THYSANOPTERA

MORPHOLOGICAL IDENTIFICATION OF ADULTS AND LARVAE, WITH FOCUS ON *THRIPS PALMI* KARNY, 1925

MAIN DOCUMENT

Training session

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Anses-EURL insects & mites

CONTENT

Introduction

We know to date about 6400 species in Thysanoptera. It makes it a rather small order when compared to the major Coleoptera, Diptera, Hymenoptera, Lepidoptera, Hemiptera[1]... More, thrips go often un-noticed because of their tiny size and their lifestyle, crawling on or within vegetation. But this relative discretion is inversely correlated with their economic importance, since Thysanoptera also include very harmful phytophagous pests that cause millions dollars of losses in agricultural products and related human tragedies (Agroinsurance 2022).

This overview of the order is an introduction to the training session devoted to the morphological identification of Thysanoptera. It quickly presents various aspects of their biology, ecology and taxonomy. Noticeably, it cannot give a complete glance of the large diversity exhibited by Thysanoptera. Most especially, they present a large panel of behaviors, including complex ones as lekking, egg-mass defense, parental care, sociality and even eusociality). Readers may refer to the resources listed in the final section for further details. If not otherwise specified, the provided information is extracted from Lawrence Mound's general publications (Mound 2003, Mound 2005, Mound & Morris 2007, Mound 2018, Mound *et al.* 2022).

Taxonomy

The order is clearly defined from an unique synapomorphy: all thrips have a single developed mandible, the left one (Fig. 1), the right one being resorbed during embryogenesis. The traditional taxonomy of Thysanoptera is based on Mound et al. 1980's classification. It is still commonly accepted and used, since others discussed propositions exist (Bhatti 1988, Zherikin 2002). This classification divides the order into Tubulifera and Terebrantia.

Sub-order Tubulifera

The suborder includes a single family, the Phlaeothripidae. However, it is clearly polyphyletic, containing for example the large wastebasket genus *Haplothrips*. Actually, this sub-order received far less attention, because it contains very few species of any economic importance. Actually, the family gathers about 3 600 species, of which around 60% feed on fungi on dead leaves or branches. It also contains some impressive of co-evolutions and insect-plant interactions, as are the about 200 Phlaeothripinae species related to the genus *Acacia* in Australia.

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1 about 380K, 150K, 130K, 160K and 100K described species, respectively Wikipedia (2022) Wikipedia, The Free Encyclopedia.

Table 1. Taxonomical summary of the Thysanoptera order (Mound & Morris 2007, updated with ThripsWiki 2022 data).

Sub-order	Family	Sub-family	Genera	Species
Tubulifera	Phlaeothripidae	Phlaeothripinae	375	3062
		Idolothripinae	82	744
Terebrantia	Uzelothripidae		1	1
	Merothripidae		3	18
	Melanthripidae		4	70
	Aeolothripidae		23	221
	Fauriellidae		4	5
	Stenurothripidae		3	6
	Heterothripidae		4	89
	Thripidae	Thripinae	229	1762
		Sericothripinae	3	174
		Panchaetothripinae	42	144
		Dendrothripinae	12	109
Total			785	6392

Sub-order Terebrantia

This second suborder is slightly smaller, containing about 2800 species (Table 1). However, it was much more studied because 95% of these species are related to plants, of which many are cultivated. Hence Terebrantia contains nearly all the pest thrips of economic importance.

Terebrantia are subdivided into eight families. The largest one, Thripidae, includes the most famous pests, with some large genera like *Thrips* (301 species), *Frankliniella* (236 species), *Scirtothrips* (106 species)… (ThripsWiki 2022). A second family, Aeolothripidae, contains a bit more than 200 species. They are predators, though many of them may also feed on plant tissues. The six remaining minor families gather less than 200 species.

Economic importance
Some beneficials

Though largely reputed as pests, thrips may also have a beneficial impact in agriculture. First, many species are predators, as are the entire Aeolothripidae family. Their preys are mites or other thrips species. However, there are very few obligate predators. Most of predator species are opportunistic so, *i.e.* they may feed on both animal or plant tissue depending on the immediate environment. Noticeably, some well-known pests as *Thrips palmi* or *Frankliniella* spp. may shift to leaf mites.

