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Special Topics

Weed Hosts of Root-Knot Nematodes and Their Distribution in Fiji

Sunil K. Singh, Uma R. Khurma, and Peter J. Lockhart*

Weeds can act as reservoir hosts of a range of pests and diseases. Information and knowledge on the host status of weeds to common pests and diseases can be used to develop integrated weed and pest management strategies. As part of a survey on the distribution and diversity of root-knot nematodes on crops in Fiji, the root-knot nematode host status of weeds was also studied. Weeds growing in root-knot nematode infested farms ($n = 189$) and bioassay pot soil samples ($n = 277$) were identified, and their host status was determined on the basis of a root gall and egg-mass index scale from 0 to 5. A total of 45 weed species were recorded as potential weed hosts of root-knot nematodes with a gall index from 1 to 5. Using the weed and tomato bioassay method, a total of 11 nonhost weed species were recorded with a gall index of 0, relative to infected tomato growing in pot soil samples. Common weeds infected by root-knot nematodes on farms and in bioassay pot soil included slender amaranth, old world diamond-flower, tropic ageratum, sicklepod, mimbra, balsamapple, purple bushbean, little ironweed, ivy gourd, and cutleaf groundcherry. The presence of egg masses on the weed hosts indicated their ability to sustain root-knot nematode populations and, thus, their potential to act as reservoir hosts.
Nomenclature: Root-knot nematodes, *Meloidogyne* Göldi; balsamapple, *Momordica charantia* L.; cutleaf groundcherry, *Physalis angulata* L.; ivy gourd, *Coccinia grandis* (L.) J. Voigt; little ironweed, *Cyanthillium cinereum* (L.) H. Rob.; old world diamond-flower, *Oldenlandia corymbosa* L.; purple bushbean, *Macroptilium atropurpureum* (Moc. & Sesse ex DC.) Urb.; seedbox, *Ludwigia hyssopifolia* (G. Don) Excell apud A. R. Fernandes; sicklepod, *Senna obtusifolia* (L.) H.S. Irwin & Barnaby; slender amaranth, *Amaranthus viridus* L.; tropic ageratum, *Ageratum conyzoides* L.; garden tomato, *Solanum lycopersicum* L.
Key words: Nematode management, *Meloidogyne*, reservoir host, nematodes.

Las malezas pueden actuar como hospederas de una amplia variedad de plagas y enfermedades. La información y los conocimientos acerca de la manera en que las malezas pueden ser hospederas de las plagas y enfermedades comunes pueden utilizarse para desarrollar estrategias integradas del manejo de éstas mismas. Como parte de una encuesta acerca de la distribución y diversidad de los nemátodos del género Meloidogyne en cultivos producidos en Fiji, también se estudio el grado en que las malezas funcionan como hospederas de dichos organismos. Se identificaron malezas que crecen en granjas infestadas con Meloidogyne ($n = 189$) y en muestras de ensayo de tierra de macetas ($n = 277$) y su estatus de hospederas fue determinado en base a los nódulos radiculares y a la masa de huevecillos presente en una escala de 0 a 5. Un total de 45 especies de maleza fueron identificadas como hospederas potenciales de Meloidogynes con un índice de nódulos radiculares de 1 a 5, usando el método de bio-ensayo en malezas y tomate, un total de 11 especies de malezas no hospederas se registraron con un índice de nódulos de 0, en relación al tomate infectado que crecía en las muestras de tierra de macetas. Las malezas comunes infectadas con Meloidogynes en granjas y en muestras de suelo de macetas, incluyeron *Amaranthus viridus* L., *Oldenlandia corymbosa* L.; *Ageratum conyzoides* L.; *Senna obtusifolia* (L.) H.S. Irwin & Barneby; *Ludwigia hyssopifolia* (G.Don) Excell apud A. R. Fernandes; *Momordica charantia* L.; *Macroptilium atropurpureum* (Moc. & Sesse ex DC.) Urb.; *Cyanthillium cinereum* (L.) H. Rob.; *Coccinia grandis* (L.) J. Voigt y *Physalis angulata* L. La presencia de masas de huevecillos en las malezas hospedaras indica su habilidad para mantener poblaciones de Meloidogynes y por lo tanto, su potencial para actuar como hospederas.

