayotova-Pencheva 2024).

5.4. Immunization and genetic resistance

Research on immunization has grown in attention as a strategy to manage parasitic nematodes in animals. Certain nematode vaccines have been created for household animals and have the potential to be used in wildlife. Vaccines that target *Haemonchus contortus* in sheep, for example, have been shown to drastically lower parasite burdens; similar vaccines are currently being investigated for wild ruminants (Adduci et al. 2022). The broad application of this technique is, however, constrained by practical issues, such as delivering vaccinations to wildlife that is free to roam (Rupprecht et al. 2023). For captive or semi-wild populations, selective breeding initiatives to improve genetic resistance to nematodes offer a workable alternative. According to studies, certain species are inherently resistant to particular nematodes; populations can become more resilient by selectively breeding individuals with these qualities. For instance, selective breeding has improved the general health of controlled populations of several deer species by lowering nematode loads (Panayotova-Pencheva 2024).

5.5. Integrated pest management (IPM) approach

To manage parasites sustainably, an Integrated Pest Management (IPM) strategy integrates several control techniques. This approach figures out infection levels through diagnostic monitoring and adjusts control measures accordingly. By combining biological agents, selective chemical treatments, and environmental management, IPM reduces dependency on any one technique and aids in keeping parasite populations below harmful thresholds (Singh et al. 2023). IPM techniques for animal protection include habitat management, treatment rotation, and the sporadic use of biological agents. In addition to aiding in nematode population management, this strategy supports ecological stability and biodiversity. Research has demonstrated that IPM can successfully lower the incidence of nematodes in animals without resulting in the environmental damage linked to heavy pesticide use (Abd-Elgawad 2024).

6. Problems with conventional control methods of nematodes

Nematode infestations in agriculture and wildlife can be handled using a variety of techniques, each having exclusive hazards and effects on the ecosystem. Normally used chemical anthelmintic therapies, such ivermectin and benzimidazoles, can become the reason for environmental pollution, parasite population resistance, and detrimental effects on beneficial microbes and soil biodiversity (Navrátilová 2024). Rotational grazing and controlled burning are two environmental administration techniques that decline nematode habitats but have the potential to upset local ecosystems by affecting food webs and biodiversity. Although large-scale application may unintentionally harm non-target creatures, biological control, which targets nematodes without leaving chemical residues, provides an environmentally acceptable option by utilizing nematophagous fungus like *Duddingtonia flagrans* (Jones 2020). These techniques are used in Integrated Pest Management (IPM), which takes a more sustainable approach by employing focused tactics to lower chemical reliance, boost biodiversity, and manage parasite numbers (Deguine et al. 2021). For animal conservation initiatives, IPM's intricate coordination in natural environments is still difficult.

7. Control of nematodes by medicinal plants

Gastrointestinal (GI) parasite infections continue to be a major global health concern, particularly in impoverished nations, despite advancements in contemporary human and veterinary treatment. Research on alternative treatments for parasitic diseases has grown as a result of mounting evidence of these parasites' multidrug resistance and the adverse effects of currently available synthetic pharmaceuticals (Harhay et al. 2010). In this situation, investigating possible botanical antiparasitics that are widely available and reasonably priced might be a viable substitute (Ranasinghe et al. 2023). Many ancient medicinal systems across the world, including Chinese (Ayurveda), Arabic (Unani), and Indian (Ayurvedic), have their roots in plants. These ancient medical systems' treatments are grounded on thousands of years' worth of empirical research, some of which has been thoroughly documented (Patwardhan 2014). Similarly, a nematode known as *Hemonchus contortus* causes severe infections in almost all wildlife ruminants and in most of the cases it causes death of the animals. Many other nematodes are acknowledged to cause severe loss to wildlife, and many control methods are suggested but with their limitations (Emery et al. 2016).

8. Use of various plants to treat nematode infections

Various botanicals of therapeutic and medicinal effects have been used over the years to treat different infections and diseases in humans and animals. They have significant importance in the veterinary field to treat bacterial, viral, fungal, and parasitic infections. These medicinal values of the plants are due to the presence of various phytochemicals in them. Various laboratory examinations have confirmed the presence of active chemical components such as polyphenols, saponins, monocarboxylic acids, alcohols, flavonoids, terpenes, sesquiterpenes etc. However, control of nematodal infections by these phytochemicals is eco-friendly and cost saving. Various studies have been carried out to check the effect of different plants on the parasites of animals and some are listed below in the Table 2.

9. Compounds of plant origin with anthelmintic action

The molecules that have been shown to have nematocidal action are members of a broad family of compounds known as secondary metabolites, which are derived from the allelopathic interactions that occur between plants and their surroundings. As the name implies, these compounds are not connected to the main metabolism of plants, hence they are not essential to their growth (Santos et al. 1999). They are created by alternate cell metabolic processes that use amino acids and shikimic acid and are stored in sections like trichomes (epidermal appendages) or cellular vacuoles, depending on their chemical makeup, frequently in cells and organs far from the site of synthesis (Wink 2008). In response to diverse stimuli, plants simultaneously create a variety of chemicals. In certain situations, the same stimulus or agent might cause the production of many substances (KhokharVoytas et al. 2023). Every part of the plant, including the leaves, stems, roots, flowers, and seeds, can create secondary metabolites, and the species, stage of development, and geographical and climatic factors all influence concentration (Luz et al. 2010).

10. Mode of action

Secondary metabolites are ideal candidates for phytotherapeutic treatments because they can be linked to defense mechanisms against infection and other plants vying for resources, and solar protection. Additionally, they can serve to distribute seeds and draw pollinators to blooms and to encourage the fixation of nitrogen (Acamovic and Brooker 2005). These chemicals can interact with many molecules in