Another aspect that is most often overlooked is their role in pollination. Their interference with bees' activity has been acknowledged since a long time, but thrips had been for long
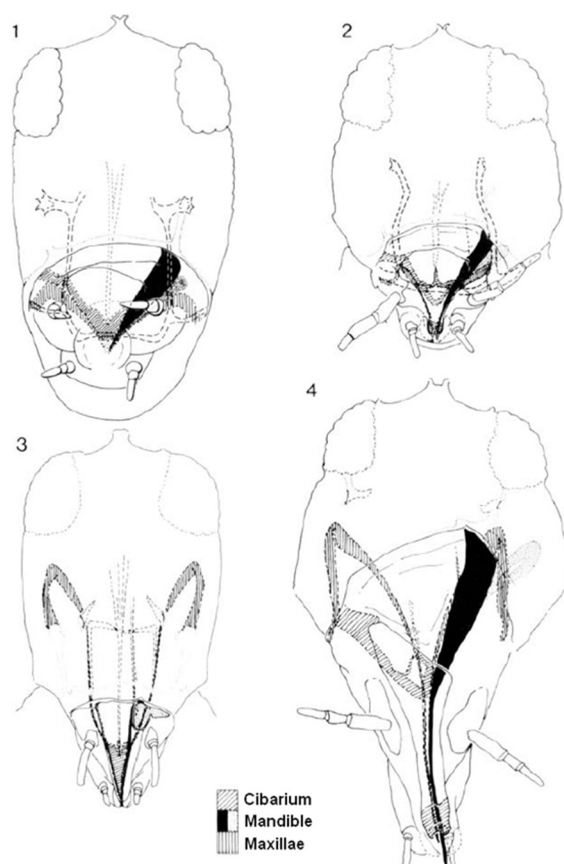

Figure 1. Heads of adult Thysanoptera (ventral aspect). 1.1: *Merothrips williamsi* (Merothripidae); 1.2: *Melanthrips juscus* (Aeolothripidae); 1.3: *Haplothrips verbasci* (Phlaeothripidae); 1.4: Chilochripspini (Thripidae) (Mound *et al.* 1980).

considered as too minute to carry a significant quantity of pollen when visiting flowers. Some species have also been reported as feeding on pollen grains. However, quantitative experiments showed that on the contrary they may transport large amounts because of their high number. More, some facts support the hypothesis that Thysanoptera were the earliest pollinating insects, and increasing numbers of plant species are shown to be associated with a given thrips species. Actually, their importance in pollination is still ill-understood but more and more evidences show that it should not been underestimated.

Mostly phytophagous pests

On the other hand, Thysanoptera are of course more famous as crop pests. In most cases, this pest status however largely depends on local conditions (Table 2). As mentioned above, many species behave as opportunistic predators on other arthropods. Most of recorded "pests" are also only locally or punctually acknowledged as such, *i.e.* having a significant negative impact on yield value. In some cases, finally, the damaging impact of a given species seems to have been artificially though incidentally selected. That is the example of *F. occidentalis* that spread in late 70's from its original distribution area to become a major crop pest: the now worldwide insecticide resistant strain(s) has putatively been selected by intensive chemical protection in horticulture (Kirk & Terry 2003).

