African Crop Science Conference Proceedings, Vol. 11. pp. 355 - 360
Printed in Uganda. All rights reserved
ISSN 1023-070X/2013 $ 4.00

Potential use of *Momordica balsamina* ethnomedicinal plant as alternative crop in soil with high population densities of *Meloidogyne incognita* race 2

K.M. POFU[1,2] *& P.W. MASHELA*[2]
[1]Agricultural Research Council, Roodeplaat-VOPI, Private Bag X293, Pretoria 0001, South Africa
[2] School of Agricultural and Environmental Sciences, University of Limpopo, Private Bag X1106, Sovenga 0727, South Africa
Corresponding author: wmashela@gmail.com

Abstract Bitter gourd (*Momordica balsamina*) plant, indigenous to Africa, has various ethnomedicinal practices. A greenhouse study was conducted to determine the plant-nematode relation between *M. balsamina* and *Meloidogyne incognita* race 2. Seven treatments, *viz*., 0, 250, 650, 1 050, 1 450, 1 850 and 2 250 eggs and second-stage juveniles (J2s) of *M. incognita* race 2, were arranged in a randomised complete block design, with 12 replications. At 56 days after inoculation, the reproductive factor values at all levels of inoculation were less than one, while nematode infection had no effect on yield components of *M. balsamina*. In plant-parasitic nematology, when RF values are less than unity and nematode infection has no effect on growth of the test plant, the plant is resistant to the test nematode. In conclusion, *M. balsamina* was resistant to *M. incognita* race 2 and could therefore be used as an alternative crop for managing population densities of *M. incognita* race 2.

Key words: Indigenous medicinal plants, nematode resistance, relative penetration index

Introduction

Trade in ethnomedicinal plants in South Africa constitutes a multi-billion industry (over R2.9 billion), which represented at least 5.6% of the national health budget (Dold & Cocks, 2002; Anon., 2005; Mander *et al*., 2005). Consequently, trade in ethnomedicinal plants could be viewed as a potential rural development industry (Moeng & Potgieter, 2011). However, due to the destructive harvesting nature of plants from the wild, certain indigenous ethnomedicinal plants had since become extinct and/or are facing the prospects of extinction (Anon., 2005; Wiersum *et al*., 2006). Attempts to propagate threatened indigenous ethnomedicinal plants are derailed by the existence of virulent root-knot (*Meloidogyne* species) nematodes (Perry *et al*., 2009). In addition to the widest host range, the genus has more than 63 species (De Waele & Elsen, 2007), with the highest number of biological races (Robertson & Diez-Rojo, 2008). For instance, the southern root-knot nematode (*Meloidogyne incognita*), has at least six races (Devran & Sogut, 2011; Hartman & Sasser, 1985; Robertson & Diez-Rojo, 2008), which constitute serious challenges in managing the genus using nematode-resistant genotypes. In South Africa, *M. incognita* race 2 is the most widely distributed race in all agro-systems (Kleynhans *et al*., 1996). Infection by *Meloidogyne* species induces formation of root galls, with various interactive mechanisms resulting in stunted plant growth, decreased water uptake, imbalances of essential nutrient elements, low evapotranspiration and increased root exudation of amino acids (Bird, 1974; Maqbool *et al*., 1987; Mashela, 2002), all of which may negatively affect the quality of medicines derived from the infected plants. The Cucurbitaceae Family, with 118 genera (Pitrat *et al*., 1999), contains several genera which are indigenous to Africa, with potent ethnomedicinal properties. The bitter gourd (*Momordica balsamina* L.) within the family, indigenous to the drier parts of tropical Africa (Gruben & Denton, 2004), has a wide range of potential benefits in ethno- and veterinary-medicines (Hutchings *et al.*, 1996). Leaves, fruits, seeds and bark of *M. balsamina* contain resins, alkaloids, flavonoids, glycosides, steroids, terpenes, cardiac glycoside and saponins (Hutchings *et al.*, 1996). Certain organs of *M. balsamina* were shown to have anti-plasmodial, shigellocidal, anti-diarrheal, anti-septic, anti-bacterial, anti-viral, anti-inflammatory, anti-microbial, hypoglycemic, anti-oxidant, analgesic and hepatoprotective chemical capabilities (Roodt, 1998; Thakur *et al*., 2009).

