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Weeds as Hosts of Plant Parasitic Nematodes in Subtropical Agriculture Systems

Habraham F. Lopez[1], Pushpa Soti[2*], Ganpati B. Jagdale[3], Parwinder Grewal[4], and Alexis Racelis[5]

[1]School of Earth Environment and Marine Sciences, University of Texas Rio Grande Valley, Edinburg, TX, USA; [2]Biology Department, University of Texas Rio Grande Valley, Edinburg, TX, USA; [3]Departmetn of Plant Pathology, University of Georgia, Athens, GA, USA. [4]University of Texas Rio Grande Valley, Edinburg, TX, USA

*Corresponding author email: Pushpa.soti@utrgv.edu

ABSTRACT

With a nearly year-round growing season, tropical and subtropical regions are plagued with a myriad of agronomic challenges, including near-continuous weeds and invertebrate pests including plant-parasitic nematodes (PPNs). A poor understanding of the presence and geographical distributions of these pests complicate their management, especially in organic farming systems. This work attempts to document the interaction of PPNs with the major weeds in the semi-arid region of south Texas. Five organically managed farms were surveyed for four weeds of regional agronomic importance including silverleaf nightshade (*Solanum elaeagnifolium*), common sunflower (*Helianthus annuus*), false ragweed (*Parthenium hysterophorus*), and London rocket (*Sisymbrium irio*). Soil and root samples were collected to determine the presence of economically important PPNs in the roots and rhizosphere of the selected weed species. Eight different nematode genera associated with the selected weed species, including*Pratylenchus* spp., *Trichodorus* spp., *Criconemella* spp., *Helicotylenchus* spp., *Xiphinema* spp., *Dorylaimus* spp., *Aphelencoides* spp., and *Tylenchus* spp. were recorded. Four of the major economically important nematode genera *(Pratylenchus* spp, *Trichodorus* spp., *Criconemella* spp., *Xiphinema* spp.) were found in the rhizosphere of all four weeds. The two major PPN genera *Helicotylenchus* spp. and *Pratylenchus*spp., were largely associated with common sunflower, a major weed in the region. Our results indicate that these weed species can present additional challenges in agriculture, not only as direct competitors for resources to agronomic crops, but also as potential hosts for PPNs.

Additional index words: plant parasitic nematodes, organic farming, subtropics, weedy plants

Farmers in the tropics and subtropics face a wide range of biotic and abiotic constraints including weeds, insect pests, and poor soil fertility. With a year-round growing season, the potential for economic gain is limited by consistent pressure from insect pests and weeds. Economic losses due to weeds are exceptionally high, especially for organic growers (Soti and Racelis 2020). Weeds not only compete with crops for resources such as nutrients, water, and sunlight, they can also provide shelter for pathogens and pests (Norris and Kogan 2005), which cause a significant reduction in crop growth and yield. While non-crop plants and arthropods can potentially provide some agroecological benefits to farms in some instances (Mkenda et al. 2019), facilitative interactions between the non-crop plants and pests complicates the management of these pests (Norris and Kogan 2005).

Weeds are known to host several species of plant parasitic nematodes (Wrather 1992; Gazaway and McLean 2003; Myers et al. 2004; Thomas et al. 2005; Quénéhervé et al. 2006; Frankenberg et al. 2007). Quénéhervéet al. (2006) reported more than a dozen different weed species as host to *Pratylenchus* spp., *Helicotylenchus* spp., and *Meloidogyne* spp., which cause significant damage in a wide variety of crops. Weeds not only serve as hosts to nematodes, but they are also known to interfere with nematode management by providing protection from pesticides and adverse environmental conditions (Thomas et al. 2004, 2005). The global crop loss associated to plant parasitic nematodes (PPNs) are estimated at $80 billion annually, with 14% of that on the most economically important crops such as fruits, vegetables, and nonedible field crops (Nicol et al. 2011). Many PPN species cause direct damage as ectoparasites, endoparasites and semi-endoparasite by feeding externally on the root cortex, internally on stele region of the root and inside roots by penetrating half of their anterior portion of body into roots (Jenkins 1964). Major belowground symptoms caused by PPNs include root galling