Weeds are often found on farms in between the furrows, along farm boundaries, and on fallow farms. Weeds growing with crop plants not only compete for light, space, water, and nutrients but also can serve as reservoirs of pests, including plant-parasitic nematodes (Davis and Webster 2005). In the absence of a suitable crop host, weeds can act as alternative hosts for plant-parasitic nematodes and help to maintain nematode populations targeted for suppression by various management strategies (Thomas et al. 2005). The presence of weed hosts reduces the efficacy of nematode management strategies (Belair and Benoit 1996; Thomas et al. 2005); hence, nematode management and weed management require simultaneous consideration.

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Most of the crop damage by plant-parasitic nematodes is caused by a relatively small number of nematode genera, which include root-knot nematodes (*Meloidogyne* spp.), cyst nematodes (*Globodera* and *Heterodera* spp.), and migratory endoparasitic nematodes (*Pratylenchus* and *Radopholus* spp.) (Bird and Kaloshian 2003). Root-knot nematodes are well known for their widespread distribution and extremely polyphagous nature with more than 3,000 plant hosts, including crops, ornamentals, medicinal and aromatic plants, and weeds (Abad et al. 2003; Haseeb and Pandey 1995; Sharma and Rich 2005). The significance of weeds acting as reservoir hosts is being increasingly recognized, and more than 226 weed species have been studied for their host suitability to different root-knot nematodes worldwide (Rich et al. 2009).

The loss of broad-spectrum soil fumigants, such as methyl bromide, has increased reliance on integrated nematode management techniques, which require detailed knowledge of nematode biology and ecology. A number of research studies recommend the use of both nematicides and herbicides

for effective control of nematodes. However, nematicide and herbicide use is costly, is less ecofriendly, and is not suited for subsistence and organic farming. Knowledge of the host status of weeds common in agricultural systems can be used to develop targeted weed management to improve efficacy of the nematode management strategies used, especially in subsistence and organic farming. In a preliminary survey, root-knot nematode infections were noted from a number of commonly grown crops and a few weed species in Fiji (Khurma et al. 2008). This led to a more extensive and systematic survey of root-knot nematode diversity and distribution in Fiji. While surveying the farms in the current study, the main focus was on the crop hosts, but encountering a number of weeds infected with root-knot nematodes led to examining weeds in conjunction with crop hosts on all farms. This article reports findings on weed hosts of root-knot nematodes and discusses the implications with respect to nematode management and farming practices in Fiji. Our findings describing morphological and molecular characterizations of Fijian root-knot nematodes are presented elsewhere.

Materials and Methods

As part of a root-knot nematode survey, along with crop hosts, weeds growing on farms were also sampled from Viti Levu, Fiji. Fiji islands cover a land area of 18,333 km^2 and are located in the tropical region between 174°E and 178° W of Greenwich and between latitudes 12°S and 22°South (Fiji Islands Bureau of Statistics 2008). There are two major islands: Viti Levu, which covers an area of 10,429 km^2, and Vanua Levu, which covers an area of 5,556 km^2. The climatic conditions are tropical, with two seasons. The cool and dry season is from May to October, with an average temperature range of 19 to 22 C, whereas the hot and wet season is from November to April, with an average temperature range of 31 to 34 C (Fiji Islands Bureau of Statistics 2008).

