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Plant Roots Increase Bacterivorous Nematode Dispersion through Nonuniform Glass-bead Media

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Abstract: Dispersion of bacterivorous nematodes in soil is a crucial ecological process that permits settlement and exploitation of new bacterial-rich patches. Although plant roots, by modifying soil structure, are likely to influence this process, they have so far been neglected. In this study, using an original three-compartment microcosm experimental design and polyvinyl chloride (PVC) bars to mimic plant roots, we tested the ability of roots to improve the dispersion of bacterivorous nematode populations through two wet, nonuniform granular (glass bead) media imitating contrasting soil textures. We showed that artificial roots increased migration time of bacterivorous nematode populations in the small-bead medium, suggesting that plant roots may play an important role in nematode dispersion in fine-textured soils or when soil compaction is high.
Key words: dispersion, ecology, glass-bead media, migration time, colonization time, plant roots.

Bacterivorous nematodes are widely distributed soil organisms involved in key terrestrial ecosystem functions such as soil fertility and plant productivity (Anderson et al., 1978; Ferris et al., 1998; Djigal et al., 2004; Blanc et al., 2006; Bonkowski et al., 2009; Irshad et al., 2011). By releasing nutrients (nitrogen and phosphorus) immobilized in bacterial biomass in the vicinity of plant roots, they largely contribute to soil nutrient availability (Anderson et al., 1983; Ferris et al., 1998) and plant nutrition and growth (Bonkowski and Clarholm, 2012; Irshad et al., 2012; Trap et al., 2015).

The positive effects of bacterivorous nematodes on soil and plant functions are conditioned by their ability to move within heterogeneous soils (Griffiths and Caul, 1993). Dispersion of nematodes from one bacterial site to new resource patches is a crucial ecological process facilitating ecosystem functions (Hassink et al., 1993a; Savin et al., 2001; Rodger et al., 2004; Horiuchi et al., 2005). It is strongly determined by soil conditions such as bulk density (Portillo-Aguilar et al., 1999; Hunt et al., 2001), soil water content (Young et al., 1998) or temperature (Hunt et al., 2001), soil texture and hence porosity (Prot and Van Gundy, 1981; Georgis and Poinar, 1983; Young et al., 1998; Portillo-Aguilar et al., 1999), bacterial species (Young et al., 1998; Rodger et al., 2004), salt gradients (Le Saux and Queneherve, 2002), or soil water runoff (Chabrier et al., 2009).

In most experiments, bacteria–nematode effects on soil nutrient availability have been studied in bulk soils and root exudates were mimicked by providing carbon as an energy source for bacteria, usually as glucose (Cole et al., 1978; Coleman et al., 1978; Anderson et al., 1983; Ferris et al., 1997, 1998). Possible physical influences of roots on nematode dispersal, and the subsequent effects on soil nutrient availability, have thus not been represented. Moreover, in experiments with plants (Djigal et al., 2004; Bjornlund et al., 2012), shifts in both energy supply and porosity induced by roots are confounded, limiting our ability to decipher mechanisms by which roots impact nematode-driven ecological functions. In this study, using an original three-compartment microcosm experimental design, we tested the ability of roots to improve the dispersion of nematodes and their associated bacteria through two wet granular media made from glass beads of different sizes in order to mimic two contrasting soil textures.

