Archives of Agriculture and Environmental Science 8(2): 250-258 (2023)
https://doi.org/10.26832/24566632.2023.0802023

e-ISSN: 2456-6632

This content is available online at AESA

Archives of Agriculture and Environmental Science

Journal homepage: journals.aesacademy.org/index.php/aaes

REVIEW ARTICLE

# Prevalence of potato cyst (*Globodera* spp.) nematode and potato root-knot (*Meloidogyne* spp.) nematode in Kenya and potential management strategies: A review

**Hillary M.O. Otieno**

Department of Plant Science and Crop Protection, University of Nairobi, Kenya
E-mail: hillarymoo@yahoo.com

**ARTICLE HISTORY**

Received: 22 April 2023
Revised received: 03 June 2023
Accepted: 12 June 2023

**Keywords**

Cyst nematodes
*Globodera rostochiensis*
*Globodera pallida*
Integrated pest management
*Meloidogyne* spp.
Root-knot nematodes

**ABSTRACT**

Nematodes are very diverse and could be free-living or plant parasite species. Amongst the existing categories, the most aggressive ones are the root lesion nematode (*Pratylenchus spp.*), root-knot nematode (*Meloidogyne spp.*), and cyst nematode (*Globodera spp.*). These categories affect over 2000 susceptible crop species causing varying yield losses, reaching 100% under heavy infestations in potatoes. The common root-knot nematode and cyst nematode hosts include tomato (*Solanum lycopersicum*), African eggplant (*Solanum aethiopicum*), eggplant (*Solanum melongena*), and other solanaceous plants, including *Physalis spp.*, *Datura spp.*, *Hyoscyamus spp.*, *Physoclaina* spp., *Salpiglossis* spp., and *Saracha* spp. These nematodes are disseminated mainly through irrigation water, rainfall runoffs, infested soil particles, commercial seed potato tubers, contaminated footwear, animal hooves, farm implements, and machinery. Effective control of nematodes requires farmers to practice integrated nematode management systems with a combination of at least two management practices. Several cultural and agronomic practices have shown some decent levels of efficacy, thus recommended for adoption. Timely application of these practices is critical for achieving better outcomes. Among the management strategies, applying nematicides is the most effective in the short term. It is important to be cautious when using these chemicals, as they pose significant risks to humans and the environment. Again, these products are costly, especially those within moderate to low toxicity, making them unsustainable and out of reach for most small-scale farmers.



**Citation of this article:** Otieno, H. M. O. (2023). Prevalence of potato cyst (*Globodera* spp.) nematode and potato root-knot (*Meloidogyne* spp.) nematode in Kenya and potential management strategies: A review. *Archives of Agriculture and Environmental Science*, *8* (2), 250-258, https://dx.doi.org/10.26832/24566632.2023.0802023

## INTRODUCTION

Nematodes are diverse, microscopic multicellular animals that could be free-living or plant-parasitic species and are found almost everywhere. They parasitize a wide range of monocot and dicot plant species, causing substantial annual yield losses. Nematodes can be broadly categorized into several groups. Out of the groups, cyst nematode (CN: *Globodera spp*), root-knot nematode (RKN: *Meloidogyne spp.*), and root lesion nematode (RLN: *Pratylenchus spp.*) are the most lethal, attacking and causing significant crop yield losses (Abrantes *et al.*, 2023). Each of the three categories has several nematode species. Amongst these categories, RKN and CN are the most aggressive ones affecting over 2000 susceptible crop species (Moens *et al.*, 2007; Abrantes *et al.*, 2023). The genus *Meloidogyne* comprises about 100 species, with *M.arenaria*, *M. incognita*, *M. hapla*, and *M. javanica* considered the main species (Elling, 2013). Twenty-two out of the 100 identified *Meloidogyne* species occur in Africa, posing a significant threat to crop production in small-holder farms (Karuri *et al.*, 2017). In Kenya, *M. incognita*, *M.javanica*, *M. hapla*, *M. africana*, *M. acronea*, and *M. kikuyensis* are the most commonly reported species (Van den Berg *et al.*, 2001; Onkendi *et al.*, 2014; Karuri *et al.*, 2017). On the other hand, two types of CN species are prevalent in Kenya: *Globodera rostochiensis* and *Globodera pallida* (Mwangi *et al.*, 2015; Mburu *et al.*, 2018; Mburu *et al.*, 2020).

These parasitic nematodes are limiting factors for the production of potatoes, one of the most important food crops in the region and the world (Karuri *et al*., 2017; Otieno and Mageto, 2021; Mbiyu *et al*., 2022). The losses in potatoes are characterized by decreased yields, physical and chemical changes in tubers, poor tuber quality, and malformations, which make them unmarketable (Pinheiro *et al*., 2015; Medina *et al*., 2016). These yield losses vary from low to 70% and to a complete loss in some cases (Sparkes, 2013; Mburu *et al*., 2020; Mbiyu *et al*., 2022). The levels of damages and losses, however, depend on a combination of factors, such as population build-up of the pest, number of nematode generations per year, length of potato growing season, time of planting, soil moisture, soil temperature, soil structure, and host factors (Greco, 1993; Gomes and Souza, 2003; Sparkes, 2013). For instance, according to Brown and Sykes (1983), a loss of approximately 2.75 tons per hectare of potatoes can occur for every 20 viable potato CN eggs per gram of soil. This translates to losses of 4.2-21.8 t/ha (Mburu *et al*., 2020), equivalent to a relative yield loss of 6.3% to 80.5% during the long rain season and from 5.5% to 73.3% during the short rain season in Kenya (Mbiyu *et al*., 2022). Establishing a national management strategy for mitigating RKN and CN threats in potato production requires determining the extent of infestations, identifying the geographical distributions, and assessing the various agronomic and social factors contributing to their spread. This information is crucial in devising and implementing effective measures to manage RKN and CN.

## PREVALENCE OF NEMATODES IN KENYA

In Kenya, *M. incognita*, *M.javanica*, *M. hapla*, *M. africana*, *M. acronea*, and *M. kikuyensis* are the most commonly occurring RKN species (Van den Berg *et al*., 2001; Onkendi *et al*., 2014; Karuri *et al*., 2017), while *Globodera rostochiensis* and *Globodera pallida* are the most detected species of potato CN (Mburu *et al*., 2020). These species affect potatoes and other crops almost in all growing areas. In Kenya, the first *Globodera rostochiensis* occurrence was detected and reported in Nyandarua County by Mwangi *et al*. (2015), while *Globodera pallida* were first detected and reported in Nyandarua County by Mburu *et al*. (2018). These parasites then spread to several other potato-producing areas across the country. Between 2016 and 2018, Mburu *et al*. (2020) conducted a nationwide survey to determine the diversity, prevalence, and distribution of PCN species. They detected PCN in all 20 potato-producing counties, with Nyeri County presenting the lowest incidence (53%), while Trans Nzoia, Taita Taveta, and West Pokot counties had 100% incidence (Figure 1). The extent of the impact and spread of this nematode can be observed even under certified fields, where, of the 22 certified seed farms sampled, 82% of farms were infested with PCN (Mburu *et al*., 2020). Regarding composition, Mburu *et al*. (2020) found that most PCN populations, 99.9%, were *G. rostochiensis*, while *G. pallida* were retrieved from one field. Haukeland (2016) has confirmed even much higher infestation in over 80% of all potato farms in Kenya. The RKN prevalence is also widespread in the country; in the Central Highlands (Wanjohi *et al*., 2018), *M. incognita* infestation in Central, Nyanza, Western, Eastern, Rift Valley, and Coast regions (Figure 2) (Hallmann and Sikora, 1994; Mbogoh *et al*., 2013; Karuri *et al*., 2017). In fact, the widespread and high infestations by root-knot nematodes, e.g., *M. incognita*, and *M. javanica* had been reported a long time ago in Kenya (Whitehead and Kariuki, 1960; Kanyagia, 1979).