Table 2 Different medicinal plants and their target parasites

Plant species	Effective plant part	Parasites and associated diseases	Reference
Acokanthera oppositifolis	Leaves	Gastrointestinal nematodes	(Sanhokwe et al. 2016)
Allium cepa	Cloves	G.I nematodes	(Cheraghipour et al. 2019)
Allium sativum	Cloves	G.I nematodes	(Soliman et al. 2023)
Azadirachta indica	Leaves	nematode infestation and toxemia	(Ranasinghe et al. 2023)
Canabis sativa	Resin	GIT nematodes	(Guerra et al. 2023)
Dature metel	Garden fresh fruits	GIT nematodes	(Chatterjee et al. 2022)
Mentha spicata	Entire plant	GIT nematodes	(El Menyiy et al. 2022)
Musa paradisiaca	Leaves	GIT nematodes	(Oguntibeju 2019)
Nicotiana tabacum	Leaves	Ectoparasites and GIT nematodes	(Nouri et al. 2016)
Piper nigrum	Seeds	IDyspepsia, diarrhea, flatulence, and poisoning	(Saad et al. 2022)
Senna italica	Entire plant	G.I parasite and ectoparasites	(Yongwa et al. 2020)
Trachyspermum ammi	Seeds	Ectoparasites and endoparasites	(Abbas et al. 2019)
Trianthema portulacastrum	Entire plant	G.I parasite	(Hussain et al. 2011)
Veronia anthelmintic	seeds	G.I parasite	(Dogra et al. 2020)

mammals, such as hormone receptors, neurotransmitters, and enzymes, hence it is uncommon for a cell to location that does not have any active secondary metabolites (Rodrigues and Guedes 2006). Secondary metabolites include tannins, non-protein amino acids, alkaloids, saponins, lignin, glycosides, and other polyphenols. Among these, anthelmintic properties are most closely linked to tannins (Rodrigues and Guedes 2006). It is generally recognized that some proteins can be linked to polyphenol chemicals, particularly tannins. Tannins are divided into condensed and hydrolysable categories based on their molecular structures. Tropical forage grasses often contain these compounds, which are further classified into different groups, highlighting the potent anthelmintic properties of procyanidins and prodelfinidins (Baxter et al. 1997). The rumen of ruminants is where binding of procyanidins and prodelfinidins dietary proteins, particularly those high in proline, takes place. In the abomasum, the protein/tannin macromolecular complex is broken down due to the low pH, and the proteins are broken down and taken up in the digestive system. As a result, a diet high in protein strengthens the immune system's defense against dangerous substances, including nematodes, which is an indirect effect of these metabolites on helminth infections (Waterman et al. 2010). Because of their propensity to attach to the parasite's proteins, tannins have a direct action that alters the cuticle structure and degenerates the intestinal and muscular cells. Because of the metabolic changes brought on by the cuticle's structural breaking, these injuries may cause the worm to become less motile (Brunet et al. 2011). Destructuring the reproductive appendage of females might also hinder their ability to deliver eggs, and abnormalities in the front end can influence the parasite's feeding. The interaction of these metabolites with the L3 larvae's sheath results in another impact of tannins, which hinders their ability to penetrate the host's gastrointestinal tract by blocking their exsheathment (Hoste et al. 2012).

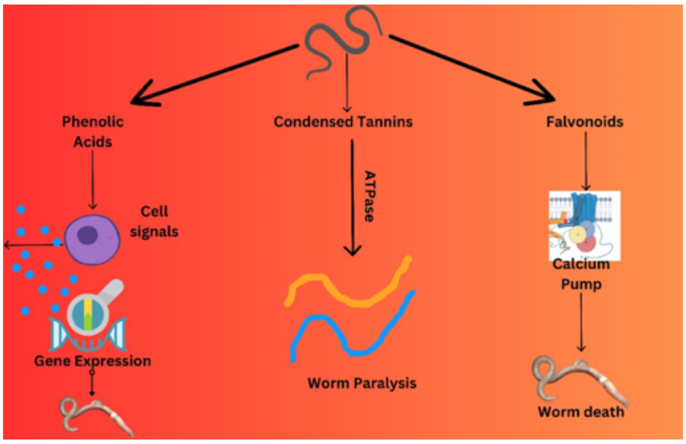

***Fig. 1**: Schematic representation of the mechanism of action and different pharmacological targets of plant extracts/compounds*

11. In vitro trials of the anthelmintic effectiveness of phytotherapy substances

Conducting in vitro studies of plant extracts, excluding host and environmental factors, and focusing solely on the target parasite and candidate plant, is the first stage in the process of confirming phytotherapeutic compounds. Numerous plant species have been assessed for their anthelmintic properties in this regard (Table 3). Much little research has been done on other livestock species, with the vast bulk of studies examining the impact of phytotherapeutic compounds on parasites concentrating on sheep. This study bias can be attributed to two factors: the higher pathogenicity of the parasites and the increasing frequency of reports of resistance (Borges and Borges 2016).

12. Limitations in use of plant-based medicines and future prospective

The use of plant-based medications to treat and prevent parasitic illnesses shows great promise. However, bioavailability can restrict their use. The main phytochemicals, such as flavonoids, glycosides, and tannins, are poorly soluble in water and lipids, which restricts their capacity to pass through biological membranes and causes inadequate absorption (Gao and Hu 2010). To get bioactive components, plants are also put through a variety of processes, including fermentation, distillation, purification, concentration, and extraction. The stability of active ingredients is questioned because they are subjected to oxidation

Table 3 Different plant species and their target parasitic life stage

Plant species	Target parasite	Life stage of parasite	Reference
Acacia baileyana	Cyathostominae sp.	Larvae	(Payne et al. 2013)
A. holorecicea	Cooperi sp. & *Haemonchus contorus*	L3 larvae	(Moreno et al. 2010)
A. meanrsnii	*H. contorus*	Eggs and larvae	(Yoshihara et al. 2014)
A. Melanoxylon	Cyathostominae sp.	Larvae	(Payne et al. 2013)
A. podalyrrifolia	Cyathostominae sp.	Larvae	(Waterman et al. 2010)
A. polycacantra	*Caenorhabiditis elegans*	Juveniles	(Payne et al. 2013)
A. salicina	*Cooperi asp.* and *H. placei*	L3 larvae	(Moreno et al. 2010)
Alectryon oleifolius	Cyathostominae sp.	Larvae	(Elghandour et al. 2023)
Allocasuarina torulosa	*H. placei*	L3 larvae	(Riley 2019)
Anogejssus lelocarpus	*C. elegans*	Juveniles	(Maroyi 2017)
Artemisia annua	*Bunostomum sp.*	Eggs and larvae	(Ekanem and Brisibe 2010)
Bridelia micranta	*C. elegans*	Juveniles	(Kevin et al. 2023)
B. micranta	*C. elegans*	Juveniles	(Waterman et al. 2010)
Cambretum nigricans	*C. elegans*	Juveniles	(Ignagli et al. 2024)
Casuarina cunninghamiana	*H. placei*	L3 larvae	(Moreno et al. 2010)
Citrus sinesis	*H. contorus*	Eggs and larvae	(Githiori et al. 2002)
Duboisia hopwood	Cyathostominae sp.	Larvae	(Payne et al. 2013)
Eucalyptus gomphocephala	Cyathostominae sp.	Larvae	(Ishaq 2014)
Flemingia vestita	Trematode and Cestoda	Adults	(Tandon et al. 1997)
Grewia bicolor	*C. elegans*	Juveniles	(Waterman et al. 2010)
Jatropha curcas	*H. contorus*	Eggs and larvae	(Monteiro et al. 2011)
Markhamia obtusifolia	*Trichostrongylus colubriformis*	Eggs	(Nchu et al. 2011)
Melaleuca quinquenervia	*H. contorus*	Eggs and larvae	(Githiori et al. 2002)
Melia azedarach	*H. contorus*	Eggs and larvae	(Kamaraj et al. 2010)
Petophorum africanum	*T. colubriformis*	Eggs and L1, L2 and L3 larvae	(Bizimenyera et al. 2006)
Phillyrea latifolia	*T. circuncincta*	Eggs and larvae	(Azaizeh et al. 2013)
Pistacia lentiscus	*Teladorsagia circuncincta*	Larvae	(Azaizeh et al. 2013)
Santalum spicatum	Cyathostominae sp.	Larvae	(Payne et al. 2013)
Strychnous spinosa	*C. elegans*	Juveniles	(Waterman et al. 2010)
Tabernaemontana citrifolia	*H. contorus*	Eggs, larvae, and adults	(Marie-Magdeleine et al. 2010)
Ziziphus mucronate	*C. elegans*	Juveniles	(Waterman et al. 2010)