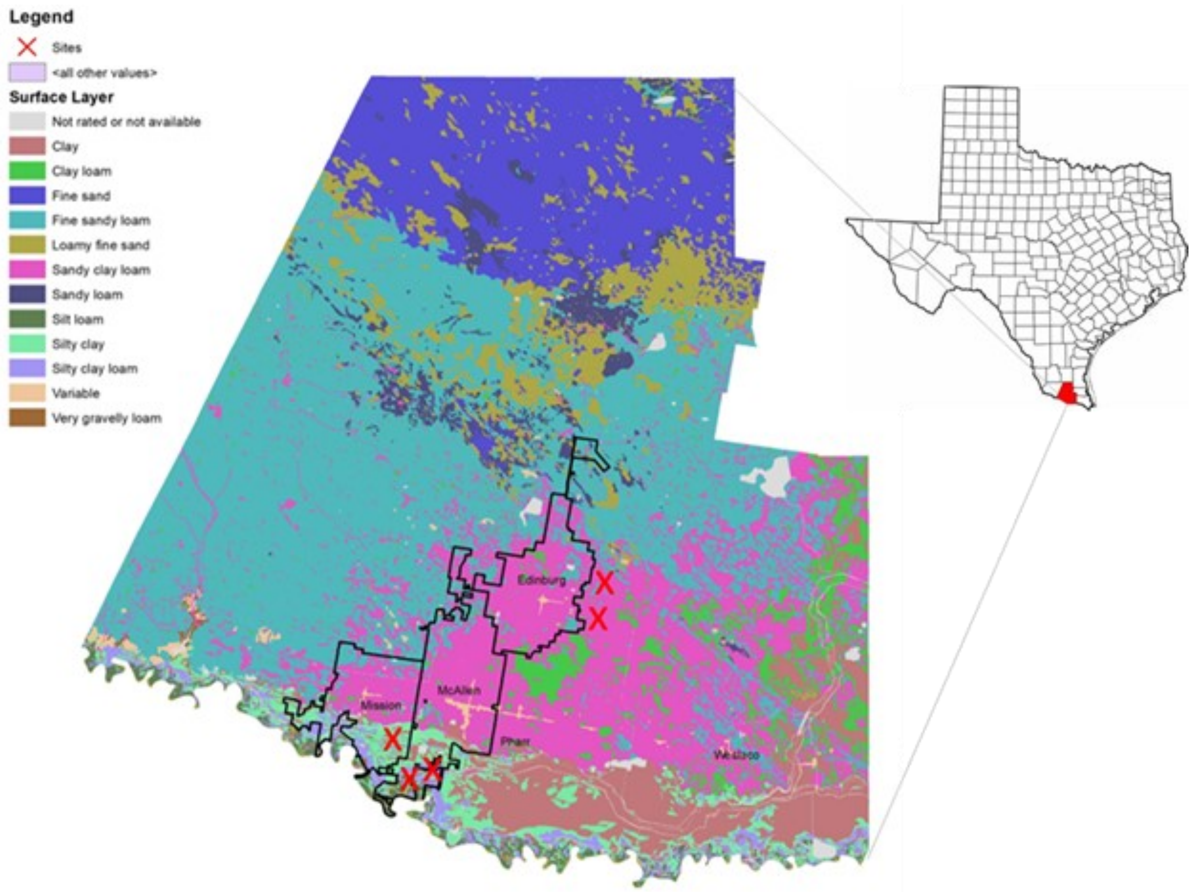

Fig. 1. Soil texture map of Hidalgo County, TX (Soil Survey Staff, 2019) marked with major city limits (for reference) and study site locations. (Texas state map inset).

(root-knots), root-lesions, and stubby-roots caused by *Meloidogyne* spp., *Pratylenchus* spp. and *Paratrichodorus* spp. nematodes, respectively. Above-ground symptoms include poor plant growth, yellowing, stunting, higher mortality, premature decline, and severely reduced yields (Dropkin 1989). In addition, injuries caused by PPNs including *Heterodera spp. Meloidogyne spp., Rotylenchulus* spp.,and*Pratylenchus* spp. on the roots allow the infection of many soilborne pathogens such as *Fusarium*, *Verticillium, Pythium, Phytophthora*, and *Rhizoctonia which cause disease complexes in crops* (Inserra and Lehman 1994; Robison et al. 1997). PPN species such as *Xiphinema*spp. and *Paratrichodorus* spp. are also known to serve as vectors of plant viruses (McGuire 1964; Brown 1989).