The most commonly grown crops in Fiji include sugarcane (*Saccharum officinarum* L.), kidney beans (*Phaseolus vulgaris* L.), eggplant (*Solanum melongena* L.), okra [*Abelmoschus esculentus* (L.) Moench], bele [*Abelmoschus manihot* (L.) Medik.], coco yam [*Colocasia esculenta* (L.) Schott], cassava (*Manihot esculenta* Crantz), sweet potato [*Ipomoea batatas* (L.) Lam.], lesser yam [*Dioscorea esculenta* (Lour.) Burkill], cultivated tobacco (*Nicotiana tabacum* L.), garden lettuce (*Lactuca sativa* L.), cabbage (*Brassica oleracea* L.), garden tomato, garden cucumber (*Cucumis sativus* L.), winter squash (*Cucurbita maxima* Duchesne), watermelon [*Citrullus lanatus* (Thunb.) Matsum. & Nakai], papaya (*Carica papaya* L.), banana (*Musa* L.), coconut palm (*Cocos nucifera* L.), garden ginger (*Zingiber officinale* Roscoe), and kava (*Piper methysticum* G. Forst.). The farms, which primarily grow vegetables, from all 10 localities on Viti Levu, Fiji (Figure 1), were sampled for more than a 1-yr period (April 2007 to April 2008), covering both cropping and noncropping seasons. The farms were selected randomly with a distance of at least 2 km separating the farms. Different weeds that were present on farms (n = 675) on Viti Levu were dug out, and the soil adhering to the root system was removed by hand before examining the entire root system for the presence of root galls,

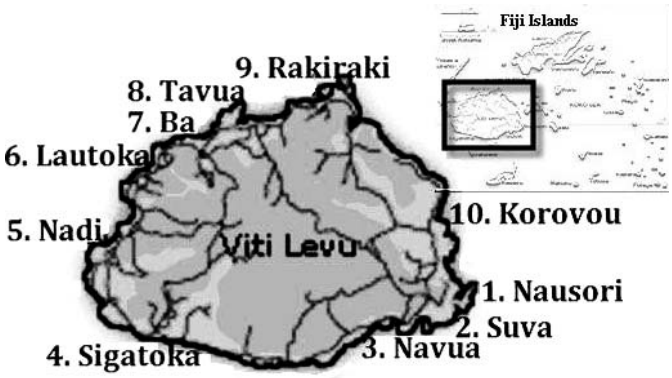

Figure 1. Map showing approximate locations of the 10 sampled localities.

characteristic of root-knot nematode infection. Weeds and crop plants from various different points on the farm were inspected, starting from the farm boundaries and moving across the length of the field in a zigzag pattern. In addition to examining the roots of the weed and crop plants, composite soil samples of approximately 2 kg consisting of 10 subsamples were taken from depths of 0 to 30 cm using a hand shovel from the root rhizosphere region of the sampled plants across the length of the fields surveyed.

The weeds found infected by root-knot nematodes in the field were brought to the University of the South Pacific (Suva, Fiji) research laboratory, where the nematodes were identified, and the number of root galls and egg masses were counted during examination with a stereoscopic microscope.[1] The field soil samples were placed in 2-kg plastic pots and kept in a screen house to allow weeds to germinate. A bioassay for the presence of root-knot nematodes in the soil sample was done by planting a susceptible host plant (*Solanum lycopersicum* 'Moneymaker') in the soil samples. The tomato plant and the germinating weeds were allowed to grow in the soil samples for 8 wk to enable root-knot nematode juveniles to infect and mature to adults on susceptible weeds and the tomato plant. The pots were kept 30 cm apart on a raised bench and watered daily. Normally, the thinning was done to at least two weed and two host plants per pot. Multiple species were assessed in the pots where more than one species had emerged. In such pots, plants were thinned to at least one weed plant per species to allow growth of multiple weed species and two tomato host plants. The germinating weed species were not always the same as those found in the field.