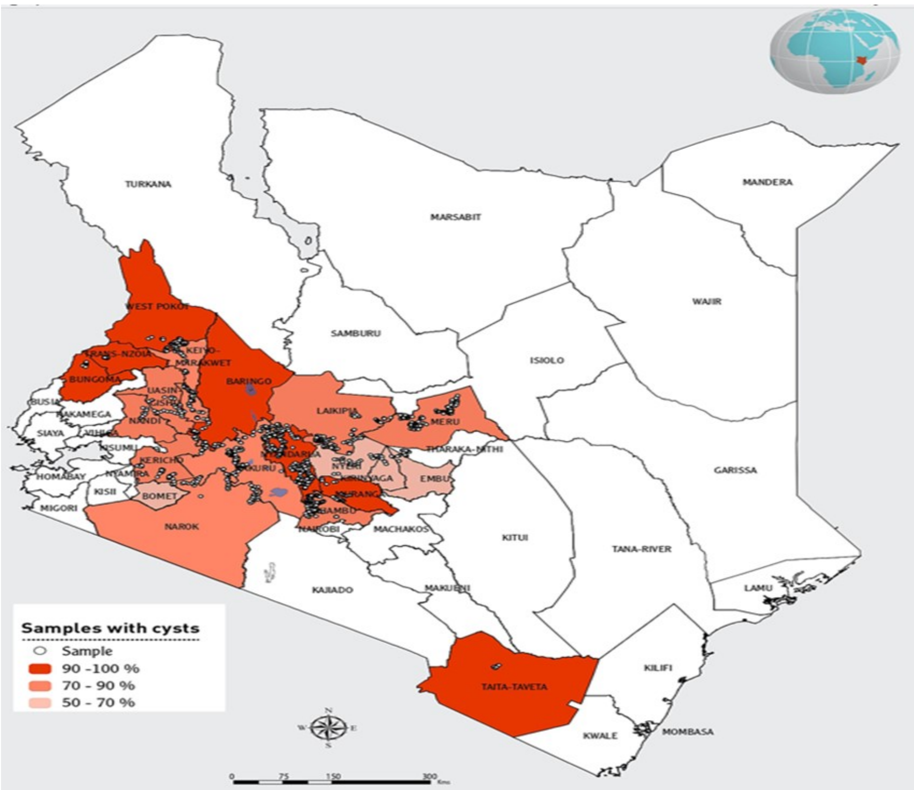


**Figure 1.** *Potato cyst nematode distribution and incidence level classification across the 20 counties in Kenya (Mburu et al., 2020).*

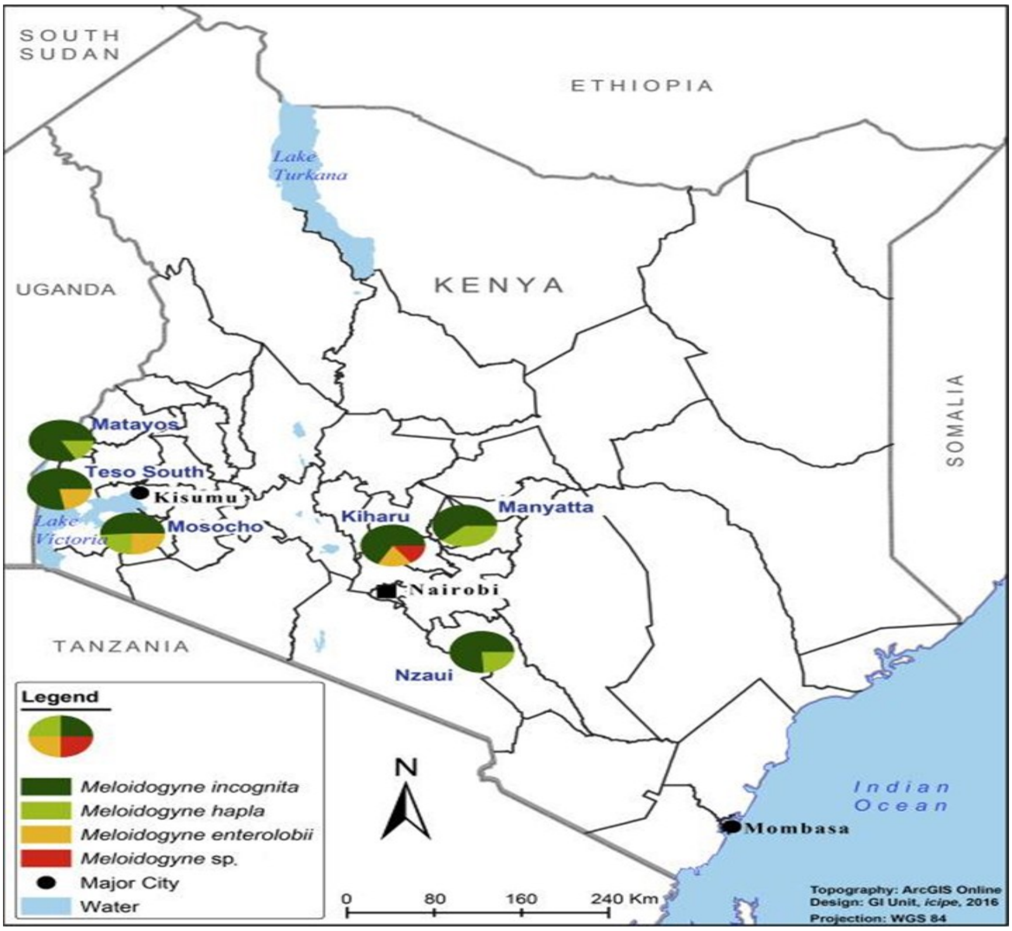


**Figure 2.** *Map of Kenya showing the distribution of Meloidogyne species in Kiharu, Manyatta, Matayos, Mosocho, Nzaui, and Teso South sub-counties, Kenya (Karuri et al., 2017).*

## NEMATODE HOST PLANTS AND DISSEMINATION

Nematodes have various growth stages that can be common or distinct depending on the species. The dormant stage of the eggs inside the cysts can last up to 20 years, even in the absence of a host or under adverse environmental conditions (Caixeta *et al*., 2016). This ability to survive adverse conditions for an extended period explains why controlling nematode parasites is always complex, challenging, and expensive. Nematodes can spread and colonize other fields in different ways. The most common dissemination methods include irrigation water, runoff, soil particles, seed tubers, footwear, animal hooves, farm implements, and machinery (Jatala and Bridge, 1990; Henrique, 2012). At the farms, several plant species play host to nematodes, and depending on the species, the hosts could be narrow to broad. Of all the categories of nematodes, root-knot nematodes have the widest host range (Mitkowski and Abawi, 2003). Common host plants to potato cyst, root-knot, and root lesion nematodes include tomato (*Solanum lycopersicum*), *Physalis spp*., African eggplant (*Solanum aethiopicum*), *Physoclaina* spp., eggplant (*Solanum melongena*), *Datura spp*., *Hyoscyamus spp*., *Salpiglossis* spp., and *Saracha* spp. (Senasica, 2013). In other countries, species such as *D. ferox*, *Nicotiana acuminata*, *S. ligustrinum* and *S. pinnatum* have also been reported as hosts of *G. rostochiensis* patotype Ro1 (Sullivan *et al*., 2007).