Materials and Methods

The study was conducted in sterile three-compartment 90-mm petri dishes (compartments labeled A–C). We designed six treatments (Fig. 1). The first two treatments corresponded to negative (NC) and positive (PC) controls, respectively. In NC, compartments were not connected (compartments were independent), whereas in PC and for the four other treatments, short gates (~5 mm width) were opened between compartments A and B and between compartments B and C by melting the plastic walls separating compartments (Fig. 1). In all treatments, compartments A and C were filled with 10 ml tryptic soy broth (TSB)-A (3 g/liter TSB Fluka 22092 and 1% agar w/v supplemented with cholesterol 5 mg/liter). The compartment B was filled with 10 ml TSB-A in NC and PC treatments, whereas in the other four treatments, it was filled with 15 g of nonuniform (polydisperse) glass beads (Abralis, France), either of small size (SB: mean diameter 130 μm, minimum-maximum diameters of 60–260 μm, porosity 40%) or large size (LB: mean diameter 600 μm, minimum-maximum diameters of 300–1100 μm, porosity 32%). Bead size was measured using a laser granulometer (Mastersizer APA2000, Malvern Instruments Ltd., Malvern, United Kingdom) while the distribution of pore size was approximated using the Finney model (Finney,

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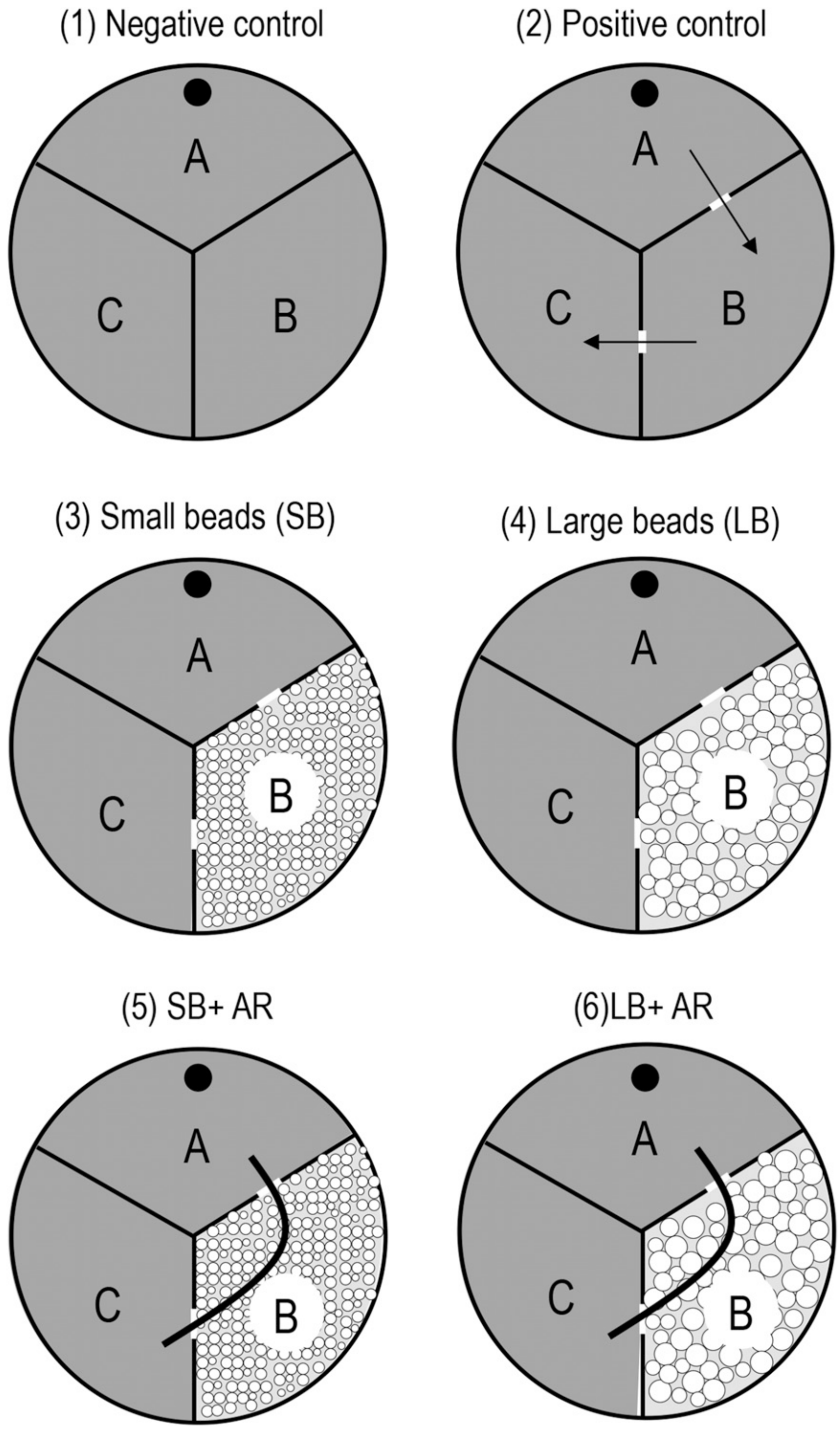