In Limpopo Province, South Africa, plants of *M. balsamina* are interplanted with crops for the repellence of the greenhouse whitefly (*Trialeurodes vaporariorum* Westwood) (Mashela, 2012, unpublished data). The greenhouse whitefly is currently an economic pest in tomato, cucurbit, lettuce, pepper, potato, strawberry and sweet potato production under both greenhouse and field conditions (Cohen *et al*., 1992; Winter *et al*., 1992; Celix *et al*., 1996; Duffus *et al*., 1996a,b; Wisler *et al*., 1998; Tzanetakis, 2004). This whitefly is a viral vector of the genus *Crinivirus*, which contains viruses that belong to the family Closteroviridae, with linear RNA single-strands, renowned for their unusual capacity to multiply rapidly (Tzanetakis, 2004). The listed viruses reduce the longevity and productivity of various crops (Tzanetakis, 2004). During the use of *M. balsamina* as interplants to repel this insect pest, it was observed that although the tomato roots were highly infected by *M. incognita* race 2, roots of *M. balsamina* had no root galls, with subsequent extractions suggesting that there were no nematodes from roots (Mashela, 2012, unpublished data). The nematode-

resistance of *M. balsamina* could be appealing among smallholder farmers for using this plant species as an alternative crop for ethnomedicinal purposes in areas heavily infested with *Meloidogyne* species, more especially because most of the highly effective fumigant nematicides had been withdrawn from the agro-chemical markets due to their environment-unfriendliness (Speth, 2004). Generally, the absence of galls under field conditions does not imply that the plant is resistant to *Meloidogyne* species since it could purely be due to the spatial distribution of nematodes in that particular field. The objective of this study therefore, was to determine the host-status and host-sensitivity of *M. balsamina* to *M. incognita* race 2 under greenhouse conditions.

Materials and methods

The experiments were conducted concurrently in the greenhouse at the Plant Protection Skills Centre, University of Limpopo, South Africa (23°53'10"S, 29°44'15"E) and at the Agricultural Research Council - Industrial Crops Institute, Rustenburg, North West Province (25°43'40"S, 27°17'30"E). Ambient day/night temperatures averaged 29/22°C, with maximum temperatures controlled using thermostatically-activated fans. The trials were conducted in early summer (October – December) 2012 and repeated in late autumn (January-March) 2013.

Ripe fruit were harvested from locally-raised *M. balsamina* plants, cut into pieces, seeds removed and shade-dried for six days. Seeds were sown in seedling trays containing Hygromix growing medium (Hygrotech, Pretoria North, South Africa). Twenty-cm-diameter plastic pots, filled with 2 700 ml steam-pasteurised (300ºC for 1 h) river sand and Hygromix at 3:1 (v/v) ratio, were placed on greenhouse benches at 0.3 m inter-row and 0.3 m intra-row spacing, with 5 g 2:3:2 (22) NPK fertiliser mixed into the topsoil of each pot. Uniform four-week-old, nematode-free *M. balsamina* seedlings were transplanted to the pots one day after irrigating the growing medium to field capacity. When required, *M. incognita* race 2 inocula were prepared by extracting eggs and second-stage juveniles (J2s) from roots of greenhouse-grown nematode-susceptible kenaf (*Hibiscus cannabinus* L.) plants in 1% NaOCl solution (Hussey & Barker, 1973).

Seven treatments, *viz*. 0, 250, 650, 1 050, 1 450, 1 850 and 2 250 eggs and J2s, were arranged in a randomised complete block design, with six replicates. Two days after transplanting, pots were each infested by dispensing approximate numbers of *M. incognita* race 2 eggs and J2s using a 20-ml plastic syringe by placing into 5-cm-deep holes on the cardinal points of the stem of the plants per replication. The zero untreated control plants received filtrate (25-ìm-mesh sieve) of nematode suspension to establish any microbes associated with *Meloidogyne* species in their rhizosphere. Two sets of Hadeco moisture meter were inserted to 20-cm depths in randomly selected pots of each treatment to monitor soil moisture tension. Plants were irrigated with 500 ml tapwater as soon as 50% of the moisture meters have readings just below 2 units.

