FIELD GUIDE TO INVASIVE ALIEN INVERTEBRATES IN THE SOUTH ATLANTIC UK OVERSEAS TERRITORIES

PART 5 – INVERTEBRATES (except insects) & REFERENCES

Chris Malumphy, Sharon Reid, Rachel Down, Jackie Dunn, Debbie Collins and June Matthews
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### 6.27 Potato Cyst Nematode

**Order:** Tylenchida  
**Family:** Heteroderidae  
**Species:** 
- *Globodera rostochiensis* (Wollenweber)  
- *Globodera pallida* (Stone)

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#### Background

Potato cyst nematodes (PCN) comprise of two very closely related species, *Globodera rostochiensis* and *Globodera pallida*, which co-evolved with the potato in South America but have subsequently been introduced elsewhere with the production of potatoes. As such, these species are considered quarantine pests of actual or potential economic importance wherever potatoes are grown or traded throughout the world (EPPO, 2017; van Riel and Mulder, 1998). They occur in large soil masses and also adhere to potato tubers as microscopic cysts, which represent a complex of morphologically identical, but behaviourally different virulence groups or pathotypes. This presents major problems for their detection, identification and management (EPPO, 2017; van Riel and Mulder, 1998).

#### Geographical Distribution

PCN originated in the Andes Mountains in South America, from where it has spread with the introduction of potatoes to other regions. The present distribution covers temperate zones down to sea level and in the tropics at higher altitudes. In these areas, distribution is linked with that of the potato crop (Sullivan et al., 2007; van Riel and Mulder, 1998). *G. pallida* has been recorded as present in the Falkland Islands since 1992 (CABI, 2018; Zaheer et al., 1992).
Hosts

The major hosts of *G. pallida* and *G. rostochiensis* are restricted to the Solanaceae, in particular potato (*Solanum tuberosum*), tomato (*S. lycopersicum*) and aubergine (*S. melongena*) (Roberts and Stone, 1981; Sullivan et al., 2007).

Description

The female colour at the appropriate stage of development can be used as an indication of species: a female which changes during maturation from white to yellow, then into a brown cyst, is likely to be *G. rostochiensis* (Fig. 5.32.2), while one which changes from white directly to brown is likely to be *G. pallida* (Fig. 5.32.3) (Stone, 1973a and Stone 1973b).
In general, *Globodera* cysts are rounded with a small projecting neck, no terminal cone is present, diameter ± 450 μm, and they have a tanned brown skin (Fig 5.32.4). The cuticle surface has a zigzag pattern of ridges and a distinct D-layer is present. The eggs are retained within the cyst, no egg-mass is present (Stone 1973a; Stone 1973b; EPPO 2017).

The second-stage juvenile, the infective stage in the life cycle, hatches directly from the egg where it has already undergone a moult. They are mobile, vermiform in shape, annulated and taper at head and tail regions. The juveniles of *G. pallida* and *G. rostochiensis* are very alike, but *G. pallida* juveniles are generally longer as are their stylets (approximately length of juveniles: 445-510 μm long) (Stone 1973a; Stone 1973b; EPPO 2017).

Males are vermiform and relatively more abundant when environmental conditions are poor as they require less food than females and do not feed during the free-living stage (Evans, 1970). The vermiform males have a short life span of ten days or so and, during this time, will mate with as many available females as possible which secrete a pheromone to attract them (Evans, 1970; Green and Miller, 1969).

**Biology**

PCN cysts can contain as many as 500 eggs and can remain viable for many years before gradually deteriorating. Egg hatch is dependent on soil moisture, temperature, chemical exudates from potato roots, and day length. *G. pallida* eggs are adapted to hatch and develop at cool temperatures (10-18°C) whereas *G. rostochiensis* eggs are adapted to a higher temperature range of 15-25°C (Franco, 1979; Hominick, 1986).

The juvenile nematodes move between soil particles and locate and invade potato roots. Once inside the root, the nematode punctures the plant cells and feeds with its needle-like stylet. Feeding induces changes in the plant root cells which become abnormally large. Female nematodes which establish feeding sites on the roots become sedentary and progressively enlarge, rupturing the outer root tissue. Slender, male nematodes leave the roots and mate with females which now only have their heads embedded in the root and the posterior parts of their bodies exposed. Once fertilised, eggs develop inside the females which die about the same time as the potato crop flowers, becoming the brown cysts on the outside of the roots. When potato plants are lifted, the mature cysts drop off and remain dormant in the soil until further crops of potatoes are grown. In general, only one life cycle occurs on each growing crop and takes from 38-48 days to complete (CABI, 2018).

**Dispersal and Detection**

Natural dispersal is limited as the infective second stage juveniles only move at a maximum of about 1 m in the soil. However, cysts can be transported by wind and flood water. Most movement to new localities is by passive transport. The main routes of spread are infested seed potatoes and movement of soil (e.g. on farm machinery, boots, bins and plants) from infested land to other areas. Infestation occurs when the second-stage juvenile hatches from the egg and enters the root near the growing tip by puncturing the epidermal cell walls, and then internal cell walls, with its stylet. Eventually it begins feeding on cells in the pericycle, cortex or endodermis (EPPO, 2013).