and hydrolysis during these procedures (Rangari 2009). Additionally, plant products frequently deteriorate, especially when stored, which results in the loss of active ingredients and the creation of inactive metabolites (Thakur et al. 2011). Concerns about the safety of plant-based medications are becoming more prevalent as their use grows worldwide. Despite their widespread use and enticing potential, many plants have not yet been confirmed safe or poisonous. This results in a lack of awareness regarding their possible side effects and makes it challenging to determine the safest and most efficient treatments (Gao and Hu 2010).

The research discussed here supports more research into plants or plant derivatives as sources of innovative treatments for nematodal parasite infections. The studies do, however, also highlight a number of areas that need more research (Liu 2020). Future study is encouraged by the majority of the studies in literature to find the ideal dosage for maximizing the efficiency of the plants under investigation. It is also crucial to translate the findings of in vitro research into in vivo experiments. Furthermore, to prove the new compounds' safety and effectiveness, clinical trials involving successful animal experiments using either the compounds alone or in combination with well-established antiparasitic medications are necessary.

13. Conclusion

In order to find new medications and lead compounds, this study focused on research that assessed plants and plant products as nematode antiparasitic medication. Although there are few studies on some species of nematodes affecting animals, plants or their isolates have been tested against all prevalent parasites. Given the circumstances, the outcomes of this review offer insightful data about genetic structures in Table 1 that can guide the development of future research concerning procedures, dosages, and experimental setups. According to this review, nematode parasites are significantly affected by plants and chemicals resulting from plants both in vitro and in vivo. Broad-spectrum antiparasitic medications and many plant extracts have shown comparable benefits. Although this element needs to be examined, the traditional use of plants offers vital evidence for finding and creating synergistic medications.

Declarations

Funding: Not applicable

Conflict of interest: All authors declare no conflicts of interests

Acknowledgements: None

References

Abbas A, Abbas RZ, Khan MK, Raza MA, Mahmood MS, Saleemi MK, Sindhu ZUD. (2019). Anticoccidial effects of *Trachyspermum ammi* (Ajwain) in broiler chickens. Pakistan Veterinary Journal 39(2): 301-304. http://dx.doi.org/10.29261/pakvetj/2019.056

Abd-Elgawad MMM. (2024). Upgrading Strategies for Managing Nematode Pests on Profitable Crops. Plants (Basel) 13(11): 558. https://doi.org/10.3390/plants13111558

Acamovic T, Brooker J. (2005). Biochemistry of plant secondary metabolites and their effects in animals. Proceedings of the Nutrition Society 64(3): 403-412. https://doi.org/10.1079/pns2005449

Adduci I, Sajovitz F, Hinney B, Lichtmannsperger K, Joachim A, Wittek T, Yan S. (2022). Haemonchosis in sheep and goats, control strategies and development of vaccines against Haemonchus contortus. Animals 12(18): 2339. https://doi.org/10.3390/ani12182339

Ahmad K, Wahid Ullah QA, Adeel M, Fahad S. (2024). A cross-sectional study of prevalence of gastrointestinal parasites in captive wild animals in Pakistan zoological gardens. World's Veterinary Journal 14(2):234-241. https://dx.doi.org/10.54203/scil.2024.wvj29

Akhoundi M, Downing T, Votýpka J, Kuhls K, Lukeš J, Cannet A, Ravel C, Marty P, Delaunay P, Kasbari M, Granouillac B, Gradoni L, Sereno D. (2017). Leishmania infections: Molecular targets and diagnosis. Molecular Aspects of Medicine 57(1): 1–29. https://doi.org/10.1016/j.mam.2016.11.012

Azaizeh H, Halahleh F, Abbas N, Markovics A, Muklada H, Ungar E, Landau S. (2013). Polyphenols from *Pistacia lentiscus* and *Phillyrea latifolia* impair the exsheathment of gastro-intestinal nematode larvae. Veterinary Parasitology 191(1-2): 44-50. https://doi.org/10.1016/j.vetpar.2012.08.016

Baxter NJ, Lilley TH, Haslam E, Williamson MP. (1997). Multiple interactions between polyphenols and a salivary proline-rich protein repeat result in complexation and precipitation. Biochemistry 36(18): 5566-5577. https://doi.org/10.1021/bi9700328

Berhanu M, Gebeyaw DT, Kefale D, Kang Y. (2024). Overview of nematophagous fungi, isolation techniques, and their role in biological control of helminthic parasites: A literature review. Acta Entomology and Zoology 5(1): 133-143. https://doi.org/10.33545/27080013.2024.v5.i1b.133

Beveridge I, Gasser RB. (2014). Diversity in parasitic helminths of Australasian marsupials and monotremes: a molecular perspective. International Journal for Parasitology 44(12): 859-864. https://doi.org/10.1016/j.ijpara.2014.06.001

Biswas JK, Pramanik S, Kumar M. (2023). Fish parasites as proxy bioindicators of degraded water quality of River Saraswati, India. Environmental Monitoring and Assessment 195: 818. https://doi.org/10.1007/s10661-023-11411-6