FIG. 1. Experimental setup with three-compartment petri dishes used to assess the effect of roots and medium porosity on nematode dispersal and colonization. In all treatments but the negative control (treatment 1), compartments A and C were connected by gates opened through the wall, with or without an artificial root "AR" (2-mm-diameter PVC bar) added to cross the compartment B. Compartments A and C were filled with TSB-A (see text for composition). Depending on the treatment, B was filled with TSB-A (treatments 1 and 2) or with small beads (SB) (treatments 3 and 5), or large beads (LB) (treatments 4 and 6) in sterile deionized water. Only compartment A was inoculated with *Bacillus subtilis* and 15 bacterial-feeding adult nematodes belonging to the *Rhabditis* sp., as a spot dropped from the corner of the compartment at the center of the petri dish (closed circle).

1970; Frost, 1978) for uniformly sized (monodisperse) granular media with the average size of beads as the most representative bead size for each medium.

Before use, glass beads were acid-washed using HCl 1M, rinsed with sterile deionized water, and saturated at 100% of their holding capacity. In two of the glass-bead treatments (5 and 6), a flexible PVC bar (2 mm diameter, 4 cm length), previously sterilized in bleach and washed with sterilized deionized water, was used to mimic roots and placed in compartment B (Fig. 1). The ends of the PVC bar were inserted through the gates, thus linking compartment A with C (Fig. 1). In compartment B, the PVC bar was placed inside the bead medium. This PVC bar was used to test physical effects of roots on nematode dispersion without interfering with carbon supply by rhizodeposition. Each treatment was replicated five times (30 microcosms).

For all microcosms, compartment A was inoculated with 100 μl of fresh gram-positive *Bacillus subtilis* (strain 111b) culture and 15 adult bacterial-feeding nematodes belonging to *Rhabditis* sp., together as a spot dropped from the corner of the compartment at the center of the petri dish (Fig. 1). Bacteria and nematodes for experiments were isolated from an ectomycorrhizal root tip and the soil collected in a maritime pine forest, respectively (Irshad et al., 2011). Nematodes were maintained in our laboratory by transferring individuals onto new TSB-A plates containing *B. subtilis* (Irshad et al., 2011). Nematodes multiplied in the dark at 20°C. Nematodes used in the inoculation experiments were prepared by removing them from the breeding TSB-A plates by washing the surface with a sterile NaCl solution (1%). They were washed from most *B. subtilis* by centrifugation (1,000 rpm, 5 min) and resuspended in sterile deionized water.

Every morning for 3 wk, microcosms were carefully inspected using a binocular microscope and the number of individuals in compartment C was counted. We defined the "migration time" as the number of days required to observe one individual (juvenile or adult) in compartment C. We also assessed the "colonization time" as the number of days required for nematodes to exploit the whole compartment C and reach the maximal carrying capacity set at 400 individuals (corresponding to a homogeneous distribution of nematodes in the compartment). Means and standard deviation were calculated for each treatment and significant differences were tested using one-way analysis of variance and Tukey honest significance test tests. Normality of residuals was checked using Shapiro test. All tests were computed with the R freeware (R, 2008) and statistical significance was set at $P < 0.05$.