Detection based on host plant symptoms are not specific. General symptoms include patches of poor growth in the crop, with plants sometimes showing yellowing, wilting or death of foliage; tuber size is reduced as a result, sometimes even when only minor symptoms are visible. However, many other causes can lead to these symptoms. Plants should therefore be lifted for a visual check on the presence
of cysts and young females on the roots, or a soil sample should be taken for testing. Young females and cysts are just visible to the naked eye as tiny white, yellow or brown globes on the root surface. Detection by lifting plants is only possible during a very narrow time lapse and is time-consuming. Soil testing is therefore the best way of determining the presence of potato cyst nematodes. Statutory sampling procedures and details of sampling and extraction methods can be found in EPPO Standard PM 3/30 (OEPP/EPPO, 1991). Detection based on host plant symptoms and identification by morphological and molecular methods is detailed in EPPO, 2017.

**Economic and other Impacts**

Potatoes are one of the top five important food and cash crops. PCN cause extensive damage, particularly in temperate areas and particularly when virulent PCN populations occur and any resistance has failed. The situation is worse with *G. pallida*, where commercial cultivars with good resistance are still limited and those that do, have undesirable characteristics. Economic loss due to PCN is estimated at around £50 million per annum and for Europe several times this amount (CABI 2018). PCN damage is related to the number of viable eggs per unit of soil and is reflected in the weight of tubers produced (Been et al., 1995; Elston et al., 1991; Philips et al., 1991).
6.28 Brown centipede

Order: Lithobiomorpha
Family: Lithobiidae
Species: *Lithobius forficatus* (Linnaeus)

**Background**

*Lithobius forficatus*, most commonly known as the brown centipede or stone centipede, is a common European centipede of the family Lithobiidae. It has been spread around the world by human activity as it commonly occurs in urban areas, in buildings and in waste and storage areas. Being a predatory organism that feeds on a variety of invertebrate taxa, it may have an ecological effect on the abundance of native soil-dwelling invertebrates in areas it invades.

*Lithobius forficatus* is morphologically highly variable, it has been hypothesized that *L. forficatus* is actually a complex of cryptic species, this has been supported by molecular research (Spelda et al., 2011; Giska & Babik, 2016). Eason (1969) discussed characters used to separate “subspecies”.

**Geographical Distribution**

*Lithobius forficatus* is native to the Holarctic region and is present throughout Europe and parts of North Africa. It was introduced to North America where it is widespread. It spread south to the South American Andean region where it has been recorded from Argentina, Chile, Colombia, Ecuador, Peru and Uruguay (Prado et al., 2018). It has also spread to Australia, New Zealand and South Africa. Of the UKOTs, it has been reported from Bermuda and Saint Helena (since 1865) (Varnham, 2006). Widespread in the higher parts of the island, it is one of three non-native Lithobiomorphs to have
invaded Saint Helena (Gray, 2018). Varnham (2006) notes an unconfirmed report of it on Ascension but lists it as doubtful. There was a find of a single female *L. forficatus* in an industrial area of Stanley, Falkland Islands (F.I.) in 2013. Discovered underneath plywood, it was to send to Fera Science for identification. Initially it was thought to be another European species, *L. melanops*, which established on the F.I (Barber, 2011; Fera unpublished records) and Tristan da Cunha (Lawrence, 1956). It is undetermined whether *L. forficatus* is established in the F.I. The “native” *Anopsobius macfaydeni* is much smaller, up to 9.2mm, yellow to pale brown and known from both West & East Falkland (Barber, pers. comm)

**Hosts**

Centipedes are nocturnal, opportunistic carnivorous predators, feeding on a range of small ground-dwelling invertebrates, including earthworms, small millipedes, enchytraeids, collembola, insect larvae and spiders. They are known to ingest some leaf litter on occasion (Coleman et al., 2004), Lewis (1965) notes that *L. forficatus* takes litter throughout the year.

**Description**

Centipedes are distinguished from superficially similar organisms by the presence of forcipules, the modified legs of the first trunk segment upon which the head rests. This segment bears the pincerlike fangs (poison claws) which have venom ducts opening at their tips (Coleman et al, 2004).

The following description is taken from Barber (2009). *Lithobius forficatus* adults range from 18mm-30 mm long and are chestnut brown in colour (freshly moulted specimens are pale blue). They have 15 pairs of legs as adults, but hatch from the egg with a reduced number of pairs (7 plus a pair of limb-buds) and go through a series of larval stages during which further legs are added at each moult. During these stages and the succeeding post-larval ones, the number of ocelli, teeth on the forcipular coxosternite, coxal pores etc. tend to show an increase in number until they reach adulthood.

Their head is slightly broader than long, the antennae have 35-43 articles and make up approximately a third of the body length. They have 20-30 ocelli on each side of the head arranged in 5 or 6 irregular rows. The forcipular coxosternite has 5 + 5 to 6 + 7 teeth. The 9th, 11th and 13th tergites have prominent projections at their posterior angles. The female genitalia have two conical spurs, occasionally a third on one side only. The claw of the gonopod has distinct dorsal and lateral denticles.

*Lithobius forficatus* can be separated from *L. melanops*, another stone millipede that has invaded the Atlantic UKOTS (namely Falkland Islands, Saint Helena and Tristan da Cunha) by its size and colour. *Lithobius forficatus* is the larger species and is more or less uniform chestnut brown, reaching 30mm at maturity whereas *L. melanops* is quite light but with a broad, dark longitudinal stripe and only reaches a maximum of 17mm in length (Barber, pers. comm). Other confirmatory characters include the presence of 5-7 teeth on each side of the forcipular coxosternite for *L. forficatus*, whereas *L. melanops* usually has 2, occasionally 3 and double claws on the 15th legs (they are single in *L. forficasus*).