At the end of the eighth week, the weeds and tomato plants were gently removed from the pot, and their roots were examined under a stereoscopic microscope to determine the presence or absence of root galls and egg masses. The weeds found infected were identified, and the number of root galls and egg masses were recorded and rated on a root gall and egg mass index scale from 0 to 5 (0, no galls or egg masses; 1, 1 or 2 galls or egg masses; 2, 3 to 10 galls or egg masses; 3, 11 to 30 galls or egg masses; 4, 31 to 100 galls or egg masses; and 5, more than 100 galls or egg masses), as outlined by Taylor and Sasser (1978). The count was based on a minimum of 10 specimens per weed species. Weeds that allowed the development of egg masses with an index range of 1 to 5

Table 1. Potential weed hosts found infected via direct examination method from farms on Viti Levu, Fiji.[a]

Weed species	Family	Common name	*n*	Root-gall index[b]	Localities[c]	*Meloidogyne* spp.[d]
Ageratum conyzoides L.	Asteraceae	ageratum, tropic	80	5	1, 4, 5,7	Ma, Mi, Mj
Amaranthus viridus L.	Amaranthaceae	amaranth, slender	80	5	2, 4, 5, 7	Mi, Mj, Mx
Coccinia grandis (L.) J. Voigt	Cucurbitaceae	gourd, ivy	24	4	2, 7, 10	Mi, Mj
Commelina benghalensis L.	Commelinaceae	dayflower, Benghal	21	2	1, 2, 3	Mi, Ma
Cyperus compressus L.	Cyperaceae	sedge, annual	27	2	1, 4, 7	Mi, Mj
Echinochloa colona (L.) Link	Poaceae	junglerice	34	2	4, 6, 7	Mi
Eleusine indica (L.) Gaertn.	Poaceae	goosegrass	68	2	4, 5, 7, 8, 9	Mi, Ma
Impatiens walleriana (Hook.f) (source: PIER)	Balsaminaceae	busy lizzy, balsam	17	5	4	Ma, Mi
Ipomoea obscura (L.) Ker Gawl. (source: PIER)	Convolvulaceae	morningglory, obscure	28	2	2, 3, 8	Mj
Ipomoea quamoclit L.	Convolvulaceae	morningglory, cypressvine	39	2	4, 5, 6, 7	Mi, Mj
Ipomoea triloba L.	Convolvulaceae	morningglory, threelobe	32	2	1, 2, 4, 5	Mi
Ludwigia hyssopifolia (G. Don) Exell apud A. R. Fernandes	Onagraceae	seedbox	44	3	1, 2, 3	Mi, Mj, Mx
Malvastrum coromandelianum (L.) Garcke (source: PIER)	Malvaceae	broomweed	57	2	1, 4, 7	Mi, Ma
Mikania micrantha Kunth	Asteraceae	mile-a-minute	36	1	1, 4	Mi
Momordica charantia L.	Cucurbitaceae	balsamapple	15	3	5	Mj
Oldenlandia corymbosa L.	Rubiaceae	diamond-flower, old world	56	5	1, 2, 4	Mi, Mj, Mx
Physalis angulata L.	Solanaceae	groundcherry, cutleaf	52	5	2, 3, 4, 5, 8	Mi, Mj
Piper aduncum L.	Piperaceae	pepper, spiked	22	2	1, 10	Mj
Senna obtusifolia (L.) H.S. Irwin & Barnaby	Fabaceae	sicklepod	68	4	5, 6, 9	Mi, Mj, Ma
Senna occidentalis (L.) Link	Fabaceae	senna, coffee	46	2	4, 5, 7, 8	Mi
Setaria palmifolia (J. Koenig) Stapf	Poaceae	palmgrass	31	1	2, 4, 9	Mj, Ma
Solanum torvum Sw.	Solanaceae	turkey berry	21	3	1, 4	Mi
Stachytarpheta cayennensis (Rich.) Vahl (source: PIER)	Verbenaceae	blue rat's tail	25	2	4, 6	Mi, Mj
Synedrella nodiflora (L.) Gaertn. (source: PIER)	Asteraceae	nodeweed	70	1	1, 2, 4	Mi
Triumfetta rhomboidea Jacq.	Tiliaceae	paroquet-bur	37	3	4, 6, 7	Mi
Cyanthillium cinereum (L.) H. Rob.	Asteraceae	ironweed, little	70	5	1, 3, 4, 7	Mi, Mj

[a] All the weeds except 9, 10, 11, 15, 20, and 22, also emerged in the bioassay pots. The *n* column refers to the sample size of weeds. Weed scientific names, families, common names, and authorities have been cited from the following sources listed in the Literature Cited: WSSA 2009, USDA 2009, and PIER 2009.