Results and Discussion

After 3 wk of incubation, no nematode was observed in compartment C in the NC (Fig. 2A), confirming that nematodes were not able to cross the walls separating compartments. In the PC, around 8 days were required to observe individuals in C, whereas 10 days were required in both large- and small-bead treatments. Our findings are in agreement with those obtained by Wallace (1958) that showed that the pore size in a saturated 75- to 150-μm soil fraction (similar to our small-bead medium) approaches that through which *Heterodera schachtii* larvae are unable to pass. In his study, a maximum of 10% of the nematodes migrated farther than 5 cm from the inoculation site for this soil fraction, whereas ~35% of the population migrated farther than 5 cm in the 150- to 200-μm fraction.

When an artificial root was added across compartment B, the mean migration time decreased to 9 days and 8 days for large and small beads, respectively. The effect of the artificial root on nematode migration time was thus observed for both bead sizes, but the effect was significant for small beads only. By creating macropores

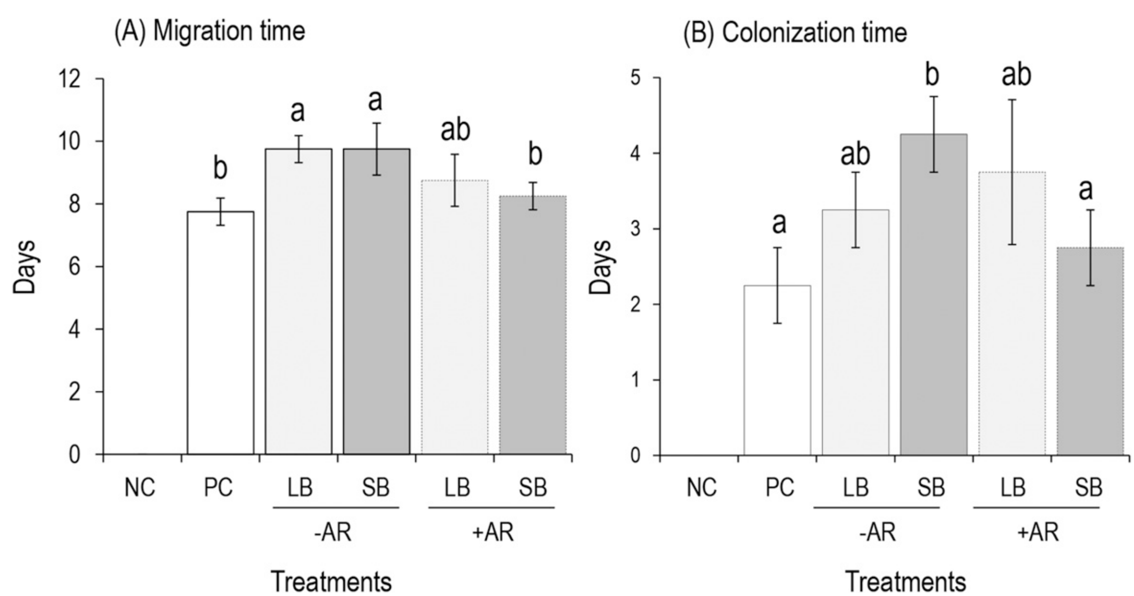

Fig. 2. A. Migration time and B. colonization time in days according to treatments. NC: negative control; PC: positive control (white); LB: large beads (light gray); SB: small beads (dark gray); −AR: without artificial root (solid line); +AR: with artificial root (dotted line). Different letters (a and b) indicate significance among treatments according to one-way analysis of variance and Tukey honest significance test post hoc tests ($P < 0.05$, $n = 5$).