There are other large brown *Lithobius* species, two of which (*L. pilicornis* and the vagrant *L. peregrinus* - see Barber, 2009) have been recorded in Britain where they are generally synanthropic and might possibly reach the South Atlantic UKOTS from there or by other routes (Barber, pers comm). South Atlantic climatic conditions are probably less favourable to them though, *L. pilicornis* is limited to the South West in Britain, and *L. peregrinus* has only been recorded on development sites, in port towns in southern England and on plant imports (Fera unpublished records). There are other lithobiids around the world with various degrees of similarity to *L. forficatus* (Barber, pers comm).
Biology

Lithobiomorpha generally inhabit leaf litter, moss, bark and under fallen tree trunks and stones in a variety of environments. Like other centipedes, they favour moist environments, as they lack a waxy coating on their exoskeleton that helps to prevent water loss. In its native or invasive distribution, *Lithobius forficatus* is often very common, especially in human influenced habitats and is sometimes found inside houses, other buildings and inside glasshouses (Barber, 2008; Barber, pers. comm.).

Lithobiomorph eggs are laid singly, and are carried at first in the genital claspers. *Lithobius forficatus* appears to lay eggs for a considerable part of the year, though there appears to be only a short period of sperm transfer in spring (Lewis, 1965). Lithobiomorphs go through a series of developmental instars and characters of the adult form with each moult (ecdysis). The earliest larval stages have fewer leg pairs (seven in *L. forficatus* hatchlings). The second developmental phase, the epimorphic phase, consists of 4 immature stages, in all of which there are fifteen pairs of legs and the sexually mature adult appears at the last one. (Cloudsley-Thompson, 1945). Development from egg to mature centipede takes roughly three years and *L. forficatus* have been known to live five to six years (references in Cloudsley-Thompson, 1945).

Small centipedes’ poison fangs are usually too weak to penetrate human skin (except perhaps where it is relatively soft e.g. between the bases of the fingers) (Barber, pers.comm), and their bite typically yields only minor discomfort, no worse than that caused by a bee sting. Centipede poison, used in the capture of prey, is a cocktail of several substances, including histamine, serotonin, cardiotoxin, and a quinoline alkaloid (Coleman et al., 2004).

Dispersal and Detection

*Lithobius forficatus* are not likely to disperse over long distances naturally, although they can move quickly when disturbed.

Accidental introduction within building material, bulk goods, plants, vegetables and hitchhiking on vehicles etc. is the most likely means of dispersal. Centipedes are nocturnal and tend to hide in small crevices and so transport via ship, trucks, containers can easily facilitate their spread. Barber (2011) noted that *L. melanops* has dispersed around the world by a result of human activity and that it most likely arrived in the Falkland Islands in material transported from the UK.

Detection during daylight hours is best done by visual inspection of suitable habitats, dark moist places such as under loose bark, stones, pots, logs etc. However, detection within shipments is more difficult due to their cryptic nature.

Economic and other Impacts

*Lithobius forficatus* is unlikely to have any major social or economic impacts should it invade a new geographical region such as one of the Atlantic UKOTs it has not already colonised, although centipedes can and do enter houses sometimes to the consternation of residents (Barber, pers. comm.) However, being a predatory organism that feeds on a variety of invertebrate taxa, it may have an ecological effect on the abundance of native soil-dwelling invertebrates. A major challenge in biodiversity studies is that in most cases, less than 20% of soil fauna taxa are described taxonomically (Coleman, et al. 2004), and for the microarthropods, such as the springtails, only about 10% of populations have been explored, and perhaps only 10% of species described (André et al., 2002).
The soil fauna consumes a wide range of organic material and are arranged in complex food webs. Macrofauna, such as insects are "bioturbators," literally movers of soil. Earthworms are even more influential, and considered primary "ecosystem engineers," moving plant and microbial residues and creating biopores in soil. A decrease in abundance of these important soil invertebrates could in turn may affect nutrient cycling and peat formation.

In the eastern USA Hickerson et al. (2005) studied the competitive relationships of the native centipede *Scolopocryptops sexspinus*, and the invasive *L. forficatus*. They found that both species consumed similar prey, but the native species was competitively superior as it predated on *L. forficatus*. *Scolopocryptops sexspinus* is a significantly larger species therefore this is unsurprising. Anecdotal evidence from Britain suggests that, in the locations where it occurs, the larger *L. pilicornis* may possibly out-compete *L. forficatus* (Barber, pers. comm).
6.29 Antarctic Soil Mite

Order: Sarcoptiformes  
Family: Nanorchestidae  
Species: *Nanorchestes antarcticus* (Strandtmann)

**Background**

*Nanorchestes antarcticus* is a minute Antarctic mite that lives in the soil near the surface or underneath rocks where they graze on algae (Block, 1980). The mite was selected to represent all soil-living mites which pose a threat to the pristine environment of the British Antarctic Territory and South Georgia. While this datasheet was being written, published records were found that indicate *N. antarcticus* already occurs to a limited extent in BAT and South Sandwich Islands (Pugh & Convey, 2000). This datasheet is therefore intended to highlight the risks of non-native soil mites in general.

**Geographical Distribution**

*Nanorchestes antarcticus* is the southernmost occurring arthropod in the world (Block, 1980; Fitzsimmons, 1971a). It is the most widely distributed Antarctic arthropod and is found over much of the Antarctic and sub-Antarctic regions (Block, 1980; Pugh & Convey, 2000).

**Host Plants**

*Nanorchestes antarcticus* are phytophagous, feeding predominantly on gelatinous red snow algae (*Ochromonas* sp.) but the algal diet is largely determined by abundance and availability rather than by preferential selection (Block, 1980; Fitzsimons, 1971b).