Monophagous species rarely pose problems, *Liothrips karnyi* (Phlaeothripidae) being the exception. Thrips that are serious crop pests are usually highly adaptable and polyphagous species. This adaptability is reflected not only in their capacity to feed on various sources, but also in variation in length of larval life, body size at pupation, pupation site, and threshold temperatures for development. In substance, significant pests belong in majority to the Thripinae, a subfamily that gathers also nearly all the virus-vectors (Table 2).
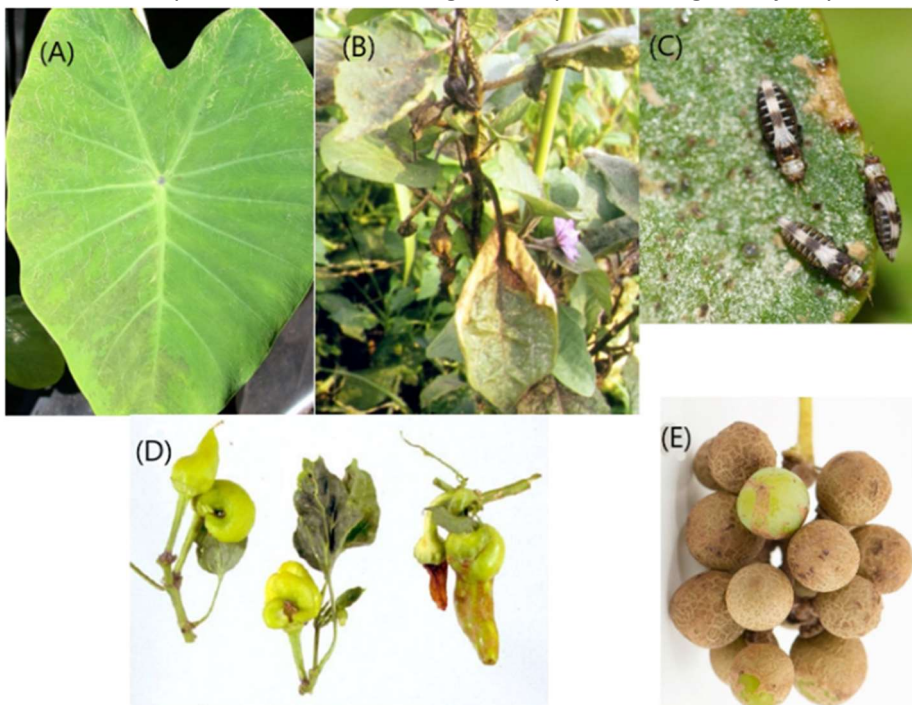
Thysanoptera may first cause direct damages when feeding or ovipositing on plants (Fig. 2). The value of a nectarine can be seriously reduced through a single thrips larva feeding on the fruit when it is young. Cucumbers, capsicums, and strawberries are badly distorted at times because of the feeding activity of thrips. Roses, carnations, and chrysanthemum flowers can be devalued through thrips feeding damage, and table grapes burst and become fungal-infected through thrips oviposition scars.

Figure 2. Feeding damage. (A) *Biltothrips minutus* markings on leaf of Taro (*Colocasia esculenta*). (B) *Thrips palmi* damage to aubergine crop (*Solanum melongena*). (C) *Dendrothrips ornatus* on *Ligustrum* leaf. (D) Damage to *Capsicum* fruits by *Franklliniella occidentalis* and Orthotospovirus. (E) Surface damage to grapes (*Vitis vinifera*) by *Scirtothrips dorsalis* (Mound *et al.* 2022).

But the main damages are caused by *Orthotospovirus* vectors. Currently, 30 species and one genus of Tospoviridae (Bunyavirales) are recognized. The genus is almost exclusively vectored by 15 thrips species, of which 14 are Thripinae members. They acquire the virus during larval stage only, when feeding on an infested plant. Noticeably, these vectors are not closely related, suggesting that the association of thrips and *Orthotospovirus* has arisen independently at different times and places. Among the most damaging *Orthotospovirus* one may cite the Tomato spotted wilt orthotospovirus (TSWV), Soybean vein necrosis orthotospovirus (SVNV), or Impatiens necrotic spot orthotospovirus (INSV). The only available management method against them is prophylaxy, *i.e.* vector suppression and infected plants removal.

Bio-ecology

Reproduction.

Thysanoptera have a biological cycle intermediate between hemi- and the holometabolous: larvae pretty different from imagoes, and pupae separate both, but juveniles and adults share a similar ecology. Two larval stages occur, followed a prepupal and a pupal stages. A second pupal stage is also differentiated in Phlaeothripidae. The morphology of all stages is detailed in the dedicated section.

As a rule, Thysanoptera are haplo-diploid, *i.e.* males are haploid and are produced by unfertilized ovocyts whereas diploid females hatch only from eggs. Further, thelytokous parthenogenesis (production of females without fecundation) has been observed in some species and is apparently linked to *Wolbachia* incidence (Arakaki *et al.* 2001). Finally, besides commonest oviparity, some cases of near ovo-viviparity are known in the larger Idolothripinae, fertilized eggs starting their development within female abdomen and hatching soon after being laid.

Feeding

As mentioned above, a rough half of thrips feed on fungi developing on dead plant tissues. Most of the remaining ones feed on plants, half on flowers and the other half on leaves. Finally, a few are obligate predators. The feeding behavior is similar whatever the ecology: the host surface is pierced using the functional mandible, then the feeding tube is inserted in the resulting hole. Saliva is then injected through the tube and the resulting mixture of saliva and host tissues is pumped back. If present, viruses are transmitted to the plant at this stage.