At 56 days after inoculation, vine length was measured from the terminal end of the short stem to the tips of the vines per plant. Stems were cut at the soil level, shoots were oven-dried for 72 h at 70°C and weighed. Root systems were removed from pots, immersed in water to remove soil particles, blotted dry and weighed to facilitate the calculation of nematode density per total roots per plant. Root galls, when necessary, were assessed using the North Carolina Differential Scale of 0 = no galls, 1 = 1 - 2 galls, 2 = 3 – 11 galls, 3 = 11 – 30 galls, 4 = 31 - 100 galls and 5 = > 100 galls (Taylor & Sasser, 1978). All collected roots were separately weighed. Nematodes were extracted from total roots per plant by maceration and blending for 30 seconds in 1% NaOCl (Hussey & Barker, 1973) and passed through top-down nested 150-ìm and 25-ìm mesh sieves. Contents of the 25-ìm mesh sieve were poured into 100-ml plastic containers for counting under a stereomicroscope. Soil per pot was thoroughly mixed and a 250-ml soil sample was collected. Nematodes were extracted from soil subsamples using the modified sugar-floatation and centrifugation method (Coolen & D'Herde, 1972). Nematode numbers from roots were converted to nematodes per total root system per plant, whereas soil nematode numbers were converted to 2700 ml soil per pot. The reproductive factor (RF = Pf/Pi), described as final population density of nematodes (Pf)/initial population density of nematodes (Pi), were computed.

Data were subjected to analysis of variance (ANOVA) through the SAS software (SAS Institute, Inc., Cary, NC., U.S.A.) to determine the effects of Pi on the RF values and the yield components. Mean separation for significant (P d" 0.05) treatments was achieved through the Duncan's multiple-range test. Lines of the best fit were determined for RF values over the $\log_{10}(Pi + 1)$. Unless otherwise stated, only treatments that were significant at the probability level of 5% were discussed.

Results and discussion

Treatment had significant effect on RF values, contributing 69% to total treatment variation in RF values (data not shown). However, treatment effects were not significant for all plant variables. At all levels of inoculation, RF and RPI values were each less than one (Table 1). Generally, RF values provide an indication of whether a plant is a host or a non-host to the test nematode. The RF values of less than one suggest that the nematode failed to feed and then reproduce in a given plant species (Seinhorst, 1967; Windham & Williams, 1988). Observations in *M. balsamina* confirmed those in other indigenous ethnomedicinal plants, namely, wild watermelon (*Cucumis africanus*) and wild cucumber (*C. myriocarpus*), which were also highly resistant to *M. incognita* race 2 (Pofu *et al*., 2009; Pofu *et al*., 2010a,b). Incidentally, the two *Cucumis* species are also in the Cucurbitaceae Family. Suppression of population densities of *M. incognita* race 2 by *M. balsamina* support *in vitro* observations that extracts from the plant were nematicidal to a free-living nematode, *Caenorhabditis elegans* (Beloin *et al.*, 2005)

and that they were also effective in controlling gastrointestinal nematodes in cattle (Amin *et al.*, 2009).

The relationship between the RF values and $\log_{10}$ (Pi + 1) of *M. incognita* race 2 on *M. balsamina* was quadratic – with a downward facing slope (Fig. 1). The observed quadratic relationship suggested that density-dependent growth (DDG) principles were in place (Salisbury & Ross, 1992; Pofu & Mashela, 2013). The DDG principles are depicted through three responses to abiotic and biotic factors, namely, stimulation, neutral and inhibition (Liu *et al*., 2003). In the current study, all inoculation levels were already beyond the neutral or equilibrium (E) point for *M. incognita* race 2 in *M. Balsamina*, which suggested a low E point relative to the used inoculum levels. This could also be viewed to imply that resistance of *M. balsamina* to *M. incognita* race 2 was quite strong.

In the current study, the aggregated relative penetration index (RPI) was less than one. Pofu & Mashela (2011) developed the RPI to estimate whether resistance was pre- or post-infectional. Generally, in plant-parasitic nematodes forms of resistance are either pre- or post-infectional (Ibraham *et al*., 1980; Steele & Savitsky, 1981; Weischer, 1982; Acedo *et al*., 1984; Huang, 1986; Raja & Dasgupta, 1986; Kaplan & Davis, 1987). In pre-infectional resistance nematicidal active nematicidal chemicals are released into the rhizosphere, with nematode population densities reduced in the soil (Griffin & Waite, 1971; Richard & DuPree, 1978; Motsinger *et al*., 2007; Wang *et al*., 2007). In contrast, in post-infectional resistance, nematodes penetrate the roots, resulting in the activation of passive chemicals – the phytoalexins (Harborne, 1999), which could constitute time-linked resistance (Thurau *et al*., 2010) or time-unlinked resistance (Wallace, 1973). In our study, the aggregated RPI at all levels of inoculation was less than one, suggesting that the form of resistance was pre-infectional (Pofu & Mashela, 2011). Unlike post-infectional resistance, pre-infectional resistance is not introgressible (Kaplan & Keen, 1988).