**Description**

*Nanorchestes antarcticus* is a small, soft bodied, prostigmatid mite (Fig. 6.29.1). The adults have a bluish body content against a red body colouring and have legs adapted for jumping. Their cuticle is flexible with regular folding over the entire body surface (Lindsay, 1972, Rounsevell & Greenslade,
1988). Females are approximately 269µm in length. During development the mite passes through four active immature instars: larva, protonymph, deutonymph and tritonymph which resembles the adult. The eggs are approximately 105.2µm in length and are red in colour (Lindsay, 1972).

**Biology**

*Nanorchestes antarcticus* are ovoviviparous, that is the eggs develop and hatch within the body of the female. The female can incubate several eggs simultaneously which may partly account for the success of this species in its Antarctic environment. When the larvae are born, they are enveloped in two coverings – the cuticle of the pre-larva and the egg shell. The larvae have only six legs. All nymphal and adult stages have eight legs. The protonymphs are approximately 172µm long, the deutonymphs approximately 191.8µm long, and the tritonymphs 222.1µm long. As the nymphs develop certain leg setae branch and one solenidion (a chemosensory sensilla which can be used for taxonomic identification) is added (Lindsay, 1972).

The habitat of *N. antarcticus* provides a highly predictable seasonal water supply and with few interacting micro-arthropods this species is an extreme A-strategist, i.e. it is long lived with low fecundity and with very limited powers of dispersal (Rounsevell, 1977). It can tolerate a wide range of environmental temperatures (-23°C to +31°C) (Block, 1980; Fitzsimons, 1971a) but are mostly active in the Antarctic summer, from October to March, when temperatures exceed 0°C diurnally (Rounsevell, 1977). During the Antarctic winter the mites are dormant in cold, dry soil just above the permafrost to a depth of 5cm (Rounsevell, 1981).

**Dispersal and Detection**

Dispersal of Antarctic arthropods is mediated by mechanisms beyond the control of individuals. The main natural dispersive agents are wind, water and birds. However, wind mediated transport is not likely for these mites as they live protected under stones and amongst vegetation and would not survive the extreme low temperatures and pressures associated with high speed/high altitude winds responsible for transoceanic dispersal (Marshall & Pugh, 1996; Pugh & Convey, 2000). Nevertheless, they may be dispersed locally by passive fallout of ice and snow in the Antarctic areas (Block, 1980). Ocean currents, or hydrochory, may be the most probable means of dispersal of these mites as they do inhabit coastal regions and can tolerate seawater. There are no reports of direct bird-mediated transportation although mites are highly likely to occur in bird colonies due to their organic enrichment of the habitat. There are no reports of human mediated dispersal to date, but with the increased diversity and intensity of human activities within this region it is only a matter of time before human mediated dispersal occurs.

Due to their small size, detection is by filtering ice/surface substrate and analysing these for presence of mites by use of a microscope.

**Economic and other Impacts**

*Nanorchestes antarcticus* does not directly impose any serious economic or other impact. However, global warming could cause ice to break and ocean currents to change, and with the ability of this species to withstand a broad temperature range, and to successfully colonise new areas, it may be able to invade other regions in the Antarctic and sub-Antarctic including other UKOT territories. In pristine environments such as BAT with low natural biodiversity it is important to prevent the introduction of any alien species of soil mite.
6.30 **Sheep Tick**

Order: Ixodida  
Family: Ixodidae  
Species: *Ixodes ricinus* (Linnaeus)

**Background**

The sheep tick *Ixodes Ricinus*, also commonly known as the castor bean or deer tick, is a small hard-bodied tick that infests livestock, deer, dogs and a wide variety of other species including humans. This tick acts as both a vector and reservoir for a series of wildlife zoonotic pathogens, especially the agents of Lyme disease, tick borne encephalitis and human granulocytic ehrlichiosis (Rizzoli *et al.*, 2004). Each active life stage (larva, nymph and adult) attaches to a single host and feeds on blood for a period of days, before detaching and then moulting or producing eggs. Whilst attached and feeding on their host they can be dispersed over large distances. All life stages will then ‘quest’ for a new host using an ambush technique whereby they climb up vegetation and wait for a host to brush past. However, during ‘questing’ the tick loses moisture, so they descend into the vegetation to rehydrate. The ticks require a relative humidity of at least 80% to survive this off-host period and are therefore restricted to areas with moderate to high rainfall (Medlock *et al.*, 2013).

Adult *I. ricinus* climb higher in the vegetation and usually only attack large animals from the size of a hare upwards. All three stages are known to bite humans. This rather indiscriminate feeding behaviour is responsible for the broad range of pathogens that this tick can transmit (CABI, 2019).

**Geographical Distribution**

*Ixodes ricinus* occurs in cool, relatively humid, shrubby or wooded areas. In addition to deciduous and mixed forests, it can be found in more open areas when the vegetation is dense, and rainfall is abundant. This tick is endemic in most of Europe (except for the Mediterranean region, which has a
warm, dry climate). It also occurs as far south as the Caspian Sea and northern Iran, as well as in forested areas of northern Africa (Sonenshine, 1991).