Weedy fallow, weeds growing along the farm edges, and resistant weeds within fields could provide a refuge for PPNs. Thus, understanding the interaction among pests is necessary to develop successful management strategies, especially for growers limited to non-chemical options. However, this association is poorly studied particularly in subtropical areas where the climatic conditions support a year-round growth of weeds and soil conditions are favorable for PPN growth (Sikora 2018). The goal of this study was to survey the potential of the four major weeds of tropical and warm temperate regions to host PPNs, including silverleaf nightshade (*Solanum elaeagnifolium*), common sunflower (*Helianthus annuus* L.), false ragweed (*Parthenium hysterophorus*), and London rocket

Table 1. Description of cultural practices and farm histories of five sampling sites/certified organic farms.

Sites	Cultural practices and farm histories
1	Seven years of continued multiple cropping (mixed species including vegetables and cover crops)
2	Vegetable field growing primarily brassicas and onions (rotation). No cover crops.
3	Vegetable field growing primarily brassicas and onions (rotation). No cover crops.
4	Vegetable field growing primarily brassicas and onions (rotation). No cover crops.
5	Multiple cropping for less than 2 years. Previously brushland, recently converted for Ag production.

(*Sisymbrium irio* L.). We collected rhizosphere soil and root samples of these weed species from five different farms and analyzed them for PPNs. These findings are discussed in the context of integrated weed and pest management strategies.

MATERIALS AND METHODS

Study site. The Lower Rio Grande Valley (LRGV) a four-county region in south Texas is characterized by a mild, subtropical climate which allows for year-round growing conditions for both agronomic crops and native and exotic weed species (Soti et al. 2020). Hidalgo county, at the center of the LRGV, has 9 certified organic farms within its borders, making it the county with highest number of certified organic growers in the state of Texas (Morris and Maggiani 2016). We conducted a field survey of PPNs in five certified organic agricultural fields across the county (Fig.1) during December 2018 through January 2019, the peak winter vegetable growing season in this region. Relevant data on various cultural practices and previous crop histories for each of these five sites are provided in Table 1.

Collection of samples. At each site 5 sampling locations with dense growth of each weed species were selected. About 25g of fresh root samples were collected from each location and mixed to make a composite root sample of each weed species. Similarly, composite rhizosphere soil samples and weed roots were collected from the selected five certified organic vegetable fields. Each composite sample consisted of 10-15soil cores collected directly from the rhizosphere of the weeds found in the field.

Cores were collected using a 1.27cm x 101.6cm step soil recovery probe (AMS; American Falls, ID). A total of 5 composite samples (one for each weed species) were collected from each of the 5 organic fields (a total of 25 samples). Root and soil samples were placed in a plastic bag and transported in a cooler to the lab, where soil samples were passed through a 2mm sieve to get rid of plant debris. Roots and soil samples were stored in a refrigerator at 4°C for 2-4 days, until nematode extraction was carried out. Remaining samples were analyzed for soil moisture, pH, organic matter, and texture. Soil pH was measured using an Oakton ion 700 bench meter (OAKTON Instruments, Vernon Hills, IL) in a soil: distilled water (1:2) mixture, organic matter was determined by loss on ignition method, and soil moisture was measured following the gravimetric method.

Nematode extraction and identification. Roots of the selected 4 weeds and rhizosphere soil samples were analyzed for the presence of PPNs. The nematodes were extracted from the 100 cm^3 soil sub-sample taken from each composite sample as described by Jenkins (1964). Five gram of root samples were incubated in water (23°C) for two days to extract endoparasitic migratory nematodes following the method by Young (1954). PPNs from each sample were identified morphologically to their genus level based on the morphological features following Mai's pictorial key to genera (Mai 2018) and Eisenback's identification guide (Eisencack2002) and counted using an inverted microscope (Leica DMi1, Buffalo Grove, IL). Abundance of each nematode genus and its association with each weed species was calculated as:

$$\frac{\text{Number of nematodes (genus x)}}{\text{Total number of nematodes recorded}} \times 100$$

RESULTS AND DISCUSSION

Soil characteristic was different among the different sampling sites. Soil moisture ranged from 10% to 21%, organic matter 3.15% to 4.96%. Soil pH was slightly alkaline (7.86- 8.23) in all sites. Soil texture ranged from sandy loam to silty clay (Table 2).

Table 2. Physical and chemical properties, and moisture of soil collected from each site.