## SYMPTOMS OF NEMATODE INFECTION

Distinctive signs and symptoms of nematode infestations occur in patches of poor potato growth in the field, which tend to be circular to oval and vary from a few meters across to several acres in size. The infected plants show smaller, curled, and abnormally colored leaves, with brown spots on the margins and reduced numbers and sizes of leaflets, affecting photosynthesis (Pinheiro *et al*., 2015). The infected plants show yellowish symptoms, like water and nutrient deficiencies, reduced size, and number of tubers, with small lesions, making them unmarketable (Grabau and Noling, 2019; Price *et al*., 2021). Under the ground, RKN induces galling on tubers due to hypertrophy and hyperplasia of infected cells, making tubers look warty or bumpy (Lima *et al*., 2018). This galling provides positive diagnostic confirmation of RKN presence, infection severity, and potential for crop damage (Grabau and Noling, 2019). Plants with damaged roots wilt during warmer periods of the day and may remain wilted even with irrigation (Grabau and Noling, 2019). The root systems become less developed, and plants produce more lateral roots, leading to stunting, nonresponsive to fertilizations, and premature death (Lima *et al*., 2018). Specific to RKN infection, the attached plants show yellowing, stunting growth, wilting, brown spots, and tuber rotting. The numbers and sizes of galls vary depending on the susceptibility of the cultivar, favorable temperatures, and population density (Hunt and Handoo, 2009).

## INTEGRATED NEMATODE PEST MANAGEMENT (IPM) STRATEGIES

Once the nematodes are reported in the fields, management measures should be deployed immediately to avoid further dissemination and population build-up, thus avoiding yield losses. The success in reducing the population of nematodes varies and depends on the initial population density, soil type, plant genotype, and temperature (Ferris *et al*., 1994). The control methods for nematodes include quarantine regulations, planting tolerant varieties, crop rotation, crop succession, solarization, use of resistant varieties, and application of nematicides. This section discusses various IPM practices in detail- the practice, key components, and scientifically proven potential impact.

### Use of disease-free planting materials from tolerant varieties and weed management

Cultural practices help reduce nematode build-up when adequately carried out. Sourcing healthy planting materials from tolerant varieties is the first step in managing nematodes. Healthy tubers and seeds ensure nematode-free plants are used for production, thereby eliminating the potential of introducing pathogens to the fields. Using resistant or tolerant cultivars provides the most effective, low-cost, and environmentally sound strategy to control nematodes. These varieties must have been tested for resistance to the pathogen and proved to produce a good performance in a particular region where potatoes and other targeted crops are produced (Minnis *et al*., 2004). However, in potatoes, currently, there is no commercial cultivar available that is resistant to *Meloidogyne* spp. (Bali *et al*., 2021) even though some studies report the identification of resistant genes in wild genotypes and posterior introgression into breeding lines (Lima *et al*., 2018; Mburu *et al*., 2020). In tomatoes, some varieties have been reported to resist various nematode species, and the rootstocks are used to upgrade the resistance of the susceptible ones. For instance, grafted tomato plants using nematode-resistant rootstocks successfully control *Meloidogyne spp*. (Kaşkavalci *et al*., 2009). These researchers reported that rootstocks from Brigeor, Morgan, King-Kong, Unifort, and Emperador varieties exhibit high resistance to *M. arenaria* and *M. javanica*, but Multifort and Maxifort showed intermediate resistance. The selection of cultivars for nematode management should also consider the variety's growth cycle. For instance, late maturing potatoes planted during long rains are more likely to record yield losses caused by nematodes than short-cycle cultivars. Therefore, farmers should opt for fast-maturing varieties to minimize losses. In Kenya, some short-cycle potato varieties are Kerr's Pink, Sagitta, Shangi, and Derby, while early-medium maturing varieties include Kenya Mpya, Saviola, and Evora. The immediate destruction of volunteer potato plants, tubers, and weeds ensures that the pests do not survive and build up in the fields. Planting certified potato tubers and selecting planting dates to avoid a high RKN population during tuber growth have been suggested to help manage nematodes (Jones *et al*., 2017).

### Practice fallowing

Fallowing is a farming practice that leaves the soil uncultivated for a specific period. Two types of fallow could be practiced, namely weed-free and weedy fallow. As the names suggest, weed-free fallow is where no crop is planted, but the weeds are still controlled. This type of fellow yields better nematode control than weedy fallow but is always prone to soil erosion. In weedy fallow, no crop is grown, and weeds are allowed to grow uncontrolled- the fields are covered under weeds for the entire fallow period. This type of fallow may allow nematode numbers to increase as many plant-parasitic nematodes survive on weed hosts (Schroeder *et al*., 1993). The effectiveness of fallow systems is contingent upon the duration of the fallow period—a longer fallow period result in improved nematode control. To successfully grow an annual crop susceptible to root-knot nematodes, fallowing for at least a year is necessary as it lowers the nematode population (Perry, 2010). Weaver *et al*. (1995) found that fallows successfully improved yields despite a mixed population of the cyst and root-knot nematodes. The efficacy of the fallow practice could be increased through combination with other practices. For instance, Adediran *et al*. (2005) observed that, on average, siam weed fallow reduced nematode population densities by 67-79%, mucuna fallow by 64-72%, and the natural bush by 30-49% across the trial locations. During fallowing, it is crucial to maintain soil moisture to encourage egg hatching and eliminate weeds that may harbor nematodes (Perry, 2010). The eggs will hatch due to the favorable moisture levels but then perish in the absence of a food source (Widmer *et al*., 2002).