(Angers and Caron, 1998), roots increased soil porosity for these free organisms and their dispersal rate. Nematodes can also move in the water film formed around the root as a "highway" toward a new site. Here, we did not provide glucose to mimic root exudates because our aim was to discriminate energy supply from physical effects of roots on nematodes. In natural rhizospheres, the presence of root exudates is known to improve soil structure and increase aggregation, especially in clay soils (Angers and Caron, 1998; Bertin et al., 2003). It is possible that in natural conditions, the improving effect of roots on nematode dispersion could be modified by rhizodeposition rate and soil clay content (Hassink et al., 1993a).

Interestingly, the colonization time of nematode populations growing in C varied according to treatments (Fig. 2B). The lowest values were observed for PC and for small beads with an artificial root (mean ~2.5 days of colonization time), whereas the highest values were observed for small beads without an artificial root (mean ~4.2 days). Intermediate values were found in large beads, with or without artificial roots. In PC and beads with an artificial root, adults moved easily from A to C. In contrast, for the treatment with small beads and without AR, the first individuals observed in C were juveniles. This pattern can be explained by the diameter of adult nematodes after 14 days of growth oscillating around 35 μm (n = 30). Individuals with a diameter superior to ~30 μm were highly constrained by the beads (Fig. 3). In consequence, in treatments with small beads, only juveniles could move easily from A to C. Several hours and days were thus needed for juveniles to grow in C before becoming adults and reproducing, explaining why colonization was slower.

It is important to note that we did not inoculate compartments B and C with *B. subtilis* cells. The colonization time of nematode populations was thus based on their ability to transport bacteria (or spores) from compartment A to C. Several studies observed phoretic transport of bacteria by nematodes (Hallmann et al., 1998; Knox et al., 2003, 2004) or defecation of living bacterial cells or spores after their passage through the nematode gut (Laaberki and Dworkin, 2008; Rae et al., 2012). For instance, Laaberki and Dworkin (2008) showed that ingested *B. subtilis* spores were resistant to *Caenorhabditis elegans* digestion. Some studies showed that nematodes can act as vectors of rhizobium (Jatala et al., 1974; Sitaramaiah and Singh, 1975; Horiuchi et al., 2005) or plant pathogenic bacteria (Kroupitski et al., 2015). Once in C, living *B. subtilis* cells or spores attached on nematode cuticles or excreted by nematodes can proliferate rapidly on TSB-A before nematode population growth.

In conclusion, this microcosm experiment showed that the presence of small beads severely constrained adult but not juvenile dispersion. An artificial root increased bacterivorous nematode populations and associated bacterial food dispersion in wet polydisperse media, especially in small-bead media. These results suggested that plant roots can play an important role in assisting nematode dispersion in fine-textured soils or when roots penetrate in compacted soils (Iijima et al., 1991; Queneherve and Chotte, 1996). Nematode effects on nutrient cycling are known to vary according to soil texture (Hassink et al., 1993b), but our study suggests that the presence of roots may alleviate the effect of small soil pore size, enhancing local population connection and probably soil nutrient cycling (Clarholm, 1985). Our results also suggested that besides root exudates and active attraction, differences in root architecture among plant species can also explain why nematode population abundance or biomass in plant rhizospheres vary according to plant species (Griffiths, 1990; Horiuchi et al., 2005). Further studies using