Host-sensitivity describes the responses of hosts to nematode infection (Seinhorst, 1967). Generally, when the RF value is less than unity and there is no yield loss, the test plant is said to be resistant (Seinhorst, 1967). Using the Seinhorst model, *M. balsamina* seedlings were therefore, resistant to *M. incognita* race 2. In conclusion, *M. balsamina* is resistant to *M. incognita* race 2, which is widely distributed in various agricultural systems in South

Table 1. Population density (Pf), reproductive factor (RF) and relative penetration index (RPI) of *Meloidogyne incognita* race 2 on *Momordica balsamina* at 56 days after inoculation (n = 72).

Inoculation level (Pi)	Nematode population density (Pf)			Proportion	
	Pf_{soil}	Pf_{root}	Pf_{total}	RF	RPI
250	12	20	32	0.13a	0.67
650	17	23	40	0.06ab	0.35
1 050	33	25	58	0.06ab	-0.24
1 450	53	23	77	0.05ab	-0.57
1 850	12	17	28	0.02b	0.42
2 250	43	20	63	0.03ab	-0.53

RF = Pf_{total}/Pi and RPI = (Pf_{root}/Pf_{soil}) – 1.

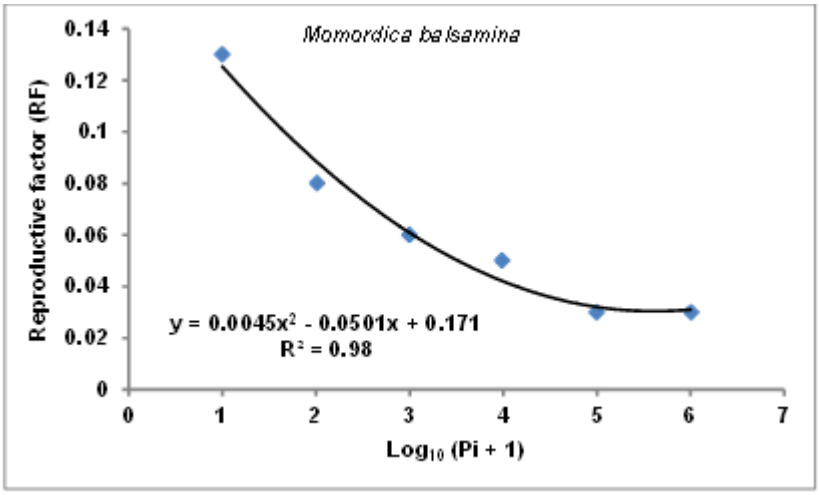

Figure 1. Relationship between the reproductive factor (RF) values and log-transformed initial population density of *Meloidogyne incognita* race 2 on *Momordica balsamina* at 56 days after inoculation (n = 72).

Africa. The observation is important in traditional medicine since it accords *M. balsamina* the status for use as an alternative crop in areas with high population densities of *M. incognita* race 2. Additionally, the observation expands the number of indigenous ethnomedicinal plants in the Cucurbitaceae Family, which could be of use as alternative crops for managing population densities of *Meloidogyne* species for various potential uses.

Acknowledgement

The authors are grateful to the Agricultural Research Council of South Africa, the Land Bank Chair of Agriculture – University of Limpopo and Flemish Interuniversity Council of Belgium for financial support.

References

Acedo, J.R., Dropkin, V.H. & Luedders, V.D. 1984. Nematode population attrition and histopathology of *Heterodera glycines*–soybean associations. Journal of Nematology **16,** 48-57.

Amin, M.R., Mostofa, M., Hoque, M.E. & Sayed, M.A. 2009. In vitro efficacy of some indigenous ethnomedicinal plants against gastrointestinal nematodes of cattle. Journal of the Bangladesh Agricultural University **7,** 57-61.