Throughout Thysanoptera, host-specificity range from strictly monophagous to highly polyphagous species. As an example, *Thrips obscuratus*, have been found on at least 225 plant species in 78 plant families. However, this has to been put in perspective by complementary observations. First, some monophagous species apparently have different host associations in different parts of their geographic range: *Apterothrips apteris* is restricted to *Erigeron* in California but is a minor pest of *Medicago* and *Allium* in Australia. Similarly, *Sericothrips gracilicornis* is specific to *Vicia cracca* (Fabaceae) in northern Europe but damages the foliage of Pinus in Spain and Italy, although the southern European reports do not indicate if this thrips is also present on *Vicia*.

Table 2. Thripidae species (by subfamily) impacting on human crop productivity (Mound *et al.* 2022).

Dendrothripinae		Thripinae		Thripinae (cont.)	
Dendrothrips latimaculatus	1	*Bathrips melanicornis*	1	*Megalurothrips distalis*	1
Dendrothrips minowai	2	*Biltothrips minutus*	1	*Megalurothrips sjostedti*	2
Dendrothrips octosparsus	1	*Anaphothrips obscurus*	2	*Megalurothrips usitatus*	2
Dendrothrips ornatus	1	*Anaphothrips sudanensis*	2	*Microcephalothrips abdominalis*	1
Leucothrips spp.	1	*Apterothrips apteris*	1	*Mycterothrips glycines*	1
Pseudodendrothrips mori	1	*Aptinothrips rufus*	1	*Odontothrips loti*	1
		Arorathrips mexicanus	1	*Organothrips bianchii*	1
Panchaetothripinae		*Aurantothrips orchidearum*	1	*Pezothrips kellyanus*	2
Anisopilothrips venustulus	1	*Bolacothrips spp.*	1	*Psydrothrips spp.*	1
Astrothrips spp.	1	*Ceratothripoides brunneus*	1	*Salpingothrips aimotofus*	1
Bradinothrips musae	1	*Ceratothripoides claratris*	2,3	*Sciothrips cardamomi*	1
Caliothrips fasciatus	2	*Chaetanaphothrips orchidii*	2	*Scirtothrips aurantii*	2
Caliothrips impurus	1	*Chaetanaphothrips signipennis*	2	*Scirtothrips bispinosus*	1
Caliothrips phaseoli	1	*Chirothrips manicatus*	1	*Scirtothrips citri*	2
Caliothrips striatopterus	1	*Corynothrips stenopterus*	1	*Scirtothrips dorsalis*	2,3
Caliothrips sudanensis	1	*Ctenothrips kwanzanensis*	1	*Scirtothrips inermis*	1
Dinurothrips hookeri	1	*Danothrips trifasciatus*	1	*Scirtothrips mangiferae*	1
Elixothrips brevisetis	1	*Dendrothripoides innoxius*	1	*Scirtothrips manihoti*	1
Helionothrips aino	1	*Diarthrothrips coffeae*	1	*Scirtothrips perseae*	2
Helionothrips annosus	1	*Dichromothrips corbetti*	1	*Stenchaetothrips biformis*	2
Helionothrips cephalicus	1	*Dichromothrips smithi*	1	*Stenchaetothrips spp.*	1
Helionothrips errans	1	*Dictyothrips betae*	1,3	*Stenothrips graminum*	1
Heliothrips haemorrhoidalis	2	*Drepanothrips reuteri*	1	*Systenothrips latens*	1
Heliothrips longisensibilis	1	*Echinothrips americanus*	2	*Taeniothrips euchariae*	1
Hercinothrips bicinctus	1	*Enneothrips flavens*	2	*Taeniothrips inconsequens*	2
Hercinothrips dimidiatus	1	*Florithrips traegardhi*	1	*Tenothrips frici*	1
Hercinothrips femoralis	1	*Frankliniella bispinosa*	2,3	*Thrips alliorum*	1
Euphysothrips subramanii	1	*Frankliniella brevicaulis*	1	*Thrips albopilosus*	1
Hoodothrips lineatus	1	*Frankliniella cephalica*	1,3	*Thrips angusticeps*	2
Panchaeothrips spp.	1	*Frankliniella fusca*	2,3	*Thrips atactus*	1
Parthenothrips dracaenae	2	*Frankliniella gardeniae*	1	*Thrips australis*	1
Retithrips syriacus	2	*Frankliniella gemina*	1,3	*Thrips calcaratus*	1
Rhipiphorothrips cruentatus	1	*Frankliniella hemerocallis*	1	*Thrips coloratus*	1
Selenothrips rubrocinctus	2	*Frankliniella insularis*	1	*Thrips flavus*	1
		Frankliniella intonsa	2,3	*Thrips florum*	1
Sericothripinae		*Frankliniella melanommata*	1	*Thrips hawaiiensis*	2
Neohydatothrips flavicingulus	1	*Frankliniella musaeperda*	1	*Thrips imaginis*	2
Neohydatothrips samayunkur	1	*Frankliniella occidentalis*	2,3	*Thrips madronii*	1
Neohydatothrips burungae	1	*Frankliniela parvula*	1	*Thrips major*	1
Neohydatothrips variabilis	1,3	*Frankliniella schultzei*	2,3	*Thrips meridionalis*	1
		Frankliniella tenuicornis	1	*Thrips nigropilosus*	2
		Frankliniella tritici	1	*Thrips obscuratus*	1
		Frankliniella williamsi	1	*Thrips orientalis*	1
		Frankliniella zucchini	1,3	*Thrips palmi*	2,3
		Fulmekiola serrata	2	*Thrips parvispinus*	2
		Kakothrips pisivorus	1	*Thrips setosus*	1,3
		Kurtomathrips morrilli	1	*Thrips simplex*	2
		Lefroyothrips lefroyi	1	*Thrips tabaci*	2,3
		Limothrips cerealium	1		