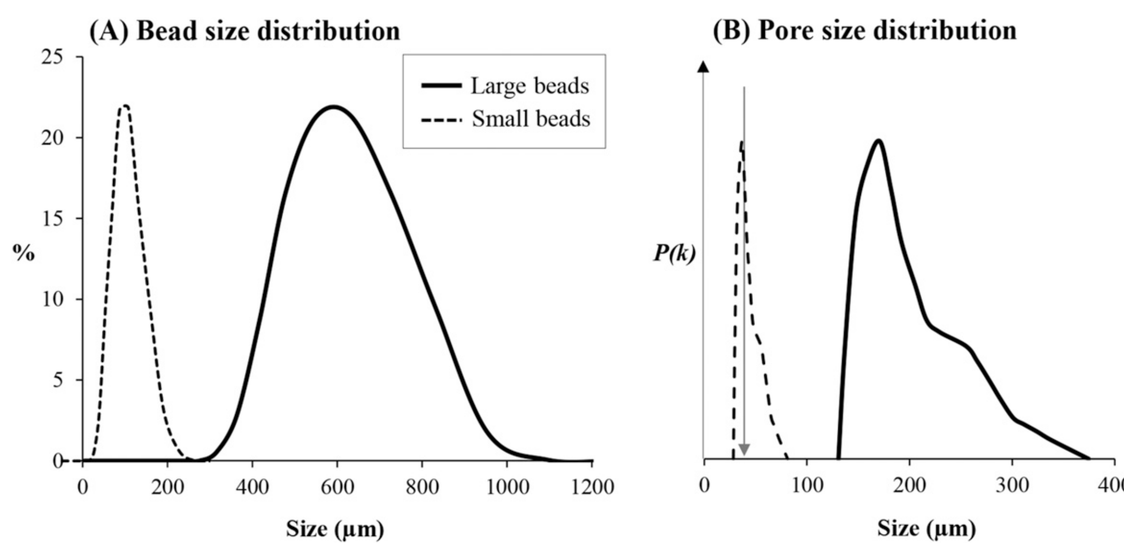

Fig. 3. A. Size distribution of beads and B. approximation of pore size distribution for small (dotted line) or large (solid line) beads. $P(k)$ is the frequency of the radius (k) of pores. The blue solid line indicates mean nematode diameter size of adults (n = 30).

similar designs could be used to disentangle physical and nutritional impacts of roots on nematode-driven transport of nutrients or organic compounds such as enzymes or pollutants.

Literature Cited

Anderson, R., Gould, W., Woods, L., Cambardella, C., Ingham, R., and Coleman, D. 1983. Organic and inorganic nitrogenous losses by microbivorous nematodes in soil. Oikos 40:75–80.

Anderson, R. V., Elliott, E. T., McClellan, J. F., Coleman, D. C., Cole, C. V., and Hunt, H. W. 1978. Trophic interactions in soils as they affect energy and nutrient dynamics. 3. Biotic interactions of bacteria, amoebae, and nematodes. Microbial Ecology 4:361–371.

Angers, D. A., and Caron, J. 1998. Plant-induced changes in soil structure: Processes and feedbacks. Biogeochemistry 42:55–72.

Bertin, C., Yang, X. H., and Weston, L. A. 2003. The role of root exudates and allelochemicals in the rhizosphere. Plant and Soil 256:67–83.

Bjornlund, L., Liu, M. Q., Rønn, R., Christensen, S., and Ekelund, F. 2012. Nematodes and protozoa affect plants differently, depending on soil nutrient status. European Journal of Soil Biology 50:28–31.

Blanc, C., Sy, M., Djigal, D., Brauman, A., Normand, P., and Villenave, C. 2006. Nutrition on bacteria by bacterial-feeding nematodes and consequences on the structure of soil bacterial community. European Journal of Soil Biology 42:S70–S78.

Bonkowski, M., and Clarholm, M. 2012. Stimulation of plant growth through interactions of bacteria and protozoa: Testing the auxiliary microbial loop hypothesis. Acta Protozoologica 51:237–247.

Bonkowski, M., Villenave, C., and Griffiths, B. 2009. Rhizosphere fauna: The functional and structural diversity of intimate interactions of soil fauna with plant roots. Plant and Soil 321:213–233.

Chabrier, C., Carles, C., Desrosiers, C., Queneherve, P., and Cabidoche, Y.-M. 2009. Nematode dispersion by runoff water: Case study of *Radopholus similis* (Cobb) Thorne on nitisol under humid tropical conditions. Applied Soil Ecology 41:148–156.

Clarholm, M. 1985. Interactions of bacteria, protozoa and plants leading to mineralization of soil nitrogen. Soil Biology and Biochemistry 17:181–187.