### Implement crop rotation

Crop rotation is the practice of planting crops from different families sequentially on the same plot of land to combat pests and diseases, among other benefits. The practice works to break the lifecyle of nematodes by depriving them of their preferred host crops. Continuous cultivation of potatoes on PCN-infested land can be devastating and lead to crop loss of up to 80% (NPCK, 2021). Crops to consider for rotation with potatoes and other Solanaceae include barley (*Hordeum vulgare*), oat cultivars (*Avena sativa*), cotton (*Gossypium hirsutum*), and some grasses, sorghum, maize, castor beans, brassica crops, including cabbage, cauliflower, mustard, and Chinese cabbage (Pinheiro *et al*., 2009; Otieno, 2019). Specifically, sorghum, maize, and castor beans have been deployed for *M. javanica* control. However, potatoes, tomatoes, eggplants, or other solanaceous species should be avoided. There is evidence of success with the rotation strategy, and the impact is greatly influenced by the rotation length, crop species involved, and soil type (Forge *et al*., 2015). Johnson *et al*. (1996) reported three years of potato yield increase when rotated with sorghum and beans in plots infested with *M. hapla* or *M. arenaria* and *M. incognita*. Lima *et al*. (2018) recommended about four years, accompanied by proper weed control, to achieve success in nematode control. Rotating potatoes with barley showed a reduction in *G. rostochiensis* up to 87% (Senasica, 2013). When using marigold for rotation, Ploeg (2002) observed a significant increase in tomato yields, which were 50% higher than after fallow. However, rotation is not very effective in the short term and requires longer periods for a meaningful reduction of nematodes. According to the modeling procedure by Trudgill *et al.* (2014), rotations longer than eight years would be required to control cyst nematode unless other effective control measures, such as growing partially resistant cultivars, were used. Due to this requirement, crop rotation may not always be economically the best way to manage these nematodes in certain cropping systems and regions. However, when looking at other soil health benefits, it is still worth practicing and could be combined with other strategies. For instance, Smiley *et al.* (2012) concluded that the management of nematodes could be achieved by combining crop rotations and genetic resistance. Chen and Tsay (2006) found that rotation with rice or taro and the cultural practice of flooding and bare fallowing for four months reduced nematode soil populations to two or fewer nematodes per 100 ml of soil.

### Plant trap and antagonistic plants

The trap strategy involves utilizing a different crop as a host to entice, disrupt, or confine specific pests to decrease the harm caused to the primary crop (Samara, 2022). By cultivating trap crops simultaneously with the main crop, it may be possible to limit the impact of nematodes by attracting nematodes towards the trap crop instead of the primary crop. This approach could prove more effective than chemical-based control methods, as trap crops are generally more attractive to nematodes than the main crop. The length of time these plants take in the field is critical and should be timely destroyed after planting to stop the nematode cycle (Halford *et al*., 1999). If these plants are not destroyed or destroyed too late, the nematode population will build up. To avoid this potential risk, trap crops should be removed from the field after 6–8 weeks (Samara, 2022). This also helps reduce the competition between the main and trap crops. Some examples of trap plants include the *Solanum Sisymbriifolium* (Lam.), which has shown a significant reduction of potato cyst nematode population density in the field by up to 80% (Timmermans *et al*., 2007). Others include *S. tuberosum*, *S. nigrum*, *S. dulcamara*, and *D. stramonium*, which have also shown promising results (Sparkes, 2013). On the other hand, antagonistic plants will initially withstand nematode infection; however, later in their cycle, plant factors will stop their further development. These plants include *Crotalaria spectabilis*, *C. juncea*, *Tagetes patula*, *T. minuta*, *T. erecta*, and *Estizolobium* spp, and have been used to manage root-knot nematodes in potato fields (Gomes and Souza, 2003; Embrapa, 2015; Lima *et al*., 2018).

### Practice solarization

Soil solarization is a method of soil disinfestation based on solar heating by mulching soil with transparent polyethylene during the hot season, thereby controlling soil-borne pests (Katan and Gamliel, 2014). This method is environmentally friendly and suitable for organic and integrated crop management systems. It effectively manages soil-borne pathogens and has long-lasting benefits for the agroecosystem, including improved soil nutrient levels and increased microbial activity. The values of the maximum soil temperature and amount of heat accumulated (duration x temperature) determine the potential of the thermal killing effect on soil-borne pests and weed seeds (Rubi *et al*., 2007). Under this method, the soils are covered with a polythene sheet for about 4-6 weeks. The method works best in warmer regions and when the soils have high organic matter content (Rubi *et al*., 2007). Gamliel and Katan (2012) have covered other basic principles of solarization. Comprehending how the management measure works, and the biological and environmental processes it affects, is a valuable resource for improving pest management, anticipating its effectiveness, and decreasing unwanted consequences. According to Katan and Gamliel (2014), the mechanisms of biological control that may be created, stimulated, or suppressed by solarization can be connected with the inoculum existing in the infested soil; suppression of reinfestation of the soils after the termination of the solarization process, leading to induced soil suppressiveness; and induced host–plant resistance.

The application of solarization in nematode management has been studied and reported. Solarization for about 30 days reduces root-knot nematodes and potato cyst nematodes from 80-100% under no solarization conditions to 1-20% (Carson and Otoo, 1996; Greco *et al*., 2000). Such control has resulted in a 28-32.3% increase in potato yields under field conditions (Greco *et al*., 2000). To apply this, farmers need to prepare their farms early by carrying out deep cultivation and allowing the field to be heated for at least 30 days before the onset of rains. Though promising and effective, resource-constrained small-scale farmers may need financial help to adopt since using polythene to cover the fields may become expensive. Again, the effects of solarization on nematodes differ across species and may not always be as potent as on bacteria and weeds. This is because nematodes are mobile and have the ability to burrow deeper into the soil to avoid heat during solarization, which allows them to recolonize soil quickly and plant roots after treatment (Elmore *et al*., 1997). Moreover, solarization is most effective in controlling nematodes within the top 12 inches of soil. Hence, nematodes that reside deeper in the soil can survive the treatment and continue to cause destruction to plants, especially those with deep root systems (Elmore *et al*., 1997).

### Adopt biological control

Biological control involves the use of living organisms to manage the nematode population. Commonly used microorganisms include Trichoderma, mycorrhizal and endophytic fungi. These organisms have been used to produce nematicides for application, which can be used solely or in combination with synthetic nematicides. The *Streptomyces sp*. strain NA-369 is able to control *M. incognita* in eggplant (Chubachi *et al*., 2003). Studies have also demonstrated the potential of combining chemical and biological nematicides to control nematodes. According to Dahlin *et al*. (2019), Velum reduced the population of nematodes at planting and reinforced the biological efficacy of

*Purpureocillium lilacinum* strain 251 (BioAct) throughout the growing season. Velum should be applied pre-plant and up to 6 weeks post-planting, whereas the nematode antagonist *P. lilacinum* should be applied before planting and during the entire crop season (Dahlin *et al*., 2019). The use of *Trichoderma harzianum* has also been tested and showed positive results. For example, Sahebani and Hadavi (2008) found that when tomato seeds were treated with *T. harzianum* under greenhouse conditions, the level of disease caused by the nematode *Meloidogyne* spp. was significantly reduced. Also, Martínez-Medina *et al.* (2017) observed that the presence of *T. harzianum* in the roots of plants hindered nematode invasion, galling, and reproduction. The use of trichoderma, mycorrhizal and endophytic fungi as biological control strategies for plant-parasitic nematodes has been reviewed in detail by Poveda *et al*. (2020).