1 Local or minor pest; 2 Frequently a pest; 3 Orthotospovirus vector.

Moreover, even in polyphagous species the ease of transfer between hosts is sometimes asymmetric, such that populations of *T. tabaci* in Greece transferred successfully from various plants onto leeks, but populations on leeks would not transfer to tobacco. Finally, from polyphagous species may appear specialized strains. This appears to be so in *Scirtothrips aurantii*, the highly polyphagous South African citrus thrips that was found in Queensland, Australia, in 2002 but restricted almost entirely to *Bryophyllum* and a few other Crassulaceae.

Natural enemies

Several groups of organisms are now recognized as useful natural enemies against pest thrips: mites and anthocorid bugs (predators), eulophid hymenopteras (parasitoids), and nematodes and fungi (pathogens) (Loomans 2003). Thus, inundative releases combining *Amblyseius* with *Orius*, or *Hypoaspis* with nematodes, are sometimes used to reduced population of *F. occidentalis* in greenhouses. Odors produced by plants and by predators can determine the behavior of thrips. There are also extensive interactions between thrips and other small arthropods. When *Orius* bugs that have fed on thrips are present, *F. occidentalis* larvae shelter on leaves under tetranychid mite webbing, and preferentially attacks the eggs of a predatory mite rather than the eggs of a relatively harmless mite. Conversely, leaves that have been infested by *F. occidentalis* or mites attract *Orius laevigatus.*

Global effect of these predators on thrips populations is however hardly assessed. Worldwide there are many records of thrips predators but few analyses of the effect of such predators on natural thrips populations. Parasitic nematodes are widely recorded, although natural populations of few thrips species have been examined. Similarly, despite the literature on the importance of mites as predators in greenhouses, there is little attempt to assess the effect of mites on natural populations, although erythraeids are common on Terebrantia and tarsonemids are common on many larger Tubulifera. Finally, let mention that not only invertebrates prey on thrips: a few minutes watching birds or lizards are also known to do, although any effects remain unrecorded.

Slide preparation

The following chapters are based on Mound & Kibby (1998) and EPPO standards (EPPO 2002, EPPO 2018) methodologies. Identification of living thrips is usually impossible, except within a local fauna that is well studied. The existence, in many common species, of light and dark forms, often associated with considerable differences in body size both within and between sexes, usually necessitates careful collection and preparation of specimens on microscope slides, with the minimum of distortion or damage. Techniques for slide mounting are best considered under two headings: those appropriate for routine identifications, and those required by taxonomic research for reference purposes.

Collection and conservation

The best method of collecting thrips, apart from hand picking individuals from plants using a small brush, is to beat vegetation over a small plastic tray, using a narrow-bladed trowel or heavy knife. Individual plants, even single flowers or leaves, can be beaten, the precise position of the thrips located, and the plants then hand searched more effectively. They can thus be removed with a fine brush into collecting vials. In another method, plant parts may be stored for 24h in sealed plastic bags with filter paper to absorb moisture. Thrips will drop from the plant and may then be collected in the bag. Finally, Berlèse funnel may be used to deal with vegetal material as flowers, herbs or even dead branches.