There are approximately 889 species of ticks worldwide. Of these, approximately 702 are classified in the family Ixodidae (‘hard-ticks’). Within this family, the genus *Ixodes* includes approximately 245 species. Of these, 14 belong to the ‘ricinus’ complex which includes four species (*I. scapularis, I. pacificus, I. ricinus* and *I. persulcatus*) that are considered responsible for the transmission of most pathogens of tick-borne diseases of animal and public health importance. These four species are widely distributed throughout the world (Swanson *et al.*, 2006). In addition, another species, *Ixodes urinae*, is found in both hemispheres and in Antarctica.
Host

*Ixodes ricinus* feed on a wide range of hosts including small rodents, passerines, and larger mammals like hedgehogs, hares, squirrels, wild boar and deer etc. The juvenile stages feed on the smaller hosts such as wood mice, whereas the adult stages feed on larger hosts like cattle and deer. Larger hosts are important in maintaining tick populations, with populations tending to be lower in the absence of larger hosts. Movement of hosts can also affect population numbers or result in the establishment of foci in new areas. *Ixodes ricinus* also has a high affinity for humans (Foldvari, 2016; CABI, 2019).

Description

*Ixodes ricinus* are hard ticks (Fig. 6.30.1) They have a dorsal shield and mouthparts that protrude forward when seen from above, they have no eyes, and the palpi are longer than they are wide. They are sexually dimorphic: the stigmatic plates are oval in males but circular in females. The ventral surface of the male has seven non-projecting, armour-like plates. The adults are red-brown in colour, but the female ticks are light grey when engorged (Fig. 6.30.2). Before feeding the males are approximately 2.5 - 3 mm long and the females 3 – 4 mm long. When they are engorged the females can be up to 10 mm in length (Kettle, 1990).

The eggs are laid in single large batches and are brown, globular, and possess a waterproofing waxy covering. The larvae are also brown, have six legs and are approximately 0.8mm in size (Fig. 6.30.2). They breathe through their skin and so water loss is exclusively through their cuticle. Once they have digested their blood meal, they moult into an octopod nymph (1.2 - 1.5mm; Fig. 6.30.2) (Kettle, 1990; Bates, 2012).
Ixodes ricinus are sometimes called castor bean ticks because the blood-gorged female resembles the mottled seeds of this plant.

Biology

Ixodes ricinus have four life stages: egg, larva, nymph and adult and a three-host life cycle (Fig. 6.30.3). Ticks must take a blood meal in order to moult to the next life stage and to produce eggs. The lifecycle is typically completed within three years but can be shorter or longer depending on climatic conditions and abundance of suitable hosts (Foldvari, 2016).

Mating usually occurs on the host and pheromones play an important role in finding a mate. Mating can last up to one week and once mated and fully engorged, an adult female will drop off a host onto the ground where she will seek conditions favourable for egg production. An engorged female will remain in this environment for four to eight weeks before eggs are produced. Up to 2000 eggs can be produced, after which the female dies and larvae hatch around eight weeks later (Foldvari, 2016).

Hungry ticks accumulate near the tips of grasses and other plants. When a host approaches, they seek to attach themselves by waving (questing) their first pair of legs. Adults seek hosts higher up the vegetation than nymphs, and nymphs higher than larvae. This is at least one of the explanations as to why adults are usually not found on rodents or lizards. The ticks remain on the host for several days (larvae 2 - 5 days; nymphs 2 - 7 days; females 6 - 11 days; and males weeks or even months). As with most members of the Ixodidae, I. ricinus spends 90-99% of its life off the host and has adaptive traits to withstand this, however, it cannot withstand long periods below 80% relative humidity (Jongejan & Uilenberg, 1994; Foldvari, 2016).

Dispersal and Detection

Ixodes ricinus larvae do not move horizontally over large distances so often remain aggregated within their environment whilst waiting for a host. Once on a host they may be dispersed by host movement while they feed, develop and moult to the nymphal stage. This is repeated at the nymphal and adult stage. The carriage of feeding ticks particularly by birds and large mammals is therefore crucial for their short and long-range dispersal. In addition, movement of infested livestock can increase their chances of long-range dispersal. However, I. ricinus is sensitive to climatic conditions and does require an animal host for all active stages for the completion of their life cycle.

Economic and other Impacts

There has been a notable increase in the prevalence of many tick-borne diseases (Parola & Didier, 2001; Randolph, 2001; Greenfield, 2011); as a result, ticks are of paramount importance globally, due to the implications on both human and livestock health.

Their ability to survive in such a diversity of habitat types and parasitize a multitude of hosts have made ticks a vector of interest over recent years. They are highly problematic in the sheep-farming and grouse-shooting industries (Davies, 2005), causing huge economic losses of several billion dollars per year, globally (Greenfield, 2011) with the deterioration of host health, including weight loss and breeding success (Balashov, 2007).
6.31 Springtail

Order: Poduromorpha
Family: Hypogastruridae
Species: Hypogastrura manubrialis (Tullberg)

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Figure 6.31.1 Dorsal view of Hypogastrura manubrialis found infesting mushrooms © 2016 Don Loarie, BugGuide

Background

Springtails are “decomposers” that thrive mostly on decaying organic matter, especially vegetable matter. They are an important component of the soil fauna biodiversity where they play a significant role in the nutrient cycle (Cassagne et al., 2003). They are good biodiversity indicators and can be used to record environmental impacts due to human activities (Fiera, 2009). Their name refers to the forked appendage on the underside of the abdomen that can be flicked backwards to fling the springtail into the air (Fig 6.31.2). Despite their abundance and significance, the collembolan fauna from many parts of the world are a relatively poorly studied, therefore a detailed account of the biology and distribution of H. manubrialis is not provided here. Hypogastrura manubrialis is a species within the large H. manubrialis ‘group’.