[b] Root gall index 0, no galls or egg masses; 1, 1 or 2 galls or egg masses; 2, 3–10 galls or egg masses; 3, 11–30 galls or egg masses; 4, 31–100 galls or egg masses; 5, more than 100 galls or egg masses.

[c] Localities on Viti Levu: 1, Nausori; 2, Suva-Nasinu; 3, Navua; 4, Sigatoka; 5, Nadi; 6, Lautoka; 7, Ba; 8, Tavua; 9, Rakiraki; 10, Korovou.

[d] Root-knot nematode (*Meloidogyne*) species parasitizing weeds: Ma, *M. arenaria*; Mi, *M. incognita*; Mj, *M. javanica*; Mx, mixed (Mi + Mj).

are listed as potential hosts (Tables 1 and 2), whereas those that did not develop root-galls or egg mass, those with an index of 0, are listed as nonhosts (Table 3) for the particular root-knot nematode species. The weed nonhosts in Table 3 are based on bioassay observations in infested soil and exclude the weeds that were found uninfected in the field to avoid false negative results.

The weeds found growing in root-knot nematode–infested soil were photographed and identified with the help of reference literature (Parham 1958; Seemann and Fitch 1865; Smith 1979; Whistler 1995), and identifications were verified in consultation with an expert botanist (A. Whistler, personal communications). The root-knot nematodes from this survey were identified by sequence-characterized amplified region (SCAR) molecular markers for the three most commonly occurring species: *M. incognita* MIF/MIR (Meng et al. 2004), *M. arenaria* Far/Rar (Zijlistra et al. 2000) and *M. javanica* Fjav/Rjav (Zijlistra et al. 2000). The morphological features of female, juvenile, and male specimens of root-knot nematode populations were also studied to verify the species identity of root-knot nematodes and to differentiate between the uncommon species (data not shown).

Results and Discussion

Cultural pest management strategies, such as rotation using nonhost crops and rotation with resistant cultivars of susceptible crops, as well as leaving farms fallow, are often implemented by farmers to reduce nematode and other pest populations. Pest management techniques that rely on starvation of pest populations will only be effective if there are no alternative hosts on the farms. The 45 species of weed hosts found in this study, belonging to 23 different families, are a clear indication that these weeds can act as alternative hosts of root-knot nematodes when present on farms (Table 1 and 2). Out of 675 farms sampled from Viti Levu, Fiji, root-knot nematodes were found on 189 farms (28%) and in an additional 88 soil samples after bioassay.

Because the evaluation of the weeds was based on field observations and bioassays, the infected weeds in this study are reported as potential hosts for the following reasons. First, the initial population densities of the root-knot nematodes was not assessed; hence, the variability in root galling and egg mass number could be due to variable levels of inoculum density in field soil samples. Second, the distribution of nematodes in the field is not uniform; hence, it may have contributed to variability in root-gall formation observed via the direct examination method. However, the effects of low population density and patchy distribution of root-knot nematodes in field soil could have been overcome to an extent because of the composite soil samples used in the bioassays. A more conclusive observation on the host status can be made using a known inoculum density, where the reproduction potential of root-knot nematodes on specific weed species can be

Table 2. Potential weed hosts found infected from soil samples collected from farms on Viti Levu, Fiji, via the bioassay method.[a]