**Table 1.** Efficacy of various nematicides on potato cyst nematode and root-knot nematodes. The efficacy levels considered included egg inhibition, nematode population reduction, reduction in galling index, and yield improvement.

| **Active ingredient** | **Product example** | **Nematode type** | **Efficacy recorded- %** | **Application method** | **Reference** |
|---|---|---|---|---|---|
| Fluopyram | Pestanal, Velum Prime | Root-knot and cyst nematode | 78-100 | Soil- furrow/pot and foliar | Arita *et al.* (2020); Silva *et al.* (2019); Faske and Hurd (2015); Dahlin *et al.* (2019) |
| Fluensulfone | Nimitz 15G®, Nimitz 480EC® | Root-knot and cyst nematode | 73-100 | Soil- furrow/pot and foliar | Arita *et al.* (2020); Silva *et al.* (2019); Morris *et al.* (2015, 2016); Grabau *et al.* (2019); Norshie *et al.* (2016) |
| Oxamyl | Nematex, Vydate 10G | Root-knot and cyst nematode | 33-80 | Soil- furrow / pot and foliar | Hajihassani *et al.* (2019); Morris *et al.* (2016); Mostafa *et al.* (2015); Norshie *et al.* (2016); Minnis *et al.* (2004); Ingham *et al.* (2000); Radwan *et al.* (2012) |
| Fluazaindolizine | Salibro SC 500, Reklemel™ | Root-knot and cyst nematode | 67-81 | Soil- furro/ pot and foliar | Hajihassani *et al.* (2019); Lahm *et al.* (2017); Wram and Zasada (2020) |
| 1,3-dichloroprene | Telone II: 94% | Root-knot and cyst nematode | 52-100 | Injection | Minnis *et al.* (2004); Grabau *et al.* (2019); Hafez and Sundararaj (2006); Ingham *et al.* (2000) |
| Ethoprophos | Savanem 20 EC, Mocap, Super Control | Root-knot and cyst nematode | 53-97 | Soil | Adamou *et al.* (2013); Mostafa *et al.* (2015); Hafez and Sundararaj (2006); Radwan *et al.* (2012); Ingham *et al.* (2000) |
| Aldicarb | Temik 10G | Root-knot and cyst nematode | 83 | Soil/ granular | Minnis *et al.* (2004); Greco *et al.* (2000); Chabrier *et al.* (2002) |
| Fosthiazate | Nemathoin 10% | Root-knot and cyst nematode | 63-97 | Soil/granular | Minnis *et al.* (2004); Norshie *et al.* (2016); Radwan *et al.* (2012); Chabrier *et al.* (2002); Ingham *et al.* (2000) |
| Carbofuran | Furadan | Root-knot and cyst nematode | 94-96 | Soil | Radwan *et al.* (2012) |
| Cadusafos | Rugby® 10G | Root-knot and cyst nematode | 77-86 | soil | Radwan *et al.* (2012) |
| Purpureocillium lilacinum strain 251 | BioAct WG | Root-knot nematode | 56-61 | Soil | Dahlin *et al.* (2019) |
| Bacillus sp BC27 &29 (from goat manure) | Lab. Isolate | Rootknot nematode | 80-100 | Soil | Chinheya *et al.* (2017) |
| Spirotetramat | Movento | Rootknot nematode | 35-78 | Foliar spray | Baidoo *et al.* (2017); Jones *et al.* (2017); Smiley *et al.* (2011); Vang *et al.* (2016); McKenry *et al.* (2009) |

### Adopt chemical control

Eliminating nematodes on some crops is essential for specific high-end market requirements, especially for high-value horticultural products like potatoes. Chemical control with nematicides has been used globally with a satisfactory control rate. However, using nematicides is costly and has severe human and ecological damage (Greco, 1993; Gowen, 1997). In order to improve control, chemicals should be used just before and after the planting of potatoes (Jones *et al*., 2017). Several compounds have been used with much success to control nematodes—some of these products including Fluopyram (Pestanal and Velum Prime), Fluensulfone (Nimitz 15G® and Nimitz 480EC®), Fluazaindolizine (Salibro SC 500 and Reklemel™), and Tioxazafen (for seed treatment) (Table 1).

The efficacy of various nematicides on cyst nematodes and root-knot nematodes can be assessed based on egg inhibition, nematode population reduction, reduction in the galling index, and yield improvement. Under foliar application, it is vital to consider the phytotoxicity characteristics of the products. For example, fluensulfone can cause phytotoxic effects on eggplant and tomato crops. The depth of incorporation also influences the efficacy of these nematicides. Incorporating the products to 15-20 cm with tubers planted at a depth of 10-15 cm reduced root invasions and consistently gave higher ware yields and better nematode control than the other incorporation methods (Woods and Haydock, 2000). Application time also matters a lot in influencing efficacy. Vang *et al.* (2016), Baidoo *et al.* (2017), and Jones *et al.* (2017) recommended that early application within 0-2 weeks after planting is much better and should be encouraged than later application when nematode infestation has already occurred. Some products have shown inconsistent results adding to the complexities in the nematode control. For instance, fluensulfone was less efficacious than oxamyl or fosthiazate, which suggests that the treatment may not reliably be integrated within shorter potato rotations (Norshie *et al*., 2016). Observations on nematode multiplication control indicated oxamyl generally reduced multiplication to a greater extent than the other products tested in sites of organic, silty- and sandy-loam and mineral soils (Michaelides *et al*., 2000). Regarding the speed of action, aldicarb acted faster and gave better control than ethoprophos (Michaelides *et al*., 2000).

Potato yield increase due ot nematicide application is never consistent and varies widely (negative to positive figures) depending on several factors, including soil type, soil moisture content, variety tolerance level, soil temperature, and weed control (Bui and Desaeger, 2021). The commonly reported yield increase range is between 0 and 44% among various crops (Greco, 2000; Chabrier *et al*., 2002; Norshie *et al*., 2016). Other compounds, such as abamectin, have also been tested with promising results on potato cyst nematode control (Sasanelli *et al*., 2020). According to Qiao *et al.* (2012), abamectin exhibited excellent control effects on nematodes while giving higher yields, 19-39% above control. However, this product is highly hazardous and should be avoided by small-scale farmers. Other products like spirotetramat show promising results though inconsistent (Desaeger *et al*., 2020).

### Conclusion

Potatoes are often impacted negatively by several nematode species, which can cause significant yield losses and poor qualities of tubers. The severity of this issue depends on various factors, including the nematode species, local climate, plant host factors, and management practices. Proper identification of nematode species is crucial for effective control. However, nematodes are difficult to manage or eradicate once introduced into a field due to their unique biology and behaviors. Effective control of nematodes requires farmers to practice integrated nematode management systems with a combination of at least two management practices. Several cultural and agronomic practices have shown some decent levels of efficacy and have been recommended for adoption. Timely application of these practices is critical for any better outcome to be realized. The application of nematicides is the most effective strategy. However, chemicals should be cautiously applied as they have severe human and ecological damage. The implementation of these practices must also consider their economic and technical feasibility for sustainable cropping of potato cultivars and other Solanaceae.



### REFERENCES

Abrantes, I., Almeida, M. T., Conceição, I. L., Esteves, I., & Maleita, C. (2023). Nematodes of potato and their management. In *Potato Production Worldwide* (pp. 213-240). Academic Press.

Adamou, H., Mamadou, A., Adamou, B., Ali, D., & Toudou, A. (2013). On-farm testing of savanem 20 EC (Ethoprophos) for control of plant parasitic nematodes associated with pepper (*Capsicum annuum*) in Tillaberi (Niger). *Asian Journal of Agricultural Sciences*, *5*(4), 83-87.