Sites	Soil Moisture %	Organic Matter %	P^{H}	Soil Texture
1	11	3.37	7.86	Sandy Loam
2	16	3.46	8.07	Silty Clay Loam
3	18	2.94	8.23	Silt Loam
4	21	4.96	8.17	Silty Clay
5	10	3.15	7.86	Loamy Sand

A total of 1376 plant parasitic nematodes were counted across all samples. We found 8 different PPNs, of which,6were of agronomic importance. Ring (*Criconemella* spp.), lesion (*Pratylenchus*spp.), stubby-root (*Trichodorus* spp.), spiral (*Helicotylenchus* spp.), dagger (*Xiphenema* spp.), and foliar (*Aphelencoides* spp.) nematodes were associated with the selected weeds (Table 3). We recorded a high number of lesion (*Pratylenchus*spp.) and spiral (*Helicotylenchus* spp.) in all the weed species, suggesting that these weeds may be highly susceptible hosts of these two species of nematodes. *Trichodorus*spp., *Xiphenema* spp., *Criconemella* spp, and *Phelencoides* spp. were found in relatively smaller numbers. Foliar nematodes, *Tylenchus*spp. and *Aphelenchoides* spp. (fungivores), capable of causing serious damage to . foliage of many crops and ornamental plants (Jagdale and Grewal 2002; Sánchez Monge et al. 2015), were also found in small numbers in the soil but more research is needed to confirm their pathogenicity to these weed species.

All eight PPN genera were found in the rhizosphere

Table 3. Total number of nematodes extracted from 100 cm3 soil and their relative abundance (in parentheses) of plant-parasitic nematodes on 4 weed species from 5 sites.

Nematode genus	Weed species and mean number of nematodes			
	H. annuus	*S. elaeagnifolium*	*S. irio*	*P. hysterophorus*
Pratylenchus spp.*	258 (78%)	35 (10%)	40 (12%)	16 (5%)
Trichodorus spp.*	13 (57%)	3 (13)	4(17%)	3 (13%)
Criconemella spp.*	1(50%)	1(50)	-	-
Helicotylenchus spp.*	351 (43%)	232 (29%)	90 (11%)	120 (17%)
Xiphenema spp.*	1 (9%)	4 (36%)	3 (36%)	1(18%)
Dorylaimus spp.	4 (50)	-	3 (38%)	1 (13%)
Aphelencoides spp.*	2 (22%)	4 (44%)	3 (33%)	-
Tylenchus spp.	7 (27%)	13 (43%)	5 (17%)	4 (13%)

associated with *Helianthus annus*, a major weed in the region (Soti et al. 2020). The rhizosphere of *S. elaeagnifolium* and *S. irio* had seven PPN genera while *P. hysterophorus* had the lowest, six PPN genera. Our results are consistent with previous studies that reported *H. annus* (Bolton et al. 1989; Singh et al. 2009) and *S. elaeagnifolium* (Wapshere 1988; Anwar et al. 2009) to be hosts of PPNs. We recovered a very few numbers of PPNs from the roots of the selected weeds (Table 4).

Table 4. Descriptive table of each nematode genus found in roots in relation to weed species and percent abundance. Data reported represent number found per 5g roots and percent abundance of genus.

Nematodes	Weed species			
	H. annuus	*S. elaeagnifolium*	*S. irio*	*P. hysterophorus*
Pratylenchus spp.*	-	-	2 (67%)	1 (33%)
Helicotylenchus spp.*	-	3 (17%)	1 (6%)	14 (78%)
Xiphinema spp.*	-	1 (33%)	1 (33%)	1 (33%)
T*ylenchus* spp.	1 (100%)	-	-	-

*Economically important genera (Jones 2013)

P. hysterophorus had the highest numbers (16), followed by *S. elaeagnifolium* (3) and *S. Irio* (1). In addition, only four PPN types were found in the roots of the weed species, with *Helicotylenchus* spp. being the most dominant one. Root knot nematode (*Meloidogyne incognita*) reported to cause a substantial damage to oil seed sunflower (Mukhtar 2009; Tzortzakakis et al. 2014) were not found in our study.

The nematode species recorded in this study are different from the species previously reported for this region. In their survey of cotton, citrus, and fallow fields in the LRGV, Robinson et al. (1987) recorded the presence of only three PPNs (*Rotylenchulusreniformis*, *Meloidogyne incognita*, and *Tylenchus. semipenetrans*). However, in our survey of organic vegetable farms we did not record any of the previously recorded species, reflecting how farm management and the crop selection can influence the diversity of PPNs that may exist in the area.