Weed species	Family	Common name	*n*	Root-gall index[b]	Localities[c]	*Meloidogyne* spp.[d]
Alternanthera sessilis (L.) R. Br. ex DC.	Amaranthaceae	joyweed, sessile	26	4	1, 4, 5	Mi
Clitoria ternatea L. (source: PIER)	Fabaceae	butterfly pea	23	2	4, 5	Mi
Cuphea carthagenensis (Jacq.) Macbr.	Lythraceae	cuphea, tarweed	28	4	1, 2, 6	Mi
Digitaria ciliaris (Retz.) Koel.	Poaceae	crabgrass, southern	40	2	2, 3, 4, 5, 7, 8, 10	Mi
Digitaria setigera Roth ex Roem. & Schult. (source: USDA)	Poaceae	east Indian crabgrass	35	3	3, 4, 10	Mi
Echinochloa crus-galli (L.) Beauv.	Poaceae	barnyardgrass	35	2	2, 3, 6, 7, 9	Mi, Mj
Eleutheranthera ruderalis (Sw.) Sch. Bip. (source: PIER)	Asteraceae	ogiera	35	2	2, 3, 5, 6	Mi
Fimbristylis dichotoma (L.) Vahl	Cyperaceae	fingerush, forked	28	3	5, 7, 10	Mi
Hyptis pectinata (L.) Poit.	Lamiaceae	hyptis, comb	36	5	1, 2, 4, 5	Mi
Lindernia nummulariifolia (D. Don) Wettst. (source: PIER)	Scrophulariaceae		22	2	1, 2, 3, 10	Mi
Macroptilium atropurpureum (Moc. & Sesse ex DC.) Urb.	Fabaceae	purple bushbean	23	5	4, 5, 6, 7, 8	Ma, Mi
Mollugo verticillata L. (source: USDA)	Aizoaceae	Carpetweed	13	4	2, 3, 10	Mi
Oxalis corniculata L.	Oxalidaceae	woodsorrel, creeping	30	4	1, 2, 3, 5, 8	Mi
Oxalis debilis Kunth var. corymbosa (DC.) Lourteig	Oxalidaceae	woodsorrel, pink	36	4	1, 2, 3, 4, 5, 6, 8, 9, 10	Mi
Peperomia pellucida (L.) Kunth.	Piperaceae	shiny bush	27	4	2, 3, 5, 10	Mi
Phyllanthus amarus Schumach. & Thonn. (source: PIER)	Euphorbiaceae	sleeping plant	43	5	2, 4, 5, 6, 7	Mj, Mx
Phyllanthus urinaria L.	Euphorbiaceae	chamber-bitter	35	4	4, 5, 6, 7,	Mj
Portulaca oleracea L.	Portulacaceae	purslane, common	29	4	1, 2, 4, 9, 10	Mi, Mx
Sporobolus indicus (L.) R. Br.	Poaceae	smut grass	30	2	4, 5, 6, 8	Mi

[a] The *n* column refers to the sample size of weeds. Weed scientific names, families, common names, and authorities have been cited from the following sources listed in the Literature Cited: WSSA 2009, USDA 2009, and PIER 2009.

[b] Root gall index 0, no galls or egg masses; 1, 1 or 2 galls or egg masses; 2, 3–10 galls or egg masses; 3, 11–30 galls or egg masses; 4, 31–100 galls or egg masses; 5, more than 100 galls or egg masses.

[c] Source locality of bioassay soil samples: 1, Nausori; 2, Suva-Nasinu; 3, Navua; 4, Sigatoka; 5, Nadi; 6, Lautoka; 7, Ba; 8, Tavua; 9, Rakiraki; 10, Korovou.

[d] Root-knot nematode (*Meloidogyne*) species parasitizing weeds: Ma, *M. arenaria*; Mi, *M. incognita*; Mj, *M. javanica*; Mx, mixed (Mi + Mj).

evaluated. For example, dayflower Benghal (*Commelina benghalensis* L.) has been reported as a good host for the southern root-knot nematode (*M. incognita*) based on reproductive potential (Davis et al. 2006), whereas in the current study the southern root-knot infections were not found to be as severe on the same weed species.