Anonymous, 2005. Ethnomedicinal plants trade. DWAF (Department of Water Affairs and Forestry), Forestry Sub-sector Studies-Briefing 5, Pretoria: DFID.

Beloin, N., Gbeassor, M., Akpagana, K., Hudson, J., De Soussa, K., Koumaglo, K. & Arnason, J.T. 2005. Ethnoethnomedicinal uses of *Momordica charantia* (Cucurbitaceae) in Togo and relation to its phytochemistry and biological activity. Journal of Ethnopharmacology **96,** 49-55.

Bird, A.F. 1974. Plant response to root-knot nematodes. Annual Review of Phytopathology **12,** 69-85.

Celix, A., Lopez-Sese, A., Almarza, N., Gomez-Guillamon, M. & Rodriguez-Cerezo, E. 1996. Characterization of cucurbit yellow stunting disorders virus, a *Bemisia tabaci*-transmitted Closterovirus. Journal of Phytopathology **86,** 1370-1376.

Cohen, J., Frank, A., Vetten, H.J., Lesemann, D.E. & Loebenstein, G. 1992. Closterovirus-like particles associated with a whitefly-transmitted disease of sweet potato. Annals of Applied Biology **121,** 257-268.

Coolen, W.A. & D'Herde, C.J. 1972. A rapid method for the quantitative extraction of nematodes from plant tissue. Minister of Agricultural Research and Administration. Station of Agricultural Research Centre Belgium. p.77.

Devran, Z. & Sogut, M.A. 2011. Characterising races of *Meloidogyne incognita*, *Meloidogyne javanica* and *Meloidogyne arenaria* in the West Mediterranean region of Turkey. Crop Protection **30,** 451-455.

De Waele, D. and Elsen, A. 2007. Challenges in tropical plant nematology. Annual Review Phytopathology **45,** 457-485.

Dold, A.P. & Cocks, M.L. 2002. The trade in ethnomedicinal plants in the Eastern Cape Province, South Africa. South African Journal of Science 98:589-597.

Duffus, J.E., Liu, H.Y. & Wisler, G.C. 1996a. Tomato infectious chlorosis virus: A new clostero-like virus transmitted by *Trialeurodes vaporariorum.* European Journal of Plant Pathology **102,** 219-226.

Duffus, J.E., Liu, H.Y., Wisler, G.C. & Li, R.H. 1996b. Lettuce chlorosis virus: A new whitefly-transmitted Closterovirus. European Journal of Plant Pathology **102,** 591-596.

Griffin, G.D. & Waite, W.W. 1971. Attraction of *Ditylenchulus dipsaci* and *Meloidogyne hapla* by resistant and susceptible alfalfa seedlings. Journal of Nematology **3,** 215.

Grubben, G.J.H. & Denton, O.A. 2004. Plant resources of tropical Africa 2. Vegetables. Wageningen, Netherlands: PROTA Foundation, Backhuys Publishers. p.385.

Harborne, J.B. 1999. The comparative biochemistry of phytoalexin induction in plants. Biochemistry System of Ecology **27,** 335-367.

Hartman, K.M. & Sasser, J.N. 1985. Identification of *Meloidogyne* species on the basis of differential host test and perineal pattern morphology. In: Barker, K.R., Carter, C.C. & Sasser, J.N. (Eds.). An advance treatise on *Meloidogyne,* Raleigh, USA: Carolina State University. pp. 69-77.

Huang, S.P. 1986. Penetration, development, reproduction, and sex ratio of *Meloidogyne incognita race 2* in three carrot cultivars. Journal of Nematology **18,** 408-412.

Hussey, R.S. & Barker, K.R. 1973. A comparison of methods of collecting inocula of *Meloidogyne* species, including a new technique. Plant Disease Report **42,** 865-872.

Hutchings, A., Scott, A.H., Lewis, G. & Cunningham, A.B. 1996. Zulu ethnomedicinal plants, an inventory. Pietermaritzburg: University of Natal Press.

Ibraham, I.K.A., Khalil, H.A.A. & Rezk, M.A. 1980. Development and pathogenesis of *Meloidogyne incognita* race 2 in cotton roots. Nematologia Mediterranea **8,** 29-33.