Geographical Distribution

Hypogastrura manubrialis is distributed worldwide although it is considered introduced in the Southern Hemisphere (Pacit, 1967), where it is known from Australia, Antarctic and Sub-antarctic, South America, South Africa and New Zealand (Bellinger, et. al., 2019). Greeslade & Convey (2012) list H. cf. manubrialis on Crozet Island (sub-Antarctic). There are over 7000 described spp. of springtail worldwide (Deharveng, 2004). The variety of collembolan species in an area is influenced by the soil
composition such as pH, aeration, organic matter combination, nutrient, humus type, vegetation covering and the physical characteristics of the soil etc. (Qazi and Shayanmehr, 2014).

**Figure 6.31.2** Ventral view of *Hypogastrura manubrialis* showing forked appendage © 2016 Don Loarie, BugGuide

**Figure 6.31.3** *Hypogastrura manubrialis* head with roundish mouthcone © 2016 Don Loarie, BugGuide

### Hosts

Most records of *H. manubrialis* are from compost and rich organic matter but they can also be found on the surface of puddles and water filled ditches (Hopkin, 2007). In South Africa *H. manubrialis* is typically associated with very nutrient-rich organic soils (Leinaas et al., 2015).

### Description

*Hypogastrura manubrialis* is a hexapod with a body length of about 1.5mm and are greyish or reddish blue in colour (Hopkin, 2007; Qazi and Shayanmehr, 2014). There are no larval or pupal stages; the newly hatched springtails resemble the adults and reach sexual maturity before they reach maximum size. They moult several times during their life cycle, and all stages require high humidity to survive (Qazi and Shayanmehr, 2014). Under normal conditions there are probably several generations per year (Chinery 1973).

### Biology

Springtails need conditions of high humidity to thrive and this is reflected in their choice of habitat such as soil, leaf litter etc. Despite their small size they may occur in huge populations and can be a significant component of the soil fauna (Barnard, 2011).

Their life cycle typically involves fertilisation of the female via a spermatophore delivered by the male. Mating behaviour can be simple or non-existent where the male simply deposits his spermatophore on the substratum at random leaving it to be discovered by the female (Hopkin, 2007). The eggs are deposited in clutches and take a few days or weeks to develop (Hopkin, 2007). After hatching they go through a series of instars, normally five, moulting every few days until they reach adulthood. They
may continue to moult throughout their adult lives and can survive for more than 6 months (Hopkin, 2007).

**Dispersal and Detection**

Despite having no wings, springtails have considerable powers of dispersal (Hopkin, 2007). Due to their small size they are easily carried into the atmosphere by the wind and have been trapped in nets trailed by aeroplanes at heights more than 3000 metres (Hopkin, 2007). In addition, they have been known to colonise remote islands by individuals clinging to driftwood and are easily transported globally by human activities (Hopkin, 2007).

Springtails are most obvious when they 'swarm'. Swarms occur following synchronized reproductions in conditions of ideal temperature and abundant food supply (Barnard, 2011; Hopkin, 2007).

Greenslade (2007) provides a comprehensive protocol for surveying collembola, including different collecting methods.

**Economic and other Impacts**

The invasion and establishment of an exotic species into an ecosystem has the potential to modify interactions between species at all trophic levels. This phenomenon is described as a trophic cascade (references in Greenslade, 2018).

Leinaas et al. (2015) observed high densities of *H. manubrialis* in heavily grazed vegetation in the Western Cape, South Africa, and briefly discussed the negative effect of this species on native springtail species.

Exotic invertebrates such as Collembola are a particular threat in the South Atlantic UK Overseas Territories because of the high endemism of the native fauna, in particular, to short-range endemic species by outcompeting them. Another collembolan, *H. purpurescens* is described as "highly invasive" and is thought to have displaced resident species from some habitats on the island of South Georgia (Convey et al., 1998.). *Hypogastura viatica* has been introduced to the Antarctic Peninsula and to a number of other sub-Antarctic Islands where it often dominates upper marine littoral habitats.
6.32 Springtail

Order: Poduromorpha  
Family: Onychiuridae  
Species: *Protaphorura fimata* (Gisin)

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**Figure 6.32.1** *Protaphorura armata* © Creative Commons, U. Burkhardt, 2005

**Background**

*Protaphorura fimata* is a species of springtail of European origin, and like most other collembola is an important component of the soil fauna biodiversity and are useful as bio-indicators of soil quality and can be used to record environmental impacts due to human activities such as soil pollution. However, *Protaphorura fimata* also behaves as a plant pest, as it feeds on the germinating seeds of lettuce (*Lactuca sativa*), causing severe stand losses in parts of the USA (Joseph & Bettiga, 2016).

Despite their abundance and significance, the collembolan fauna from many parts of the world are a relatively poorly studied, therefore a detailed account of the biology and distribution of *P. fimata* is not provided here. *Protaphorura fimata* is a species within the *P. armata* 'group' (Fig. 6.32.1), and morphological characters to separate species within the group are not reliable (Hopkin, 2007).

**Geographical Distribution**

*Protaphorura fimata* was originally described from Switzerland and seems to be widely distributed in Europe (de Jong et al., 2014). It was first reported from the USA under the name *P. fimata* in 2015 from a population in California, however most US records of *P. armata* appear to refer to *P. fimata*, suggesting that the species is widespread throughout the country (Joseph et al. 2015). It has invaded Australia (Greenslade, 2018) where it is recorded from agricultural land. In the Antarctic *Protaphorura fimata* has been recorded on the Macquarie Island (Australia) in 2010 (Greenslade, 2018) and has
been present on Deception Island since at least 1965, during a 2015 study, two individuals were found at Fumarole Bay in moss and two further specimens were recorded in 2017 at Kroner Lake beach with algae (Enriquez et al., 2019).