Bizimenyera E, Githiori J, Eloff J, Swan G. (2006). In vitro activity of Peltophorum africanum Sond. (Fabaceae) extracts on the egg hatching and larval development of the parasitic nematode Trichostrongylus colubriformis. Veterinary Parasitology 142(3-4): 336-343. https://doi.org/10.1016/j.vetpar.2006.06.013

Borges DGL, Borges FDA. (2016). Plants and their medicinal potential for controlling gastrointestinal nematodes in ruminants. Nematoda 3(1): e92016. http://dx.doi.org/10.4322/nematoda.00916

Bouchtaoui EM, Fahr M, Smouni A, Azim K, Lahlali R, Mokrini F. (2024). Harnessing compost and vermicompost for sustainable and effective management of plant-parasitic nematodes in agricultural systems: A critical review. Physiological and Molecular Plant Pathology 63(1): 102363. https://doi.org/10.1016/j.pmpp.2024.102363

Branciari R, Ranucci D, Miraglia D, Valiani A, Veronesi F, Urban E, Lo Vaglio G, Pascucci L, Franceschini R. (2016). Occurrence of parasites of the genus Eustrongylides spp.(Nematoda: Dioctophymatidae) in fish caught in Trasimeno lake, Italy. Italian Journal of Food Safety, 5(4): 6130. https://doi.org/10.4081/ijfs.2016.6130

Bricarello PA, Longo C, da Rocha RA, Hotzel MJ. (2023). Understanding animal-plant-parasite interactions to improve the management of gastrointestinal nematodes in grazing ruminants. Pathogens 12(4): 531. https://doi.org/10.3390/pathogens12040531

Brunet S, Fourquaux I, Hoste H. (2011). Ultrastructural changes in the third-stage, infective larvae of ruminant nematodes treated with sainfoin (Onobrychis viciifolia) extract. Parasitology International, 60(4): 419-424. https://doi.org/10.1016/j.parint.2010.09.011

Cable J, Barber I, Boag B, Ellison AR, Morgan ER, Murray K, Booth M. (2017). Global change, parasite transmission and disease control: lessons from ecology. Philosophical Transactions of the Royal Society B: Biological Sciences 372(1719): 20160088. http://doi.org/10.1098/rstb.2016.0088

Callejon R, De Rojas M, Feliu C, Balao F, Marrugal A, Henttonen H, Cutillas C. (2012). Phylogeography of Trichuris populations isolated from different Cricetidae rodents. Parasitology 139(13): 1795-1812. http://doi.org/10.1017/S0031182012001114

Calsbeek R. (2009). Sex-specific adult dispersal and its selective consequences in the brown anole, Anolis sagrei. Journal of Animal Ecology 78(3): 617-624. https://doi.org/10.1111/j.1365-2656.2009.01527.x

Chandrakar S, Khutey JK, Roy S, Singh J, Mishra B, Jambagi K, Uppal M. (2020). Prevalence of gastrointestinal nematodes in captive wild carnivores of Chhattisgarh, India. International Journal of Current Microbiology and Applied Sciences 9(3): 609-615. https://doi.org/10.20546/ijcmas.2020.903.072

Chatterjee A, Singh N, Chanu WK, Singh CB, Nagaraj VA. (2022). Phytochemical screening, cytotoxicity assessment and evaluation of in vitro antiplasmodial and in vivo antimalarial activities of Mentha spicata L. methanolic leaf extract. Journal of Ethnopharmacology 298(1): 115636. https://doi.org/10.1016/j.jep.2022.115636

Cheraghipour K, Marzban A, Ezatpour B, Moradpour K, Nazarabad

VH. (2019). The role of onion (Allium cepa) in controlling parasitic diseases: a mini review. Herbal Medicines Journal 63(1): 175-180. https://doi.org/10.22087/hmj.v4i4.749

Chilton NB, Huby-Chilton F, Beveridge I, Smales LR, Gasser RB, Andrews RH. (2011). Phylogenetic relationships of species within the tribe Labiostrongylinea (Nematoda: Cloacinidae) from Australian marsupials based on ribosomal DNA spacer sequence data. Parasitology International 60(4): 381-387. https://doi.org/10.1016/j.parint.2011.06.012

Chilton NB, Shuttleworth MA, Huby-Chilton F, Koehler AV, Jabbar A, Gasser RB, & Beveridge I. (2017). Speciation in the genus Cloacina (Nematoda: Strongylida): species flocks and intra-host speciation. Parasitology 144(13): 1828-1840. https://doi.org/10.1017/S0031182017001238

Cole R, Viney M. (2018). The population genetics of parasitic nematodes of wild animals. Parasites & vectors 11(1): 590. https://doi.org/10.1186/s13071-018-3137-5

Deguine JP, Aubertot JN, Flor RJ, Lescourret F, Wyckhuys KA, Ratnadass A. (2021). Integrated pest management: good intentions, hard realities. A review. Agronomy for Sustainable Development 41(3): 38. https://doi.org/10.1007/s13593-021-00689-w

Di Cesare A, Otranto D, Latrofa MS, Veronesi F, Perrucci S, Lalosevic D, Traversa D. (2014). Genetic variability of Eucoleus aerophilus from domestic and wild hosts. Research in Veterinary Science 96(3): 512-515. https://doi.org/10.1016/j.rvsc.2014.03.018

Dogra NK, Kumar S, Kumar D. (2020). Vernonia anthelmintica (L.) Willd.: An ethnomedicinal, phytochemical, pharmacological and toxicological review. Journal of Ethnopharmacology 256: 112777. https://doi.org/10.1016/j.jep.2020.112777

Dunams-Morel DB, Reichard MV, Torretti L, Zarlenga DS, Rosenthal BM. (2012). Discernible but limited introgression has occurred where Trichinella nativa and the T6 genotype occur in sympatry. Infection, Genetics and Evolution 12(3): 530-538. https://doi.org/10.1016/j.meegid.2012.01.004

Ekanem AP, Brisibe EA. (2010). Effects of ethanol extract of Artemisia annua L. against monogenean parasites of Heterobranchus longifilis. Parasitology Research 106(1): 1135-1139. https://doi.org/10.1007/s00436-010-1787-0

El Menyiy N, Mrabti HN, El Omari N, Bakili AE, Bakrim S, Mekkaoui M, Balahbib A, Amiri-Ardekani E, Ullah R, Alqahtani AS, Shahat AA, Bouyahya A. (2022). Medicinal uses, phytochemistry, pharmacology, and toxicology of Mentha spicata. Evidence-Based Complementary and Alternative Medicine 2022(1): 7990508. https://doi.org/10.1155/2022/7990508