The weed species used in this study are considered of major economic value which infest vegetable, grains, and cotton, the major crops grown in this region and to other subtropical areas around the world (Soti et al. 2020). Our results indicate that these weeds can potentially serve to facilitate a persistent, year-round population of PPNs. For example, LRGV is a national leader in grain sorghum production, and *H. annuus*--a major weed in sorghum--harbored a relatively high number of *Pratylenchus scribneri,* also a major pest of sorghum (Motalaote et al. 1987; Blancard 2012). Weed management is costly, particularly for organic growers.

Organic farms in general are reported to have higher weed density and diversity (Roschewitz et al. 2005; Henckel et al. 2015) compared to conventional farms. Our results support previous studies that weeds, if unmanaged, can shelter nematodes in the field margins during the cropping season and in the field during fallow period (Thomas et al. 2005; Kutywayo and Been 2006). Furthermore, many farmers adopt an economic threshold principle in making weed control decision where weed control is only done after the population reaches a certain threshold. While our samples were collected during the fallow period, our results indicate that weeds can cause a significant impact on the crops not only by competing for resources such as light, nutrients, and water but also by harboring plant parasitic nematodes. Thus, management plans, especially in organic farms should consider not only the direct benefit of weed eradication, but also consider the implication of weeds management on other pest populations and manage them simultaneously.

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LITERATURE CITED

Anwar S, Zia A, Javed N, Shakeel Q (2009) Weeds as reservoir of nematodes. Pak.J.Nematol 27(2):145 -153.

Blancard D (2012) Tomato diseases: Identification, biology and control: A colour handbook. 2nd ed CRC Press, London, UK.

Bolton C, De Waele D, Loots G (1989) Plant-parasitic nematodes on field crops in South Africa. III: Sunflower. Revue De Nématologie 12(1):69-75.

Brown D, Ploeg A, Robinson D (1989) The association between serotypes of tobraviruses and Trichodorus and Paratrichodorus species 1. EPPO Bulletin 19(3):611-7.

Dropkin VH (1989) Introduction to plant nematology. John Wiley and Sons Inc.

Eisenback J (2002) Identification guides for the most common genera of plant-parasitic nematodes. Mactode Publications.

Frankenberg A, Paffrath A, Hallmann J, Schmidt H (2007) Occurrence and importance of plant-parasitic nematodes in organic farming in Germany. Nematology 9(6):869-79.

Gazaway W and McLean K (2003) A survey of plant-parasitic nematodes associated with cotton in Alabama. Journal of Cotton Science 7(1):1-7.

Henckel L, Börger L, MeissH, Gaba S, Bretagnolle V (2015) Organic fields sustain weed metacommunity dynamics in farmland landscapes. Proceedings of the Royal Society B: Biological Sciences, 282(1808), 20150002.

Inserra R, Lehman P, Overstreet (1994) Ornamental hosts of the reniform nematode, *Rotylenchus reniformis*. Florida Department of Agriculture & Consumer Services, Division of Plant Industry, Gainesville, FL. Nematology Circular (209).

Jagdale GB, Grewal PS (2002). Identification of alternatives for the management of foliar nematodes in floriculture. Pest Management Science, 58(5), 451-458.

Jenkins W (1964) A rapid centrifugal-flotation technique for separating nematodes from soil. Plant Disease Reporter 48(9).

Jones JT, Haegeman A, Danchin EG, Gaur HS, Helder J, Jones MG, Kikuchi T, Manzanilla López R, Palomares Rius JE, Wesemael WM (2013) Top 10 plant parasitic nematodes in molecular plant pathology. Molecular Plant Pathology 14(9):946-61.

Kutywayo V, Been TH (2006) Host status of six major weeds to *Meloidogyne chitwoodi* and *Pratylenchus penetrans*, including a preliminary field survey concerning other weeds. Nematology, 8 (5), 647-657.

Mai W (2018) Plant-parasitic nematodes: A pictorial key to genera. 5th ed. Ithaca: Cornell University Press.

McGuire J (1964) Efficiency of *Xiphinema americanum* as a vector of tobacco ringspot virus. Phytopathology 54(7):799-801.

Motalaote B, Starr J, Frederiksen R, Miller F (1987) Host status and susceptibility of sorghum to pratylenchus species. Revue De Nématologie 10 (1):81-6.