**Hosts**

Most collembola feed on fungal hyphae or decaying vegetation and have been reported to control fungal diseases on some plants (Hopkin, 2007). Protaphorura species are selective fungus feeders, avoiding some (often toxic) species, and that their selective grazing can affect the outcome of competition between decomposer fungi (Shaw, 2019). Collembola are usually beneficial, however in some cases they feed directly on plant material and cause economic damage. Many species of soil-inhabiting Onychiuridae feed on the roots of plants, including seedlings of field and greenhouse crops, a few species are predators of nematodes (Alford, 2011). *Protaphorura fimata* feeds on the germinating seeds of lettuce (*Lactuca sativa* L.), causing severe stand losses in the northern Salinas Valley of California (Joseph & Bettiga, 2016).

**Description**

Springtails are small, primitive, wingless invertebrates with mainly biting mouthparts, a sucker-like tube (the collophore) is present on the underside of the first abdominal segment and a forked, tail-like springing organ (the furcula) is present on the fourth segment (Alford, 2011).

As described in Joseph et al (2015): *Protaphorura fimata* is less than 2.5 mm in length; lacks eyes, and pigmentation, and carries 33/022/33333 pseudocelli dorsally on the head and body. Unlike other springtails, *P. fimata* lacks a furcula, and when disturbed does not jump but instead curls up. There are no larval or pupal stages; the newly hatched springtails resemble the adults and reach sexual maturity before they reach maximum size. *Protaphorura fimata* is a species within the *P. armata* group, and morphological characters to separate species within the group are not reliable (Hopkin, 2007).

Kaprus et al. (2016) provides an identification key to the *Protaphorura* in the Eastern Palearctic; Christiansen & Bellinger (1998) provide a key to the collembola of North America from which *P. fimata* can be separated from another; Convey et al. 1999 provide a key to the collembola of sub-Antarctic South Georgia.

**Biology**

Collembola prefer wet or damp surroundings and are usually are soil and litter dwelling, although some species move actively over the surfaces of bark and flowers in daylight. Collembolans are major components of terrestrial ecosystems (and particularly significant members of the soil communities), constituting a significant proportion of the animal biomass and are thus frequently and easily found (Handschin, 1955).

*Protaphorura fimata* is a subterranean species which reproduces sexually, their life cycle typically involves fertilisation of the female via a spermatophore delivered by the male. Mating behaviour can be simple or non-existent where the male simply deposits his spermatophore on the substratum at random leaving it to be discovered by the female (Hopkin, 2007). The eggs are deposited in clutches and take a few days or weeks to develop (Hopkin, 2007). After hatching they go through a series of instars, normally five, moulting every few days until they reach adulthood. They may continue to moult
throughout their adult lives and can survive for several years in case of some cave dwelling species (Hopkin, 2007).

**Dispersal and Detection**

Springtails can be carried by the wind and have been recorded at heights of more than 3000 metres, they have been known to colonise remote islands by individuals clinging to driftwood (Hopkin, 2007). Most significantly, they are transported globally by numerous human activities, particularly trade in plants and produce, movement of soil contaminated vehicles or equipment and tourism.

In South Georgia, alien invasive collembolean species numbers increased significantly after the introduction of exotic weeds around dwellings (Convey et al, 1999). Greenslade and Convey (2012) suggested that the importation of root and other vegetables was the route by which these species arrived and that that increased visitation by tourists, research and maintenance personnel seems an obvious cause despite strict quarantine controls imposed on all visitors. In 1994 Pugh highlighted the ineffective control on the import of contaminated equipment or vegetation by the military garrison at King Edward Point, or by the trawlers, yachts and cruise ships that dock.

Deception Island, in the South Shetland Islands is heavily visited by tourists on a daily basis over the summer. Several exotic collembolean species are only found near the geothermal vents (Greenslade et al., 2018), where tourists flock to bath in the warm water.

Use of a Berlese or Tüllgren funnel is the most effective way to collect Collembola by extraction of soil, leaf litter or bark. But they may also be collected by hand using a mouth operated aspirator, or hand operated aspirator. Overturning rocks and other objects frequently will expose them, and pitfall traps can be used to capture collemboles (Bellinger et al., 2019). Joseph and Bettiga (2015) used beet root (B. vulgaris) and potato (S. tuberosum) slices to bait capture *P. fimata* in the USA.

Greenslade (2007) provides a comprehensive protocol for surveying collemboles, including different collecting methods.

**Economic and other Impacts**

The invasion and establishment of an exotic species into an ecosystem has the potential to modify interactions between species at all trophic levels. This phenomenon is described as a trophic cascade (references in Greenslade, 2018). The cost in lost ecosystem services that invasive species can pose in native environments has been estimated, in one fresh water environment, to be in the millions of dollars (Walsh et al. 2016).

Exotic invertebrates such as Collembola are a particular threat in the South Atlantic UK Overseas Territories because of the high endemism of the native fauna, in particular, to short-range endemic species by outcompeting them. Another collembolean, *H. purpurescens* is described as “highly invasive” and is thought to have displaced resident species from some habitats on the island of South Georgia (Convey et al., 1998.). *Hypogastura viatica* has been introduced to the Antarctic Peninsula and to a number of other sub-Antarctic Islands where it often dominates upper marine littoral habitats.