Elghandour MMMY, Maggiolino A, Vázquez-Mendoza P, Alvarado-Ramírez ER, Cedillo-Monroy J, De Palo P, Salem AZM. (2023). Moringa oleifera as a natural alternative for the control of gastrointestinal parasites in equines: a review. *Plants*, 12(9): 1921. https://doi.org/10.3390/plants12091921

Elhady A, Alghanmi L, Abd-Elgawad MM, Heuer H, Saad MM, Hirt H. (2024). Plant-parasitic nematode research in the arid desert landscape: a systematic review of challenges and bridging interventions. Frontiers in Plant Science 15(1): 1432311. https://doi.org/10.3389/fpls.2024.1432311

Emery DL, Hunt PW, Le Jambre LF. (2016). Haemonchus contortus: the then and now, and where to from here? International Journal for Parasitology 46(12): 755-769. https://doi.org/10.1016/j.ijpara.2016.07.001

Falk BG, Perkins SL. (2013). Host specificity shapes population structure of pinworm parasites in C aribbean reptiles. Molecular ecology 22(17): 4576-4590. https://doi.org/10.1111/mec.12410

Fallah FJ, Khara H, Rohi JD, Sayadborani M. (2015). Hematological parameters associated with parasitism in pike, Esox lucius caught from Anzali wetland. Journal of Parasitic Diseases 39(1): 245-248. https://doi.org/10.1007/s12639-013-0329-y

Flores-Pérez I, Hallal-Calleros C, Cervantes-Pacheco BJ, Alba-Hurtado F, Orihuela A, Castro-del-Campo N, Barajas-Cruz R. (2019). Behavioural and productive response to experimental infection with stage 3 larvae of Haemonchus contortus in feedlot bull-calves. Experimental Parasitology 197: 1-8. https://doi.org/10.1016/j.exppara.2018.12.005

Gao S, Hu M. (2010). Bioavailability challenges associated with development of anti-cancer phenolics. Mini Reviews in Medicinal Chemistry 10(6): 550-567. https://doi.org/10.2174/138955710791384081

Gianechini LS. (2024). Addressing anthelmintic resistance in parasitic nematodes by investigating the potential of a novel drug target and the dynamics of benzimidazole selection in a model parasitic nematode. PhD thesis, University of Georgia, Athens, Georgia, USA.

Githiori JB, Höglund J, Waller PJ, Baker RL. (2002). Anthelmintic activity of preparations derived from Myrsine africana and Rapanea melanophloeos against the nematode parasite, Haemonchus contortus, of sheep. Journal of ethnopharmacology 80(2-3): 187-191. https://doi.org/10.1016/s0378-8741(02)00030-2

Guerra RO, do Carmo Neto JR, da Silva PEF, Borges YLL, Franco PIR, Machado JR, da Silva MV. (2023). Cannabis sativa: A look at protozoa, helminths, insect vectors, and pests. Fitoterapia 166: 105467. https://doi.org/10.1016/j.fitote.2023.105467

Harhay MO, Horton J, Olliaro PL. (2010). Epidemiology and control of human gastrointestinal parasites in children. Expert review of anti-infective therapy 8(2): 219-234. https://doi.org/10.1586/eri.09.119

Heppert JK, Ransone EM, Grossman AS, Mauer TJ, Goodrich-Blair H. (2022). Nematodes as models for symbiosis. In: Glazer I, Shapiro-llan D, Sternberg PW, editors, Nematodes as model organisms. CABI, Wallingford, Oxfordshire, UK. Pp. 82-126. https://doi.org/10.1079/9781789248814.0005

Honcharov SL, Soroka NM, Halat MV, Dubovyi AI, Zhurenko VV, Halushko IA. (2022). Distribution of the nematodes of the genus Eustrongylides (Nematoda, Dioctophymatidae) in the world. Regulatory Mechanisms in Biosystems 13(1): 73-79. https://doi.org/10.15421/022210

Hoste H, Martinez-Ortiz-De-Montellano C, Manolaraki F, Brunet S, Ojeda-Robertos N, Fourquaux I, Sandoval-Castro C. (2012). Direct and indirect effects of bioactive tannin-rich tropical and temperate legumes against nematode infections. Veterinary Parasitology 186(1-2): 18-27. https://doi.org/10.1016/j.vetpar.2011.11.042

Hussain A, Khan MN, Iqbal Z, Sajid MS, Khan MK. (2011). Anthelmintic activity of Trianthema portulacastrum L. and Musa paradisiaca L. against gastrointestinal nematodes of sheep. Veterinary Parasitology 179(1-3): 92-99. https://doi.org/10.1016/j.vetpar.2011.02.022

Ignagali BB, Galani Tietcheu BR, Betrosse T, Kamaya B, Ndjonka D. (2024). In Vitro Filaricidal Properties of Aqueous Extracts of Combretum nigricans (Combretaceae) on Onchocerca ochengi

(Onchocercidae). Journal of Parasitology Research 2024(1): 2119056. https://doi.org/10.1155/2024/2119056

Ishaq L. (2014). The role of mycorrhizal fungi in Eucalyptus gomphocephala (Tuart) health. PhD thesis, Murdoch University, Perth, Western Australia.

Jabbar A, Beveridge I, Mohandas N, Chilton NB, Littlewood DTJ, Jex AR, & Gasser RB. (2013). Analyses of mitochondrial amino acid sequence datasets support the proposal that specimens of Hypodontus macropi from three species of macropodid hosts represent distinct species. BMC Evolutionary Biology 13: 259. https://doi.org/10.1186/1471-2148-13-259

Jones IJ. (2020). Human health and environmental sustainability in pathogenic landscapes: Feedbacks and solutions. PhD Thesis, Stanford University, Stanford, California, USA.