Cole, C. V., Elliott, E. T., Hunt, H. W., and Coleman, D. C. 1978. Trophic interactions in soils as they affect energy and nutrient dynamics. Phosphorus transformations. Microbial Ecology 4:381–387.

Coleman, D. C., Anderson, R. V., Cole, C. V., Elliott, E. T., Woods, L., and Campion, M. K. 1978. Trophic interactions in soils as they affect energy and nutrient dynamics. Flows of metabolic and biomass carbon. Microbial Ecology 4:373–380.

Djigal, D., Brauman, A., Diop, T. A., Chotte, J. L., and Villenave, C. 2004. Influence of bacterial-feeding nematodes (Cephalobidae) on soil microbial communities during maize growth. Soil Biology and Biochemistry 36:323–331.

Ferris, H., Venette, R. C., and Lau, S. S. 1997. Population energetics of bacterial-feeding nematodes: Carbon and nitrogen budgets. Soil Biology and Biochemistry 29:1183–1194.

Ferris, H., Venette, R. C., van der Meulen, H. R., and Lau, S. S. 1998. Nitrogen mineralization by bacterial-feeding nematodes: Verification and measurement. Plant and Soil 203:159–171.

Finney, J. 1970. Random packings and the structure of simple liquids. I. The geometry of random close packing. Proceedings of the Royal Society of London A: Mathematical. Physical and Engineering Sciences 319:479–493.

Frost, H. 1978. Hole statistics in dense random packing. DTIC Document.

Georgis, R., and Poinar, G. O. 1983. Effect of soil texture on the distribution and infectivity of *Neoplectana glaseri* (Nematoda, Steinernematidae). Journal of Nematology 15:329–332.

Griffiths, B. S. 1990. A comparison of microbial-feeding nematodes and protozoa in the rhizosphere of different plants. Biology and Fertility of Soils 9:83–88.

Griffiths, B. S., and Caul, S. 1993. Migration of bacterial-feeding nematodes, but not protozoa, to decomposing grass residues. Biology and Fertility of Soils 15:201–207.

Hallmann, J., Quadt-Hallmann, A., Rodríguez-Kábana, R., and Kloepper, J. 1998. Interactions between *Meloidogyne incognita* and endophytic bacteria in cotton and cucumber. Soil Biology and Biochemistry 30:925–937.

Hassink, J., Bouwman, L. A., Zwart, K. B., Bloem, J., and Brussaard, L. 1993a. Relationships between soil texture, physical protection of organic matter, soil biota, and C and N mineralization in grassland soils. Geoderma 57:105–128.

Hassink, J., Bouwman, L. A., Zwart, K. B., and Brussaard, L. 1993b. Relationships between habitable pore-space, soil biota and mineralization rates in grassland soils. Soil Biology and Biochemistry 25:47–55.

Horiuchi, J.-I., Prithiviraj, B., Bais, H. P., Kimball, B. A., and Vivanco, J. M. 2005. Soil nematodes mediate positive interactions between legume plants and rhizobium bacteria. Planta 222:848–857.

Hunt, H. W., Wall, D. H., DeCrappeo, N. M., and Brenner, J. S. 2001. A model for nematode locomotion in soil. Nematology 3:705–716.

Iijima, M., Kono, Y., Yamauchi, A., and Pardales, J. R. 1991. Effects of soil compaction on the development of rice and maize root systems. Environmental and Experimental Botany 31:333–342.

Irshad, U., Brauman, A., Villenave, C., and Plassard, C. 2012. Phosphorus acquisition from phytate depends on efficient bacterial grazing, irrespective of the mycorrhizal status of *Pinus pinaster.* Plant and Soil 358:148–161.

Irshad, U., Villenave, C., Brauman, A., and Plassard, C. 2011. Grazing by nematodes on rhizosphere bacteria enhances nitrate and phosphorus availability to *Pinus pinaster* seedlings. Soil Biology and Biochemistry 43:2121–2126.