Root-knot nematode species acted differently toward different species of weeds, with varying levels of galling and reproductive potential, as indicted by the root gall and egg-mass indices. Such differences in the host status of weeds have been documented in other studies as well (Davis et al. 2006; Desaeger and Rao 2000; Ehwaeti et al. 1999; Kaur et al. 2007). Some weed species had relatively higher root-gall and egg-mass indices that ranged from 3 to 5 (Tables 1 and 2). This level of infection produced more inoculum for the next cropping season. The weed species with root-gall and egg-mass indices that ranged from 1 to 2 (Tables 1 and 2) had relatively limited ability to sustain root-knot nematode populations.

Table 3. Weeds found to be non hosts of root-knot nematodes infesting soil on farms on Viti Levu, Fiji via bioassay method.[a]

Weed species	Family	Common name	*n*	Root-gall index[b]	Localities[c]	*Meloidogyne* spp.[d]
Chamaesyce hirta (L.) Millsp.	Euphorbiaceae	spurge, garden	30	0	1,2, 3, 4	Mi
Chamaesyce hypericifolia (L.) Millsp. (source: PIER)	Euphorbiaceae	beach spurge	37	0	2, 5, 6, 7, 9	Mi, Mj
Chamaesyce hyssopifolia (L.) Small	Euphorbiaceae	spurge, hyssop	46	0	1, 2, 3, 4, 10	Mi, Mj
Commelina diffusa Burm. f.	Commelinaceae	dayflower, spreading	28	0	1, 2, 3	Mj
Cyperus polystachyos Rottb. var. *texensis* (Torr.) Fern.	Cyperaceae	sedge, Texas	20	0	2, 3, 4	Mj
Euphorbia heterophylla L.	Euphorbiaceae	poinsettia, wild	31	0	1, 3, 4,	Mi, Mj
Kyllinga polyphylla Willid. ex Kunth (source: PIER)	Cyperaceae	Navua sedge	25	0	2, 3, 4	Mj
Mimosa pudica L.	Fabaceae	sensitiveplant	20	0	4, 5, 6, 7	Mj
Passiflora foetida L.	Passifloraceae	passionflower, redfruit	15	0	4, 5, 9	Mj
Rottboellia cochinchinensis (Lour.) W.D. Clayton	Poaceae	Itchgrass	13	0	7,	Mi, Mj
Spermacoce remota Lam. (source: PIER)	Rubiaceae	Buttonweed	28	0	2, 3, 4	Mi, Mj

[a] All the weeds except 1, 5, 7, 8, and 11 were also found in the fields and were uninfected. The *n* column refers to the sample size of weeds. Weed scientific names, families, common names, and authorities have been cited from the following sources listed in the Literature Cited: WSSA 2009, USDA 2009, and PIER 2009.

[b] Root gall index 0, no galls or egg masses; 1, 1 or 2 galls or egg masses; 2, 3–10 galls or egg masses; 3, 11–30 galls or egg masses; 4, 31–100 galls or egg masses; 5, more than 100 galls or egg masses.

[c] Source locality of bioassay soil samples: 1, Nausori; 2, Suva-Nasinu; 3, Navua; 4, Sigatoka; 5, Nadi; 6, Lautoka; 7, Ba; 8, Tavua; 9, Rakiraki; 10, Korovou.

[d] Root-knot nematode species parasitizing the tomato, but not the weed, in the pot bioassay of field soil.

The nonhost weed species (Table 3) in bioassay pots did not develop root-knot nematode populations because there were no root galls or egg masses observed, whereas, in the same pots, infections developed on the susceptible tomato host. In the field, some weeds were found uninfected in root-knot nematode–infested fields, and their host status was verified when the same weeds did not have any infection compared with the infected tomato plants in the bioassay pots. The weeds that were uninfected in the field, but which could not be verified via bioassay, have not been reported here because the lack of galling could be due to a patchy distribution of nematodes or to low population density.