Adediran, J. A., Adegbite, A. A., Akinlosotu, T. A., Agbaje, G. O., Taiwo, L. B., Owolade, O. F., & Oluwatosin, G. A. (2005). Evaluation of fallow and cover crops for nematode suppression in three agroecologies of south western Nigeria. *African Journal of Biotechnology*, *4*(10).

Arita, L. Y., da Silva, S. A., & Machado, A. C. Z. (2020) Efficacy of chemical and biological nematicides in the management of Meloidogyne paranaensis in *Coffea arabica*. *Crop Protection*, *131*, 105099.

Baidoo, R., Mengistu, T., McSorley, R., Stamps, R.H., Brito, J., & Crow, W.T. (2017). Management of root-knot nematode (*Meloidogyne incognita*) on *Pittosporum tobira* under greenhouse, field, and on-farm conditions in Florida. *Journal of nematology*, 49(2),133.

Bali, S., Zhang, L., Franco, J., & Gleason, C. (2021). Biotechnological advances with applicability in potatoes for resistance against root-knot nematodes. *Current Opinion in Biotechnology*, *70*, 226-233.

Brown, E. B., & Sykes, G. B. (1983). Assessment of the losses caused to potatoes by the potato cyst nematodes, *Globodera rostochiensis* and *G. pallida*. *Annals of Applied Biology*, 103(2), 271-276.

Bui, H. X., & Desaeger, J. A. (2021). Efficacy of nonfumigant nematicides against *Meloidogyne javanica* as affected by soil temperature under pasteurized and natural soil conditions. *Pest Management Science*, *77*(7), 3179-3186.

Caixeta, L. B., Furlaneto, C., Cares, J. E. & Gênero Heterodera (2016) In: Oliveira CMG, Santos MA, Castro LHS, editors. Diagnose de Fitonematoides. 1st ed. Campinas: Millennium; 2016.pp. 149-162

Carson, G., & Otoo, E. (1996). Application of soil solarization to control root-knot nematodes and weeds in transplanted tomato. *Ghana Journal of Agricultural Science*, *29*(2), 91-98.

Chabrier, C., Hubervic, J., & Quénéhervé, P. (2002). Evaluation of fosthiazate (Nemathorin® 10G) for the control of nematodes in banana fields in Martinique. *Nematropica*, 137-148.

Chen, P., & Tsay, T. (2006). Effect of crop rotation on *Meloidogyne* spp. and *Pratylenchus* spp. populations in strawberry fields in Taiwan. *Journal of Nematology*, *38*(3), 339.

Chinheya, C. C., Yobo, K. S., & Laing, M. D. (2017). Biological control of the rootknot nematode, *Meloidogyne javanica* (Chitwood) using *Bacillus* isolates, on soybean. Biological control, 109:37-41.

Chubachi, K., Fukuda, S., Matsumura, S., Yanagisawa, T., Itagawa, H., Shimizu, T., & Nakagawa, A. (2003). Suppression of root-knot nematode disease in eggplants by antinematodal Streptomyces NA-369 in artificially infested soil. *Biocontrol Science*, *8*(2), 93-96.

Dahlin, P., Eder, R., Consoli, E., Krauss, J., & Kiewnick, S. (2019). Integrated control of Meloidogyne incognita in tomatoes using fluopyram and *Purpureocillium lilacinum* strain 251. *Crop Protection*, *124*, 104874.

Desaeger, J., Wram, C., & Zasada, I. (2020). New reduced-risk agricultural nematicides-rationale and review. *Journal of Nematology*,52.

Elling, A. A. (2013). Major emerging problems with minor Meloidogyne species. *Phytopathology*, *103*(11), 1092-1102.

Elmore, C. L., Stapleton, J. J., Bell, C. E., & DeVay, J. E. (1997). Soil Solarization: A Non-pesticidal Method for Controlling, Diseases, Nematodes, and Weeds. *Oakland: University of California, Agriculture and Natural Resources Publication*, *21377*, 60-80.

Embrapa. (2015). Nematoides na cultura da batata. Circular Técnica.,143:1-12.

Faske, T. R., & Hurd, K. (2015). Sensitivity of *Meloidogyne incognita* and *Rotyenchulus reniformis* to fluopyram. *Journal of Nematology*, 47(4), 316–321.

Ferris, H., Westerdahl, B. B., & Carlson, H. L. (1994). Nematode population changes under crop rotation sequences: consequences for potato production. *Agronomy Journal*, *86*(2), 340-348.

Forge, T. A., Larney, F. J., Kawchuk, L. M., Pearson, D. C., Koch, C., & Blackshaw, R. E. (2015). Crop rotation effects on *Pratylenchus neglectus* populations in the root zone of irrigated potatoes in southern Alberta. *Canadian Journal of Plant Pathology*, *37*(3), 363-368.

Gamliel, A., & Katan, J. (Eds.). (2012). *Soil solarization: Theory and practice* (p. 266). St. Paul: American Phytopathological Society.

Gomes, C.B., & Souza, R.M. (2003). Doenças causadas por nematoides. In: Pereira AS, Daniels J, editors. O cultivo da batata na região sul do Brasil. 1st ed. Brasilia, Brazil: Embrapa,. pp. 321-349

Gowen, S. R. (1997). Chemical control of nematodes: efficiency and side-effects. FAO Plant Production and Protection Paper (FAO).

Grabau, Z. J., & Noling, J. W. (2019). Nematode management in potatoes (Irish or White). University of Florida publication Series no. ENY-029, USA, 12.

Grabau, Z. J., Noling, J. W., & Gine, P. A. N. (2019). Fluensulfone and 1, 3-dichloroprene for plant-parasitic nematode management in potato production. *Journal of Nematology*, *51*.

Greco, N. (1993). Reviews: nematode problems affecting potato production in subtropical climates. *Nematropica*, 213-220.

Greco, N., Brandonisio, A., & Dangelico, A.. (2000). Control of the potato cyst nematode, *Globodera rostochiensis*, with soil solarization and nematicides. *Nematologia Mediterranea*, 28:93-9.

Hafez, S. L., & Sundararaj, P. (2006). Evaluation of fumigant and non-fumigant nematicides for the control of *Meloidogyne chitwoodi* on potato. *Nematologia Mediterranea*.

Hajihassani, A., Davis, R. F., & Timper, P. (2019). Evaluation of selected nonfumigant nematicides on increasing inoculation densities of *Meloidogyne incognita* on cucumber. *Plant Disease*, *103*(12), 3161-3165.

Halford, P. D., Russell, M. D., & Evans, K. (1999). Use of resistant and susceptible potato cultivars in the trap cropping of potato cyst nematodes, *Globodera pallida* and *G. rostochiensis*. *Annals of Applied Biology*, *134*(3), 321-327.

Hallmann, J., & Sikora, R. A. (1994). Occurrence of plant parasitic nematodes and non‐pathogenic species of Fusarium in tomato plants in Kenya and their role as mutualistic synergists for biological control of root-knot nematodes. *International Journal of Pest Management*, *40*(4), 321-325.

Haukeland, S. (2016). Training of Twenty (20) Government technicians and determination of Potato Cyst Nematode status in 1200 soil samples Final Narrative Report Submitted to FAO. In FAO Report (Vol. 254, Issue March).