Mkenda PA, Ndakidemi PA, Mbega E, Stevenson PC, Arnold SE, Gurr GM, Belmain SR (2019) Multiple ecosystem services from field margin vegetation for ecological sustainability in agriculture: scientific evidence and knowledge gaps. PeerJ, 7 (3): e8091.

Morris M, Maggiani R (2016) Who are the organic farmers in Texas. National Center for Appropriate Technology. https://attra.ncat.org/downloads/TX_Organic_Farmers.pdf.

Myers L, Wang K, McSorley R, Chase C (2004) Investigations of weeds as reservoirs of plant-parasitic nematodes in agricultural systems in northern Florida. Pages 256-265 *in* 26th Southern Conservation Tillage Conference for sustainable agriculture. North Carolina State University Technical Bulletin TB: Citeseer.

Mukhtar I (2009) Sunflower disease and insect pests in Pakistan: A review. African crop science journal, 17(2).

Nicol J, Turner S, Coyne D, Den Nijs L, Hockland S, Maafi ZT (2011) Current nematode threats to world agriculture. Pages 21-43 in Genomics and molecular genetics of plant-nematode interactions. Springer.

Norris RF, Kogan M (2005) Ecology of interactions between weeds and arthropods. Annu. Rev. Entomol., 50:479-503.

Quénéhervé P, Chabrier C, Auwerkerken A, Topart P, Martiny B, Marie-Luce S (2006) Status of weeds as reservoirs of plant parasitic nematodes in banana fields in martinique. Crop Protection 25 (8):860-7.

Robinson AF, Heald CM, Flanagan SL, Thames WH, Amador J (1987) Geographical distributions of R*rotylenchulus reniformis*, *Meloidogyne incognita*, and *Tylenchulus semipenetrans* in the Lower Rio Grande valley as related to soil texture and land use. J Nematol 19(Annals 1):20-5.

Robinson A, Inserra R, Caswell-Chen E, Vovlas N, Troccoli A (1997) *Rotylenchulus* species: Identification, distribution, host ranges, and crop plant resistance. Nematropica 27(2):127-80.

Roschewitz, I., Gabriel, D., Tscharntke, T., &Thies, C.

(2005) The effects of landscape complexity on arable weed species diversity in organic and conventional farming. Journal of applied ecology, 42 (5):873-882.

Sánchez Monge GA, Flores L, Salazar L, Hocland S, Bert W (2015) An updated list of the plants associated with plant-parasitic aphelenchoides (nematoda: Aphelenchoididae) and its implications for plant-parasitism within this genus. Zootaxa, 4013(2): 207–224.

Sikora RA, Coyne D, Hallmann J, Timper P (2018) Plant parasitic nematodes in subtropical and tropical agriculture. Wallingford, UK: Cabi 3rd ed pp. 795-839.

Singh S, Tyagi S, Prasad D (2009) Distribution pattern of plant parasitic nematodes infesting sunflower. Annals of Plant Protection Sciences 17(2):443-6.

Soti P, Goolsby JA, Racelis A (2020) Agricultural and environmental weeds of south Texas and their management. Subtropical Agriculture and Environments 71:1-11.

Soti P, Racelis A (2020). Cover crops for weed suppression in organic vegetable systems in semiarid subtropical Texas. Organic Agriculture, 1-8.

Thomas SH, Schroeder J, Murray LW (2004). Cyperus tubers protect *Meloidogyne incognita* from 1, 3-dichloropropene. Journal of nematology, 36(2): 131.

Thomas SH, Schroeder J, Murray LW (2005). The role of weeds in nematode management. Weed Science, 53(6): 923-928.

Tzortzakakis EA, Anastasiadis AI, Simoglou KB, Cantalapiedra-Navarrete C, Palomares-Rius J E, Castillo P (2014) First report of the root-knot nematode, *Meloidogyne hispanica*, infecting sunflower in Greece. Plant disease, 98(5): 703-703.

Wapshere A, (1988) Prospects for the biological control of silver-leaf nightshade, *Solanum elaeagnifolium*, in Australia. Aust J Agric Res 39(2):187-97.

Wrather JA, Niblack TL, Milam MR (1992) Survey of plant-parasitic nematodes in Missouri cotton fields. J Nematol 24(4S):779-82.

Young T (1954) An incubation method for collecting migratory endoparasitic nematodes. Plant Disease Reporter 38(11):794-5.