Jorge F, Roca V, Perera A, Harris DJ, Carretero MA. (2011). A phylogenetic assessment of the colonisation patterns in Spauligodon atlanticus Astasio-Arbiza et al. 1987 (Nematoda: Oxyurida: Pharyngodonidae), a parasite of lizards of the genus Gallotia Boulenger: no simple answers. Systematic parasitology 80(1): 53-66. https://doi.org/10.1007/s11230-011-9311-1

Juillard LQ, Ramp D. (2022). The Impacts of drought on the health and demography of Eastern grey kangaroos. Animals 12(3): 256. https://doi.org/10.3390/ani12030256

Kamaraj C, Rahuman AA, Bagavan A, Mohamed MJ, Elango G, Rajakumar G, Zahir AA, Santhoshkumar T, Marimuthu S. (2010). Ovicidal and larvicidal activity of crude extracts of Melia azedarach against Haemonchus contortus (Strongylida). Parasitology Research 106(1): 1071-1077. https://doi.org/10.1007/s00436-010-1750-0

Kapinder DN, Verma AK. (2023). Drug resistance in helminth parasites: Role of plant-based natural therapeutics. In: Singh A, Rathi B, Verma AK, Singh IK, editors, Natural product based drug discovery against human parasites: Opportunities and Challenges. Springer Nature, Singapore. Pp. 553-579. https://doi.org/10.1007/978-981-19-9605-4_25

Kavetska KM, Pilarczyk B, Królaczyk K. (2012). Stomach nematodes of wild ducks (subfamily anatinae) wintering in the north-western Poland. Journal of Veterinary Research 56(1): 27-31. https://doi.org/10.2478/v10213-012-0005-5

Kevin TDA, Cedric Y, Nadia NAC, Sidiki NNA, Azizi MA, Guy-Armand GN, Sandra TNJ, Christian MN, Geraldine ESE, Romeo TT, Payne VK, Gustave LL. (2023). Antiplasmodial, antioxidant, and cytotoxic activity of Bridelia micrantha a cameroonian medicinal plant used for the treatment of malaria. BioMed Research International 2023: 1219432. https://doi.org/10.1155/2023/1219432

KhokharVoytas A, Shahbaz M, Maqsood MF, Zulfiqar U, Naz N, Iqbal UZ, Sara M, Aqeel M, Khalid N, Noman A, Zulfiqar F, Al Syaad KM, AlShaqhaa MA. (2023). Genetic modification strategies for enhancing plant resilience to abiotic stresses in the context of climate change. Functional & Integrative Genomics 23(3): 283. https://doi.org/10.1007/s10142-023-01202-0

King IL, Li Y. (2018). Host–parasite interactions promote disease tolerance to intestinal helminth infection. Frontiers in Immunology 9: 2128. https://doi.org/10.3389/fimmu.2018.02128

La Rosa G, Marucci G, Rosenthal BM, Pozio E. (2012). Development of a single larva microsatellite analysis to investigate the population structure of Trichinella spiralis. Infection, Genetics and Evolution 12(2): 369-376. https://doi.org/10.1016/j.meegid.2012.01.008

Leon-Regagnon V. (2019). Helminths of the Eurasian marsh frog, Pelophylax ridibundus (Pallas, 1771)(Anura: Ranidae), from the Shiraz region, southwestern Iran. Helminthologia 56(3): 261. https://doi.org/10.2478/helm-2019-0022

Liu M, Panda SK, Luyten W. (2020). Plant-based natural products for the discovery and development of novel anthelmintics against nematodes. Biomolecules 10(3): 426. https://doi.org/10.3390/biom10030426

Long ES, Diefenbach DR, Rosenberry CS, Wallingford BD. (2008). Multiple proximate and ultimate causes of natal dispersal in white-tailed deer. Behavioral Ecology 19(6): 1235-1242. https://doi.org/10.1093/beheco/arn082

Luz SDM, Souza Filho APDS, Guilohn G, Vilhena KDSDS. (2010). Allelopathic activity of chemical substances isolated from Acacia mangium and its variations in function of PH. Planta Daninha 28(3): 479-487. https://doi.org/10.1590/S0100-83582010000300004

Lynsdale CL, Mon NO, Franco dos Santos DJ, Aung HH, Nyein UK, Htut W, Lummaa V. (2020). Demographic and reproductive associations with nematode infection in a long-lived mammal. Scientific Reports 10(1): 9214. https://doi.org/10.1038/s41598-020-66075-w

Marie-Magdeleine C, Mahieu M, D'Alexis S, Philibert L, Archimede H. (2010). In vitro effects of Tabernaemontana citrifolia extracts on Haemonchus contortus. Research in Veterinary Science 89(1): 88-92. https://doi.org/10.1016/j.rvsc.2010.01.002

Maroyi A. (2017). Ethnopharmacology and therapeutic value of Bridelia micrantha (Hochst.) Baill. in tropical Africa: a comprehensive review. Molecules 22(9): 1493. https://doi.org/10.3390/molecules22091493

Metin S, Didinen BI, Boyacı YO, Kubilay A, Emre N, Didinen H, Emre Y. (2014). Occurrence of Eustrongylides excisus jägerskiöld, 1909 larvae (Nematoda: Dioctophymatidae) in Pikeperch (Sander lucioperca, L.) in lake Eğirdir. Süleyman Demirel Üniversitesi Eğirdir Su Ürünleri Fakültesi Dergisi 10(1): 20-24. https://doi.org/10.22392/egirdir.246371

Monteiro MVB, Bevilaqua CM, Morais SM, Machado LKA, Camurça-Vasconcelos ALF, Campello CC, Mesquita MDA. (2011). Anthelmintic activity of Jatropha curcas L. seeds on Haemonchus contortus. Veterinary parasitology 182(2-4): 259-263. https://doi.org/10.1016/j.vetpar.2011.04.010

Moreno F, Gordon I, Wright A, Benvenutti M, & Saumell C. (2010). In vitro anthelmintic effect of plant extracts against infective larvae of ruminants gastrointestinal nematode parasites. Archivos de Medicina Veterinaria 42(3): 155-163. https://doi.org/10.4067/S0301-732X2010000300006

Mukherjee A, Kar I, Patra AK. (2023). Understanding anthelmintic resistance in livestock using "omics" approaches. Environmental Science and Pollution Research 30(60): 125439-125463. https://doi.org/10.1007/s11356-023-31045-y

Muzzall PM (1999). Nematode parasites of yellow perch, Perca flavescens, from the Laurentian Great Lakes. Journal of the Helminthological Society Washington 66(2): 115-122.