As well as their impact on biodiversity, there are several reports of springtails as pests of crops reducing yields and productivity. *Protaphorura fimata* has been associated with feeding damage to germinating sugar beet seeds and sugar bean and most recently lettuce (references in Joseph et al 2015). Lettuce seedlings in fields with high densities of *P. fimata* show retarded or stunted growth and do not emerge in a synchronous pattern (Joseph et al 2015). The value of lettuce is estimated to be US$1.3 billion in the Salinas Valley (Monterey County Crop report 2012).
7. Acknowledgments

This guide was commissioned by Niall Moore and Jillian Key of the Non-Native Species Secretariat and is based on a series of projects funded by Defra delivered between 2009-2019 aimed at enhancing biosecurity in the UK Overseas Territories. We would like to express our gratitude to for their support and the many individuals in the UKOTs for their help and hospitality.

We would also like to thank Tom Prior (Fera) and Tony Barber for valuable input and reviewing some factsheets, and David Crossley (Fera) for taking most of the Crown copyright and Fera photographs, and for helping to format the final guide.

8. References


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Harris, R., Abbott, K., Barton, K., Berry, J., Don, W., Gunawardana, D., Lester, P., Rees, J., Stanley, M. Sutherland, A. & Toft, R. 2005. Invasive ant pest risk assessment project for Biosecurity New


MacGowan, J.A. 2016. Ants (Formicidae) of the south eastern United States. Mississippi State University Fact Sheet.


9. Appendices

9.1 Major sources of further information

The following list of websites provide a wealth of freely accessible information on invasive alien
species that pose a potential pant health risk to the UKOTs in the Atlantic. They also provide links to
further sources of information:

EPPO. 2019. European and Mediterranean Plant Protection Organization (EPPO)
https://www.eppo.int/
NNSS. 2019. GB Non-Native Species Secretariat – UK Overseas Territories Home –
http://www.nonnativespecies.org/ots/otsMap.cfm

9.2 Fera invertebrate plant pest identification Services
for the UKOTs

Defra provide to Fera to provide an identification service for suspected invasive alien invertebrate
plant pests found in the UKOTs. A submission form and instructions on how to submit a sample are
given below.
UK Overseas Territories Sample Submission Form

To help us with our diagnosis, please try and fill in as much information as possible?

**For Fera use only:**

### Your Details

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* Tick preferred method of contact

### The Sample

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What do you suspect the pest to be?

Approximate numbers of organisms found:

Life-stage(s) (if known):

Sample preservation: (see over for recommended methods)

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Description of host damage (if any):

Country of origin:

Other information:

I hereby authorise Fera to carry out diagnostic work on this sample. Signature:
## Sending your sample for diagnosis

### How to collect, package and send your sample

How a sample is preserved before posting is largely dependent on how long it will take to reach us. Please contact us for advice if you are unsure of how to collect and package a particular sample or if you wish to send photos of pests and/or pest damage before submitting a sample. Please email: ukot@fera.co.uk

### Collecting and Packaging:

- **Provide as many individual specimens of each suspected species as possible.** Some species are highly variable, or there may be mixed populations of related species on a host.

- **In some cases, adult life stages are necessary for species determination.** Larval forms of most Diptera (flies) and Lepidoptera (butterflies and moths) should ideally be collected with their food source and reared on before sending.

### Beetles, adult Lepidoptera, and other robust bodied invertebrates

Kill before sending, freezing is preferable. Carefully place the dead insect in a small tube or box and lightly wrap it with tissue (enough to stop it rattling around and becoming damaged). Avoid airtight containers due to possible mould formation. Prevent this by puncturing a few holes in the tube or box. Moths and butterflies can also placed in paper or glassine envelopes, but take care not to crush them. Pinning, carding and setting of organisms before posting is welcomed, please contact us if you require instructions.

### Larval invertebrates

Place in a small plastic tube filled with ethanol (70%).

### Scale insects, aphids, whitefly pupae and mites

Leave on the host where possible. Removing them can damage them making identification more difficult. Place the host leaf or stem in a plastic pot of 70% ethanol. With larger leaves or stems, or if the sample is going to take a while to reach us it is necessary to change the preservative after 4-5 days as the plant material will dilute the ethanol and the invertebrates will begin to rot.

If you have to detach specimens from the host material this is best done with a fine paintbrush or fine forceps. Place them in a small plastic container filled with ethanol (70%). Do not send specimens in formalin or any other formaldehyde based preservative.

### Soil samples

For nematode analysis. Place about 500g of soil in a strong plastic bag and seal.

### Sticky traps

Label, bend round sticky side inwards, and secure in position with adhesive tape.
Labelling and Posting:

- Each sample must be clearly labelled indicating host association, date collected, locality, reference numbers and any other pertinent information. Pictures of associated host damage are also useful.
- All samples should be put in a strong cardboard box and packaged securely. Include your Sample Submission Form licence and permits seal the box. We will provide you with a copy of our licence ‘Letter of Authority’.
- On the outside of the parcel a label should state “Dead Invertebrates for Scientific Study - No Commercial Value”. Please also place a copy of the appropriate permits and licences in and envelope and secure to the outside of the parcel.
- If the parcel is being sent by courier, please send us the tracking reference if possible.

Send your sample to: UKOT Project (Room 02FA04)
Fera Science Ltd
Sand Hutton
York
YO41 1LZ
United Kingdom

If you have any queries, contact: Sharon Reid or Dr. Chris Malumphy
Tel: +44 (0)1904 462000
E-mail: ukot@fera.co.uk