Kaplan, D.T. & Davis, E.L. 1987. Mechanisms of plant incompatibility with nematodes. In: Veech, J.A. & Dickson, D.W. (Eds.). Vistas on nematology Hyattsville, Maryland: Society of Nematologists. pp. 267-276.

Kaplan, D.T. & Keen, N.T. 1980. Mechanisms conferring plant incompatibility to nematodes. Revue de Nèmatologie **3,** 123-134.

Kleynhans, K.P.N., Van der Berg, E., Swart, A., Marais, M. & Buckley, N.H. 1996. Plant nematodes in South Africa. Handbook No. 8, Pretoria: Plant Protection Research Institute. p.165.

Lamberti, F. 1979b. Chemical and cultural control. In: Lamberti, F. & Taylor, C.E. (Eds.). Root-knot nematodes (*Meloidogyne* species) systematics, biology and control. London: Academic Press. pp. 403-423.

Mander, M., Ntuli, L., Diederichs, N. & Mavundla, K. 2005. Economics of the traditional medicine in South Africa. In: Diederichs, N. (Ed.). Commercialising ethnomedicinal plants: A Southern African guide. Stellenbosch: African SunMedia. p. 216.

Mafeo, T.P. & Mashela, P.W. 2009. Responses of germination in tomato, watermelon and butternut

squash to a C*ucumis* bio-nematicide. Journal of Agricultural and Environmental Sciences **6,** 215-219.

Maqbool, A., Aurangzeb, R.A., Habib, N.B. & Khan, I. 1987. Nutritional and organoleptic evaluation of wheat rotis supplemented with soybean flour. Pakistan Journal of Science and Industry Research **38,** 615-618.

Mashela, P.W. 2002. Ground wild cucumber fruits suppress numbers of *Meloidogyne incognita* on tomato in microplots. Nematropica **32,** 13-19.

Moeng, E.T. & Potgieter, M.J. 2011. The trade of ethnomedicinal plants by muthi shops and street vendors in the Limpopo Province, South Africa. Journal of Ethnomedicinal Plants and Research **5,** 558-564.

Mofokeng, M.A. 2005. Host-status of *Cucumis myriocarpus* to *Meloidogyne. incognita*. Master Mini-Dissertation submitted to the University of Limpopo, Sovenga, South Africa.

Mofokeng, M.A., Mokgalong, N.H. & Mashela, P.W. 2005. Host-status of wild to root-knot nematode. Proceedings of the Nematological Society of Southern Africa **17,** 12.

Motsinger, R.E., Moody, E.H. & Gay, C.M. 1977. Reaction of certain French marigolds (*Tagetes patula*) cultivars to three *Meloidogyne* species. Journal of Nematology **9,** 278.

Perry, R., Moens, M. & Starr, J.L. 2009. Root-knot nematodes. Wallingford: CABI.

Pitrat, M., Chauvet, M. & Foury, C. 1999. Diversity, history and production of cultivated cucurbits. Acta Horticulturae 492:21-28.

Pofu, K.M., Mafeo, T.P. & Mashela, P.W. 2009. The responses of *Cucumis africanus* to *Meloidogyne incognita* race 2 infection. African Crop Science Proceedings **9,** 659-661.

Pofu, K.M., Mashela, P.W. & Mokgalong, N.M. 2010a. Host-status and host-sensitivity of *Cucumis africanus* and *Cucumis myriocarpus* to M*eloidogyne incognita* race 2 under greenhouse conditions. African Journal of Agricultural Research **5,** 1504-1508.

Pofu, K.M., Mashela, P.W. & Mphosi, M.S. 2010b. Responses of *Cucumis africanus* and *Cucumis myriocarpus* to *Meloidogyne incognita* race 2 under microplot conditions. International Journal of Nematology **20,** 113-118.

Pofu, K.M. & Mashela, P.W. 2011. Using relative penetration and maleness indices in *Meloidogyne incognita* to establish resistance type in *Cucumis myriocarpus*. African Journal of Biotechnology **10,** 390-393.

Pofu, K.M. 2011. Potential uses of *Cucumis* species as nematode-resistant rootstocks in watermelon (C*itrulus lanatus*) production. Thesis submitted to the University of Limpopo, Sovenga, South Africa.

Pofu, K.M. & Mashela, P.W. 2013. Using population dynamics of *Meloidogyne javanica* on fibre hemp cultivars to standardise exposure time in host-status trials. Journal of American and Experimental Science. (In Press).