Narr LL, O'Donnell JG, Libster B, Alessi P, Abraham D. (1996). Eustrongylidiasis--a parasitic infection acquired by eating live minnows. Journal of Osteopathic Medicine 96(7): 400-402. https://doi.org/10.7556/jaoa.1996.96.7.400

Navrátilová M. (2024). Anthelmintics in the environment: circulation, metabolism and effects. PhD thesis, Charles University, Prague, Czechia. http://hdl.handle.net/20.500.11956/192396

Nchu F, Githiori J, McGaw LJ, Eloff JN. (2011). Anthelmintic and cytotoxic activities of extracts of Markhamia obtusifolia Sprague (Bignoniaceae). Veterinary Parasitology 183(1-2): 184-188. https://doi.org/10.1016/j.vetpar.2011.06.017

Nieberding CM, Durette-Desset MC, Vanderpoorten A, Casanova JC, Ribas A, Deffontaine V, Michaux JR. (2008). Geography and host biogeography matter for understanding the phylogeography of a parasite. Molecular Phylogenetics and Evolution 47(2): 538-554. https://doi.org/10.1016/j.ympev.2008.01.028

Nouri F, Nourollahi-Fard SR, Foroodi HR, Sharifi H. (2016). In vitro anthelmintic effect of Tobacco (Nicotiana tabacum) extract on parasitic nematode, Marshallagia marshalli. Journal of Parasitic Diseases 40(3): 643-647. https://doi.org/10.1007/s12639-014-0550-3

Oguntibeju OO. (2019). Antidiabetic, anti-inflammatory, antibacterial, anti-helminthic, antioxidant and nutritional potential of Musa paradisiaca. Asian Journal of Pharmaceutical and Clinical Research 12(10): 9-13. http://dx.doi.org/10.22159/ajpcr.2019.v12i10.34239

Otranto D, Deplazes P. (2019). Zoonotic nematodes of wild carnivores. International Journal for Parasitology: Parasites and Wildlife 9: 370-383. https://doi.org/10.1016/j.ijppaw.2018.12.011

Palomba M, Santoro M, Alburqueque RA, Cipriani P, Mattiucci S. (2021). First molecular detection of the parasites Molicola uncinatus and Hepatoxylon trichiuri (Cestoda: Trypanorhyncha) infecting the silver scabbardfish Lepidopus caudatus from the Central Mediterranean Sea: Implications for the seafood quality and safety. Food Control 122: 107807. https://doi.org/10.1016/j.foodcont.2020.107807

Panayotova-Pencheva MS. (2024). Control of helminth infections in captive herbivores: An overview of experience. Journal of Zoological and Botanical Gardens 5(4): 641-667. https://doi.org/10.3390/jzbg5040043

Patwardhan B. (2014). Bridging Ayurveda with evidence-based scientific approaches in medicine. EPMA Journal, 5(1): 19. https://doi.org/10.1186/1878-5085-5-19

Payne S, Kotze A, Durmic Z, Vercoe P. (2013). Australian plants show anthelmintic activity toward equine cyathostomins in vitro. Veterinary Parasitology 196(1-2): 153-160. https://doi.org/10.1016/j.vetpar.2013.01.012

Peacock D, Croxford A, Iannella A, Kovaliski J, Lavazza A, Cooke B, Spratt D, Strive T, Taggart D, Campbell, Robinson S, Sawyers E. (2022). Using genetic analysis to determine the distribution, prevalence, and diversity of Eimeria species in pest rabbits (Oryctolagus cuniculus) in Australia. Parasitology International 91: 102642. https://doi.org/10.1016/j.parint.2022.102642

Pontes T, D'Amelio S, Costa G, Paggi L. (2005). Molecular characterization of larval anisakid nematodes from marine fishes of Madeira by a PCR-based approach, with evidence for a new species. Journal of Parasitology 91(6): 1430-1434. https://doi.org/10.1645/GE-565R1.1

Postema EG. (2023). integrating multiple pressures on antipredator color traits: a conceptual framework, with experimental evidence from swallowtail caterpillars (Papilionidae). PhD thesis, University of California, Davis, USA.

Pybus MJ, Wood ME, Fox KA, Munk BA. (2023). Diseases and Parasites. In: Heffelfinger JR, Krausman PR, editors, Ecology and management of Black-tailed and Mule deer of North America. CRC Press, Boca Raton, Florida, USA. Pp. 103-124.

Quiroz-Martinez B, Salgado-Maldonado G. (2013). Patterns of distribution of the helminth parasites of freshwater fishes of Mexico. Plos One 8(1): e54787. https://doi.org/10.1371/journal.pone.0054787

Ranasinghe S, Armson A, Lymbery AJ, Zahedi A, Ash A. (2023). Medicinal plants as a source of antiparasitics: an overview of experimental studies. Pathogens and Global Health 117(6): 535-553. https://doi.org/10.1080/20477724.2023.2179454

Rangari VD. (2009). Pharmacognosy & phytochemistry, volume 1. Career publications, Nashik, Maharastra, India.

Riley IT. (2019). A case for assessing Allocasuarina and Casuarina spp. for use in agroecosystem improvement in semi-arid areas with a focus on Central Anatolia, Turkey. Frontiers of Agricultural Science and Engineering 8(4): 568-582. https://doi.org/10.15302/J-FASE-2019270

Rodrigues A, Guedes ML. (2006). Utilização de plantas medicinais no Povoado Sapucaia, Cruz das Almas–Bahia. Revista Brasileira de Plantas Medicinais 8(2): 1-7.

Rothmann W, de Waal PJ. (2017). Diversity of *Spirocerca lupi* in domestic dogs and black-backed jackals (*Canis mesomelas*) from South Africa. Veterinary Parasitology 244: 59-63. https://doi.org/10.1016/j.vetpar.2017.07.026

Rupprecht CE, Mshelbwala PP, Reeves RG, Kuzmin IV. (2023). Rabies in a postpandemic world: resilient reservoirs, redoubtable riposte, recurrent roadblocks, and resolute recidivism. Animal Diseases 3(1): 15. https://doi.org/10.1186/s44149-023-00078-8

Saad AM, Salem HM, El-Tahan AM, El-Saadony MT, Alotaibi SS, El-Shehawi AM, El-Mageed TAA, Taha AE, Alkahtani MA, Ahmed AE, Swelum AA. (2022). Biological control: An effective approach against nematodes using black pepper plants (Piper nigrum L.). Saudi Journal of Biological Sciences 29(4): 2047-2055. https://doi.org/10.1016/j.sjbs.2022.01.004

Sanhokwe M, Mupangwa J, Masika PJ, Maphosa V, Muchenje V. (2016). Medicinal plants used to control internal and external parasites in goats. Onderstepoort Journal of Veterinary Research 83(1): 1016. https://doi.org/10.4102/ojvr.v83i1.1016

Santos MJ, Henriques R, Fernandes S, Rodrigues R. (1999). Delimitation of affinity regions in western Europe for the application of the regional drought model to annual precipitation data. ARIDE Tech. Report no. 4, Portuguese Institute for Water, Lisbon, Portugal.

Singh M, Pandey N, Sharma O. (2023). IPM concept and strategies for sustainable agriculture. In: Singh DP, editor, Integrated pest management in diverse cropping systems. Apple Academic Press, New York, USA.