Jatala, P., Jensen, H. J., and Russell, S. A. 1974. *Pristionchus lheritieri* as a carrier of *Rhizobium japonicum.* Journal of nematology 6:130–131.

Knox, O. G. G., Killham, K., Artz, R. R. E., Mullins, C., and Wilson, M. 2004. Effect of nematodes on rhizosphere colonization by seed-applied bacteria. Applied and Environmental Microbiology 70:4666–4671.

Knox, O. G. G., Killham, K., Mullins, C. E., and Wilson, M. J. 2003. Nematode-enhanced microbial colonization of the wheat rhizosphere. Fems Microbiology Letters 225:227–233.

Kroupitski, Y., Pinto, R., Bucki, P., Belausov, E., Ruess, L., Spiegel, Y., and Sela, S. 2015. *Acrobeloides buetschlii* as a potential vector for enteric pathogens. Nematology 17:447–457.

Laaberki, M.-H., and Dworkin, J. 2008. Role of spore coat proteins in the resistance of *Bacillus subtilis* spores to *Caenorhabditis elegans* predation. Journal of Bacteriology 190:6197–6203.

Le Saux, R., and Queneherve, P. 2002. Differential chemotactic responses of two plant-parasitic nematodes, *Meloidogyne incognita* and *Rotylenchulus reniformis*, to some inorganic ions. Nematology 4:99–105.

Portillo-Aguilar, C., Villani, M. G., Tauber, M. J., Tauber, C. A., and Nyrop, J. P. 1999. Entomopathogenic nematode (Rhabditida: Heterorhabditidae and Steinernematidae) response to soil texture and bulk density. Environmental Entomology 28:1021–1035.

Prot, J. C., and Van Gundy, S. D. 1981. Effect of soil texture and the clay component on migration of *Meloidogyne incognita* 2nd stage juveniles. Journal of Nematology 13:213–217.

Queneherve, P., and Chotte, J. L. 1996. Distribution of nematodes in vertisol aggregates under a permanent pasture in Martinique. Applied Soil Ecology 4:193–200.

R. 2008. R: A language and environment for statistical computing. R Foundation for Statistical Computing, Vienna, Austria.

Rae, R., Witte, H., Rödelsperger, C., and Sommer, R. J. 2012. The importance of being regular: *Caenorhabditis elegans* and *Pristionchus* *pacificus* defecation mutants are hypersusceptible to bacterial pathogens. International Journal for Parasitology 42:747–753.

Rodger, S., Griffiths, B., McNicol, J., Wheatley, R., and Young, I. 2004. The impact of bacterial diet on the migration and navigation of *Caenorhabditis elegans*. Microbial Ecology 48:358–365.

Savin, M. C., Görres, J. H., Neher, D. A., and Amador, J. A. 2001. Uncoupling of carbon and nitrogen mineralisation: Role of microbivorous nematodes. Soil Biology and Biochemistry 33:1463–1472.

Sitaramaiah, K., and Singh, R. S. 1975. *Mononchus* spp as carriers of *Rhizobium japonicum* in soybean fields. Journal of Nematology 7:330–330.

Trap, J., Bonkowski, M., Plassard, C., Villenave, C., and Blanchart, E. 2015. Ecological importance of soil bacterivores for ecosystem functions. Plant and Soil. DOI: 10.1007/s11104-015-2671-6.

Wallace, H. 1958. Movement of eelworms. I. The influence of pore size and moisture content of the soil on the migration of larvae of the beet eelworm, *Heterodera schachtii* Schmidt. Annals of Applied Biology 46:74–85.

Young, I. M., Griffiths, B. S., Robertson, W. M., and McNicol, J. W. 1998. Nematode (*Caenorhabditis elegans*) movement in sand as affected by particle size, moisture and the presence of bacteria (*Escherichia coli*). European Journal of Soil Science 49:237–241.