The presence or absence of root-galls is usually a reliable method for the diagnosis of root-knot nematode infections; however, in some plants, the galls may not be well developed yet still allow reproduction by root-knot nematodes. In this study, dayflower Benghal, junglerice [*Echinochloa colona* (L.) Link], and goosegrass [*Eleusine indica* (L.) Gaertn.] were found to have very small root galls with egg masses, seen under the stereomicroscope. The presence or lack of galling, therefore, does not necessarily mean that a particular plant is a host or a nonhost because some plants, even in the presence of galls, do not support egg production, and on the other hand, some plants support egg production that can occur without galling (Shepherd 1979) or inconspicuous galling. Therefore, production of eggs by root-knot nematode populations is a more reliable indicator of the host status of a plant.

The host status, and of more importance, the susceptibility level of the weed, depends on the weed species and the *Meloidogyne* species, and sometimes, even the race of the nematode is important (Ehwaeti et al. 1999; Schroeder et al. 1993; Sharma and Rich 2005; Tedford and Fortnum 1988). The weed host, relative to crop hosts, determined on the basis of root galling and egg mass production, can be used to ascertain whether the weed, in comparison to a particular crop, is a better, poorer, or equally good host for the root-knot nematodes. The bioassay tomato plant was found to be a good host, with a root gall and egg-mass index range of 3 to 5, and provided good comparison while determining the weed host status. However, in some cases, probably because of the heavy infection by root-knot nematodes, the tomato plant died, whereas the weeds were able to survive in the bioassay pot. Ehwaeti et al. (1999) reported similar effects, in which the growth of only the susceptible tomato was reduced, whereas the growth of the weeds as nonhost, poor host, and intermediate host were unaffected by southern root-knot nematode. Such information can prove useful because some weeds might have higher tolerance levels than the crop hosts, hence, sustaining the root-knot nematode populations, even when the crop host is dead.

In Fiji, common weed species, such as slender amaranth, balsamapple, tropic ageratum, and ivy gourd, which are often allowed to grow on fallow fields because of alternative uses as vegetables or medicines, can also act as good reservoir hosts of root-knot nematodes (Table 1). Pigweeds (*Amaranthus* spp.) have been consistently reported as good hosts of root-knot nematodes in previous studies as well (Bendixen et al. 1979; Kaur et al. 2007; Khurma et. al 2008). Infected weeds allowed to decompose in the field can act as a source of root-knot nematode inoculum or contribute toward the spread of root-knot nematode infestation to noninfested areas, when infected weed root stock is blown or washed to other farms. Weeds can also act as hosts to more than one species of nematodes. Common weed species, such as slender amaranth and sicklepod, which are good hosts of root-knot nematodes, can also act as hosts of other nematode species (Powell 2001).

The presence of weed hosts can have important implications on the effectiveness of cultural and nonchemical nematode management methods. When using cultural nematode management strategies, weed hosts need to be eliminated from the farms during fallow periods, solarization, or crop rotation to increase the efficacy of the nematode management. Weed hosts along the farm boundaries and in between the furrows should also be removed to ensure that no alternative hosts are available for nematodes targeted for suppression. Weed control is useful in helping to reduce *Meloidogyne* populations (Dabaj and Jenser 1990); hence, nematicide, as well as herbicide, can be used simultaneously for managing root-knot nematode populations.

In subsistence and organic farming, weed hosts can be identified and removed either physically or mechanically. The weed hosts can be incinerated after removal to minimize chances of reinfection and spread to noninfested farms. Weed hosts can still maintain root-knot nematode populations, even when pesticides are used. Thus, management of weed hosts becomes even more important to reduce crop loss from weed-associated pests and diseases. In Fiji, farmers need to be made aware of the role weeds play in nematode management, and this study can form the basis for a more systematic investigation on weeds acting as reservoir hosts of economically important nematodes.

Sources of Materials

[1] Olympus SZ61 stereoscopic microscope, Olympus America, 3500 Corporate Parkway, Center Valley, PA 18034.

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