Sloboda M, Mihalca AD, Falka I, Petrželková KJ, Carlsson M, Ghira I, Modrý D. (2010). Are gobiid fish more susceptible to predation if parasitized by Eustrongylides excisus? An answer from robbed snakes. Ecological Research 25(1): 469-473. https://doi.org/10.1007/s11284-009-0676-4

Sokolov SG, Moshu AJ. (2014). Goussia obstinata sp. (Sporozoa: Eimeriidae), a new coccidian species from intestines of the Amur sleeper Perccottus glenii Dybowski, 1877 (Perciformes: Odontobutidae). Parazitologiia 48(5): 382-392.

Sol D, Jovani R, Torres J. (2003). Parasite mediated mortality and host immune response explain age-related differences in blood parasitism in birds. Oecologia 135(4): 542-547. https://doi.org/10.1007/s00442-003-1223-6

Soliman AA, Khattab AE, Ali HE, Elsebaay MM. (2023). Impacts of some garlic extracts on tomato growth and physio-bio chemistry under infection of root-knot nematode, Meloidogyne incognita. Egyptian Academic Journal of Biological Sciences, F. Toxicology & Pest Control 15(1): 79-86. https://doi.org/10.21608/eajbsf.2023.291960

Song J, Wang H, Li S, Du C, Qian P, Wang W, Shen M, Zhang Z, Zhou J, Zhang Y, Li C, Hao Y, Dong Y. (2024). The genetic diversity of Oncomelania hupensis robertsoni, intermediate hosts of Schistosoma japonicum in hilly regions of China, using microsatellite markers. Parasites & Vectors 17(1): 147. https://doi.org/10.1186/s13071-024-06227-3

Stear M, Bishop S, Henderson N, Scott I. (2003). A key mechanism of pathogenesis in sheep infected with the nematode Teladorsagia circumcincta. Animal Health Research Reviews 4(1): 45-52. https://doi.org/10.1079/ahrr200351

Stear M, Preston S, Piedrafita D, Donskow-Łysoniewska K. (2023). The Immune Response to Nematode Infection. International Journal of Molecular Sciences 24(3): 2283. https://doi.org/10.3390/ijms24032283

Subekti S, Kurniawan MR, Sudjarwo SA. (2020). Identification and prevalence of infection of helminth in the gastrointestinal tract swamp eel (Synbranchus bengalensis) which is marketed in Surabaya, East Java. IOP Conference Series: Earth and Environmental Science; 2nd International Conference on Fisheries and Marine Science, 26th September, Surabaya, Indonesis, 441(1): 012146. https://doi.org/10.1088/1755-1315/441/1/012146

Sukee T, Beveridge I, Jabbar A. (2018). Molecular and morphological characterisation of Pharyngostrongylus kappa Mawson, 1965 (Nematoda: Strongylida) from Australian macropodid marsupials with the description of a new species, P. patriciae n. sp. Parasites & Vectors 11(1): 271. https://doi.org/10.1186/s13071-018-2816-6

Svazas S, Chukalova N, Grishanov G, Pūtys Z, Sruoga A, Butkauskas D, Prakas P. (2011). The role of Great Cormorant (Phalacrocorax carbo sinensis) for fish stock and dispersal of helminths parasites in the Curonian Lagoon area. Veterinarija Ir Zootechnika 55(77): 79-87.

Tandon V, Pal P, Roy B, Rao H, Reddy K. (1997). In vitro anthelmintic activity of root-tuber extract of Flemingia vestita, an indigenous plant in Shillong, India. Parasitology Research 83(3): 492-498. https://doi.org/10.1007/s004360050286

Thakur L, Ghodasra U, Patel N, Dabhi M. (2011). Novel approaches for stability improvement in natural medicines. Pharmacognosy Reviews 5(9): 48-54. https://doi.org/10.4103/0973-7847.79099

Thompson R. (2023). Zoonotic helminths–why the challenge remains. Journal of Helminthology 97(1): e21. https://doi.org/10.1017/S0022149X23000020

Wasimuddin BJ, Ribas A, Baird SJ, Piálek J, Goüy de Bellocq J. (2016). Testing parasite 'intimacy': the whipworm Trichuris muris in the European house mouse hybrid zone. Ecology and Evolution 6(9): 2688-2701. https://doi.org/10.1002/ece3.2022

Waterman C, Smith RA, Pontiggia L, DerMarderosian A. (2010). Anthelmintic screening of Sub-Saharan African plants used in traditional medicine. Journal of Ethnopharmacology 127(3): 755-759. https://doi.org/10.1016/j.jep.2009.11.025

Wink M. (2008). Evolutionary advantage and molecular modes of action of multi-component mixtures used in phytomedicine. Current Drug Metabolism 9(10): 996-1009. https://doi.org/10.2174/138920008786927794

Xiong F, Li WX, Wu SG, Zou H, Wang, GT. (2013). Molecular phylogeny and host specificity of the larval Eustrongylides (Nematoda: Dioctophmidae) from freshwater fish in China. The Journal of Parasitology 99(1): 137-144. https://doi.org/10.1645/GE-3163.1

Yongwa G, Ngnoda BFNF, Ndjonka D, Saotoing P. (2020). In vitro anthelmintic activity of aqueous and ethanolic extract of Senna italica (Caesalpiniaceae) on three-stages of Haemonchus contortus. Journal of Pharmaceutical Research International 32(3): 25-34. https://doi.org/10.9734/jpri/2020/v32i330411

Yoshihara E, Minho AP, Cardim ST, Tabacow VBD, Yamamura MH. (2014). In vitro ovicidal and larvicidal activity of condensed tannins on gastrointestinal nematode infestations in sheep (Ovis aries). Semina: Ciências Agrárias 35(6): 3173-3180. https://doi.org/10.5433/1679-0359.2014v35n6p3173

Zhang S, Huang G, Li L, Liu X, Tang X, Suo X. (2021). Morphological and phylogenetic analysis of Eustrongylides sp. and Gnathostoma spinigerum parasitizing the Asian swamp eel Monopterus albus in China. Pathogens 10(6): 711. https://doi.org/10.3390/pathogens10060711

Ziegler AF, Welte SC, Miller EA, Nolan TJ. (2000). Eustrongylidiasis in eastern great blue herons (Ardea herodias). Avian Diseases 44(2): 443-448.

Citation

Sajid M, Fatima A, Mehmood T, Sirajvi MMA, Manzoor M, Zohaib M, Iqbal UZ, Khan AMA. (2025). Prevalence of nematodes in wildlife and livestock animals and their control by medicinal plants. Letters in Animal Biology 05(1): 30 – 41.