Henrique. (2012). Nematodos de quisto de batateira *Globodera rostochiensis* and *G. pallida*. Boletim Técnico – INIA. 2012;12:1-2

Hunt, D., & Handoo, Z. (2009). Taxonomy, identification and principal species. In: Perry RN, Moens M, Star JL, editors. Root Knot Nematodes. 1st ed. London: CAB International, pp. 55-88.

Ingham, R.E., Hamm, P.B., Williams, R.E., & Swanson, W.H. (2000). Control of *Meloidogyne chitwoodi* in potato with fumigant and nonfumigant nematicides. *Journal of Nematology*, *32*(4S), 556.

Jatala, P., & Bridge, J. (1990) Nematode parasites of root and tuber crops. In: Luc M, Sikora RA, Bridge J, Editors. Plant Parasitic Nematodes in Tropical and Subtropical Agriculture. 1st ed. Wallingford: CAB International; 1990. pp. 137-180.

Johnson, A.W., Dowler, C.C., Glaze, N.C., & Handoo, Z.A. (1996). Role of nematodes, nematicides, and crop rotation on the productivity and quality of potato, sweet potato, peanut, and grain sorghum. *Journal of Nematology*, *28* (3), 389.

Jones, J. G., Kleczewski, N. M., Desaeger, J., Meyer, S. L., & Johnson, G. C. (2017). Evaluation of nematicides for southern root-knot nematode management in lima bean. *Crop Protection*, *96*, 151-7.

Jones, R. K., Storey, S.G., Knoetze, R., & Fourie, H. (2017). Nematode Pests of Potato and Other Vegetables– Nematology in South Africa. A View from the 21st Century. Vol. 1. Cham, Switzerland: Springer International Publishing, 231-260.

Kanyagia, S. T. (1979). A survey of vegetable nematodes in Kenya. *East African Agricultural and Forestry Journal*, *44*(3), 178-182.

Karuri, H. W., Olago, D., Neilson, R., Mararo, E., & Villinger, J. (2017). A survey of root knot nematodes and resistance to Meloidogyne incognita in sweet potato varieties from Kenyan fields. *Crop Protection*, *92*, 114-121.

Ka⍰kavalcı, G., Tüzel, Y., Dura, O., & Öztekin, G.B. (2009). Effects of alternative control methods against Meloidogyne incognita in organic tomato production. *Ekoloji*, *18*(72), 23-31.

Katan, J., & Gamliel, A. (2014). Plant health management: soil solarization. *Encyclopedia of Agriculture and Food Systems*, *4*, 460–471.

Lahm, G. P., Desaeger, J., Smith, B. K., Pahutski, T. F., Rivera, M. A., Meloro, T., & Wiles, J. A. (2017). The discovery of fluazaindolizine: a new product for the control of plant parasitic nematodes. *Bioorganic & Medicinal Chemistry Letters*, *27*(7), 1572-1575.

Lima, F. S., Mattos, V. S., Silva, E. S., Carvalho, M. A., Teixeira, R. A., Silva, J. C., & Correa, V. R. (2018). Nematodes affecting potato and sustainable practices for their management. *Potato: From Incas to All Over the World*, *107*.

Martínez-Medina, A., Fernandez, I., Lok, G. B., Pozo, M. J., Pieterse, C. M., & Van Wees, S. C. (2017). Shifting from priming of salicylic acid-to jasmonic acid⍰ regulated defences by Trichoderma protects tomato against the root knot nematode Meloidogyne incognita. *New phytologist*, 213(3), 1363-1377.

Mbiyu, M. W., Kimenju, J. W., Muiru, W. M., Nyongesa, M. W., & Haukeland, S. (2022). Relative yield loss of potato in fields infested with potato cyst nematodes and the efficacy of selected control products in Nyandarua County, Kenya. *East African Agricultural and Forestry Journal*, *86*(3 & 4), 10-10.

Mbogoh, J. M., Omami, E., Ngode, L., & Ochuodho, J. (2013). Incidence and occurrence of root-knot nematode (Meloidogyne species) in African leafy vegetables in Western Kenya. *African Journal of Education, Science and Technology*, *1* (1), 139-152.

Mburu, H., Cortada, L., Haukeland, S., Ronno, W., Nyongesa, M., Kinyua, Z., Bargul, J. L., & Coyne, D. (2020) Potato cyst nematodes: a new threat to potato production in East Africa. *Frontiers in Plant Science*, *25*(11), 670.

Mburu, H., Cortada, L., Mwangi, G., Gitau, K., Kiriga, A., Kinyua, Z., & Haukeland, S. (2018). First report of potato cyst nematode *Globodera pallida* (Stone, 1973) infecting potato (*Solanum tuberosum* L.) in Kenya. *Plant Disease*. https://doi.org/10.1094/PDIS-11-17-1777-PDN

Medina, I. L., Gomes, C. B., Correa, V. R., Mattos, V. S., Castagnone-Sereno, P., & Carneiro, R. M. (2017). Genetic diversity of Meloidogyne spp. parasitising potato in Brazil and aggressiveness of *M. javanica* populations on susceptible cultivars. *Nematology*, *19*(1), 69-80.

Michaelides, P., Foesser, F., & Beadle, N. (2000). Nematicide efficacy tests for potato cyst nematode control in the UK and Europe. *Aspects of Applied Biology*, (59), 115-120.

Minnis, S. T., Haydock, P. P. J., & Evans, K. (2004). Control of potato cyst nematodes and economic benefits of application of 1, 3-dichloropropene and granular nematicides. *Annals of Applied Biology*, *145*(2), 145-156.

Mitkowski, N. A., & Abawi, G. S. (2003). Root-knot nematodes. The plant health instructor, 10.

Moens, M., Perry, R. N., & Starr, J. L. (2009). Meloidogyne species-a diverse group of novel and important plant parasites. In Root-knot nematodes (pp. 1-17). Wallingford UK: CABI.

Morris, K. A., Langston, D. B., Davis, R. F., Noe, J. P., Dickson, D. W., & Timper, P. (2016). Efficacy of various application methods of fluensulfone for managing root-knot nematodes in vegetables. *Journal of Nematology*, *48*(2), 65-71.

Morris, K. A., Langston, D. B., Dickson, D. W., Davis, R. F., Timper, P., & Noe, J. P. (2015). Efficacy of fluensulfone in a tomato cucumber dou ble cropping system. *Journal of Nematology*, *47*(4), 310–315.

Mostafa, F. A., Ali, R. A., & Zawam, H. S. (2015). Effect of certain commercial compounds in controlling root-knot nematodes infected potato plants. *Journal of Phytopathology and Pest Management*, *27*, 9-19.

Mwangi, J. M., Kariuki, G. M., Waceke, J. W., & Grundler, F. M. (2015). First report of Globodera rostochiensis infesting potatoes in Kenya. *New Disease Reports*, *31*, 18-18.

Norshie, P. M., Grove, I. G., & Back, M. A. (2016). Field evaluation of the nematicide fluensulfone for control of the potato cyst nematode Globodera pallida. *Pest Management Science*, *72*(10), 2001-2007.

NPCK (2021) Potato Cyst Nematode Workshop. Available online at: https://npck.org/potato-cyst-nematodeworkshop/#:~:text=PCN%20is%20primarily%20a%20potato,Globodera%20rosotchiensis%20has%20been%20detected (Accessed on 14th April, 2023).