Raja, A. & Dasgupta, D.R. 1986. Enhanced synthesis of messenger RNA in relation to resistance expression in cowpea (*Vigna unguiculata*) infected with the root-knot nematode, *Meloidogyne incognita.* Journal of Nematology **9,** 35-49.

Richard, D.A. & Dupree Jr, A.W. 1978. The effectiveness of ten kinds of marigolds and five other treatments for control of four *Meloidogyne* species. Journal of Nematology **4,** 296-297.

Robertson, L. & Diez-Rojo, M.A. 2008. New host races of *Meloidogyne arenaria*, *Meloidogyne incognita*, and *Meloidogyne javanica* from horticultural regions of Spain. Plant Disease **93,** 180-184.

Roodt, V. 1998. Common wild flowers of the Okavango Delta; ethnomedicinal uses and nutritional value. The Shell Field Guide Series: Part II.

Salisbury, F.B. & Ross, C.W. 1992. Plant physiology. Belmont California: Wadsworth.

Seinhorst, 1967. The relationship between population increase and population density in plant-parasitic nematodes. 3. Definition of the terms host, host-status and resistance. 4. The influence of external conditions on the regulation of population density. Nematologica **13,** 429-442.

Speth, J.G. 2004. Red sky at Morning: America and the Crisis of the Global Environment. New Haven: Yale University Press.

Steele, A.E. & Savitsky, H. 1981. Resistance of trisomic and diploid hybrids of *Beta vulgaris B. procumbens* to the sugar beet nematode, *Heterodera schatii*. Journal of Nematology **13,** 352-357.

Thakur, G.S., Bag, M., Sanodiya, B.S., Bhadouriya, P., Debnath, M., Prasad, G.B. & Bisen, P.S. 2009. *Momordica balsamina*: A ethnomedicinal and neutraceutical plant for health care management. Current Pharmacology and Biotechnology **10,** 667-682.

Taylor, A.L. & Sasser, J.N. 1978. Biology, identification and control of root-knot nematodes *(Meliodogyne* species*)*. Raleigh, North Caroline, USA, North Carolina State University Press. p. 111

Thies, J.A. & Levi, A. 2007. Characterization of watermelon (*Citrullus lanatus* var. Citroides) germplasm for resistance to root-knot nematodes. HortScience **42,** 1530-1533.

Thurau, T., Ye, W. & Cai, D. 2010. Insect and nematode resistance. In: Widholm, J.M. & Nagata, T. (Eds.). Biotechnology in agriculture and forestry, Heidelberg, Berlin: Springer-Verlag. 675 pp.

Tzanetakis, I.E. 2004. Molecular characterisation of criniviruses and ilarviruses infecting strawberry. Ph.D. Dissertation Submitted at Oregon State University, Corvallis, USA.

Wang, K., Hooks, C.R. & Ploeg, A. 2007. Protecting crops from nematode pests using marigold as an alternative to chemical nematicides. Co-operative Extension Services, Report. pp. 1-6.

Wallace, H.R. 1973. Nematode ecology and plant disease. London, U.K.: Edward Arnold. 228 pp.

Weischer, B. 1982. Histopathological studies on grape species with different degrees of resistance to *Xiphenema index*. Nematologica **28,** 178-179.

Wiersum, K.F., Dold, A.P., Husselman, M. & Cocks, M. 2006. Cultivation of ethnomedicinal plants as a tool for biodiversity conservation and poverty alleviation

in the Amatola region, In: Ethnomedicinal and aromatic plants. Bogers, R.J., Cracker, L.E. & Lange, D. (Eds.). South Africa: Springer. pp. 43-57.

Windham, G.L. & Williams, W.P. 1988. Reproduction of *M. javanica* on corn hybrids and inbreeds. Annals of Applied Nematology **2,** 25-28.

Winter, S., Purac, A., Leggett, F., Frison, E.A., Rossel, H.W. & Hamilton, R.I. 1992. Partial characterization and molecular cloning of a closterovirus from sweet potato infected with the sweet potato virus disease complex from Nigeria. Journal of Phytopathology **82,** 869-875.

Wisler, G.C., Duffus, J.E., Liu, H.Y. & Li, R.H. 1998. Ecology and epidemiology of whitefly-transmitted closteroviruses. Pant Disease **82,** 270-280.