Onkendi, E. M., Kariuki, G. M., Marais, M., & Moleleki, L. N. (2014). The threat of root-knot nematodes (*M. eloidogyne* spp.) in A frica: a review. *Plant pathology*, *63*(4), 727-737.

Otieno, H. M. (2019). Impacts and Management Strategies of Common Potato (Solanum tuberosum L.) Pests and Diseases in East Africa. *Frontiers in Science*, *9*(2), 33-40.

Otieno, H. M. O., & Mageto, E. K. (2021). A review on yield response to nitrogen, potassium and manure applications in potato (*Solanum tuberosum* L.) production. *Archives of Agriculture and Environmental Science*, *6*(1), 80-86.

Perry, E. J. (2010). How to manage pests, pests in gardens and landscapes. University of California, Davis, CA 95616.

Pinheiro, J.B., Lopes, C.A., & Henz, G.P. (2009). Medidas gerais de controle de nematoides da batata. Circular Técnica, 76:1-9

Pinheiro, J.B., Silva, F.O., & Pereira, R.B. (2015). Nematoides na cultura da batata. *Circular Técnica*, 143,1-12.

Ploeg, A. T. (2002). Effects of selected marigold varieties on root-knot nematodes and tomato and melon yields. *Plant Disease*, *86*(5), 505-508.

Poveda, J., Abril-Urias, P., & Escobar, C. (2020). Biological control of plant-parasitic nematodes by filamentous fungi inducers of resistance: Trichoderma, mycorrhizal and endophytic fungi. *Frontiers in Microbiology*, *11*, 992.

Price, J. A., Coyne, D., Blok, V. C., & Jones, J. T. (2021). Potato cyst nematodes Globodera rostochiensis and G. pallida. *Molecular Plant Pathology*, *22*(5), 495-507.

Qiao, K., Liu, X., Wang, H., Xia, X., Ji, X., & Wang, K. (2012). Effect of abamectin on root-knot nematodes and tomato yield. *Pest Management Science*, *68*(6), 853-857.

Radwan, M. A., Farrag, S. A. A., Abu-Elamayem, M. M., & Ahmed, N. S. (2012). Efficacy of some granular nematicides against root-knot nematode, *Meloidogyne incognita* associated with tomato. *Pakistan Journal of Nematology*, *30*(1), 41-47.

Rubi, B., Cohen, O., & Gamliel, A. (2007). Soil solarization: an environmentally-friendly alternative. intechnical workshop on non-chemical alternatives to replace methyl bromide as a soil fumigant, Budapest, Hungary, pp. 26-28.

Sahebani, N., & Hadavi, N. (2008). Biological control of the root-knot nematode *Meloidogyne javanica* by *Trichoderma harzianum*. *Soil Biology and Biochemistry*, *40*, 2016–2020.

Samara, R. (2022). Evaluation of 11 potential trap crops for root-knot nematode (RKN) control under glasshouse conditions. *Open Agriculture*, *7*(1), 61-68.

Sasanelli, N., Toderas, I., Veronico, P., Iurcu-Straistaru, E., Rusu, S., Melillo, M.T., & Caboni, P. (2020). Abamectin efficacy on the potato cyst nematode *Globodera pallida*. *Plants*, *9*(1),12.

Schroeder, J., Thomas, S. H., & Murray, L. (1993). Yellow and purple nutsedge and chile peppers host southern root-knot nematode. *Weed Science*, *41*(1), 150-156.

Senasica (2013) Nematodo dorado de la papa- Globodera rostochiensis – Mexico–Segarpa. *Ficha Técnica*, *19*, 1–24

Silva, J. D., Loffredo, A., da Rocha, M. R., & Becker, J. O. (2019). Efficacy of new nematicides for managing Meloidogyne incognita in tomato crop. *Journal of Phytopathology*, *167*(5), 295-8.

Smiley, R. W., Gourlie, J. A., Rhinhart, K. E. L., Marshall, J. M., Anderson, M. D., & Yan, G. P. (2012). Influence of nematicides and fungicides on spring wheat in fields infested with soil-borne pathogens. *Plant Disease*, 96,1537-1547.

Smiley, R., Marshall, J., & Yan, G. (2011). Effect of foliarly applied spirotetramat on reproduction of *Heterodera avenae* on wheat roots. *Plant Disease*, *95*(8), 983-989.

Sparkes, J. (2013). Potential trap crops for the control of potato cyst nematodes (PCN). *Agriculture and Horticulture Development Board*, *1*, 1-19.

Sullivan, M. J., Inserra, R. N., Franco, J., Moreno-Leheude, I., & Greco, N. (2007). Potato cyst nematodes: plant host status and their regulatory impact. *Nematropica*, 193-202.

Timmermans, B. G. H., Vos, J., Stomph, T. J., Van Nieuwburg, J., & Van der Putten, P. E. L. (2007) Field performance of *Solanum sisymbriifolium*, a trap crop for potato cyst nematodes. *Annals of Applied Biology*, *150*, 99-106

Trudgill, D. L., Phillips, M. S., & Elliott, M. J. (2014). Dynamics and management of the white potato cyst nematode *Globodera pallida* in commercial potato crops. *Annals of Applied Biology*, *164*(1), 18-34.

Van den Berg, E., Marais, M., Kandji, S., Mounport, D., Ndiaye, B., & Cadet, P. (2001). Plant-parasitic nematodes associated with maize-bean intercropping systems in Kenya. *African Plant Protection*, *7*(2), 81-84.

Vang, L. E., Opperman, C. H., Schwarz, M. R., & Davis, E. L. (2016). Spirotetramat causes an arrest of nematode juvenile development. *Nematology*, *18*(2), 121-31.

Wanjohi, W. J., Wafula, G. O., & Macharia, C. M. (2018). Integrated management of *Fusarium* wilt-root knot nematode complex on tomato in central highlands of Kenya. *Sustainable Agriculture Research*, *7*(526-2020-469), 8-18.

Weaver, D. B., Rodriguez-Kabana, R., & Carden, E. L. (1995). Comparison of crop rotation and fallow for management of *Heterodera glycines* and *Meloidogyne* spp. in soybean. *Journal of Nematology*, *27*(4S), 585.

Whitehead, A. G., & Kariuki, L. (1960). Root-knot nematode surveys of cultivated areas in East Africa. *East African Agricultural and Forestry Journal*, *26*(2), 87-91.

Widmer, T. L., Mitkowski, N. A., & Abawi, G. S. (2002). Soil organic matter and management of plant-parasitic nematodes. *Journal of Nematology*, *34*(4), 289.

Woods, S.R., & Haydock, P.P. (2000). The effect of granular nematicide incorporation depth and potato planting depth on potatoes grown in land infested with the potato cyst nematodes *Globodera rostochiensis* and *G. pallida*. *Annals of Applied Biology*, 136(1):27-33.

Wram, C. L., & Zasada, I. (2020). Differential response of *Meloidogyne*, *Pratylenchus*, *Globodera*, and *Xiphinema* species to the nematicide fluazaindolizine. *Phytopathology®*, *110*(12), 2003-2009.