Table 3. AGA, Hoyer and Faure's fluid preparations

AGA		Hoyer		Faure	
70% EtOH	8/15	Distilled water	100 mL	Distilled water	120 mL
Distilled water	5/15	Glycerin	40 mL	Acetic acid	20 mL
Glycerin	1/15	Arabic gum	60g	5% glucose solution	20 mL
Acetic acid	1/15	Chloral hydrate	400g	Arabic gum	40g
				Chloral hydrate	80g
Mix components at once		Dissolve the gum in water overnight, add other components, warm gently to complete dissolution and filter.			

The best fluid to collect into is AGA fluid (Table 3). This mixture helps to distend the body of most thrips and keeps the limbs supple. However, several laboratories have reported that AGA may act to denature the DNA of the thrips thereby hindering any subsequent molecular work. An alternative is to use non-mixed ethanol. 10% ethanol (added with wetting agent) is a good option for short conservation periods, stronger alcohols quickly make the thrips contract and become very rigid. However, 70-95% ethanol is better recommended if specimens are to be used for molecular studies. However, in this case specimens should be stored in the freezer until used, or they may prove difficult to slide mount. Specimens that are otherwise to be stored must be transferred to 60% alcohol and kept in the dark, preferably at temperatures well below 0°C, to prevent loss of color.

Temporary slide preparation (routine identifications)

The following method is rapid and thus relatively inexpensive. It allows a quick examination of both larvae and adults during routine identification work. Such slides are however not permanent and cannot be kept as references in collection. Specimens can be manipulated with precision brush or fine micro-pins, e.g. minuten pins mounted on broach holder. It is often useful to bend the apex of one of these pins slightly. A simple lifting tool to move specimens from one dish to another can be made from a small loop of fine wire (Figs 3, 4).

Remove the specimens from the collecting fluid into clean 70% alcohol. Place a drop of 30-85% lactic acid on a slide. Then transfer the specimens into the drop, dorsal side uppermost. If the specimens are reasonably flexible, attempt to open the wings and straighten the antennae using micro-pins. Cover the drop with the coverslip. Place immediately into an oven or on an heating plate at 35-40°C. Actually, specimens often collapse initially but then recover slowly when heated. Leave for 6 hours before attempting to study.

Figure 3. Mounting material. A: brush and micro-pins; B: ethanol (10%, 70% and 95% needed); C: 30% lactic acid; D: 0.1M KOH (NaOH also convenient); E: lavander oil; F: Canada balsam; G: microscope slides; H: centering template made from microscope slide; I: 12mm diameter rounded cover slips; J: 20x20mm square cover slips.

Permanent slide preparation (archiving and taxonomic research)

The idealized objective is to prepare specimens on to slides with their shape and color retained in a condition as close as possible to the natural, living state but with the body cleared so that surface detail is visible. Since this is rarely possible, a compromise is essential. A few specimens should be prepared for study without maceration in order to preserve their natural coloration, but the remainder should be macerated gently to reveal fine details of body sculpture and minute setae. Note that this preparation is not adapted for larvae that could collapse in drying balsam. See the next chapter for larvae.

Table 4. Permanent mounting: procedure summary

Step	Details
Piercing	Pierce abdomen laterally and thorax between mid coxae. Massage to expel body content.
Maceration	Minutes to hours in 0.1M KOH or NaOH
Dehydration	Several hours in 10% EtOH + wetting Several hours or days in 70% EtOH 5-10 minutes in 95% EtOH
Mounting	5-10 minutes in lavender oil Transfer into Canada balsam Weeks to months in 40° oven

Schematically, the whole method is subdivided into four steps: piercing, maceration, dehydration and mounting (Table 4). Piercing is essential to expel most of internal body content. While maintaining the specimen with a first micro-pin, puncture the abdomen laterally with a second one, and the thorax ventrally between meso-coxae. Subsequently, gently massage the specimen to expel body content through the holes.

The objective of maceration is to light the specimens by further removing the body contents. This is done by soaking the specimens in a weak alkaline solution (0.1M KOH or NaOH) for an appropriate period: half an hour for pale specimens, longer for larger or darker specimens. The length of the period of treatment must be determined by experiment. A tip is to regularly observe the specimens and carry on to the further step when eyes start to discolor. Beware that a too long exposure may damage specimens. Maceration should always be carried out at room temperature; heating causes damage to setae and the body surface.
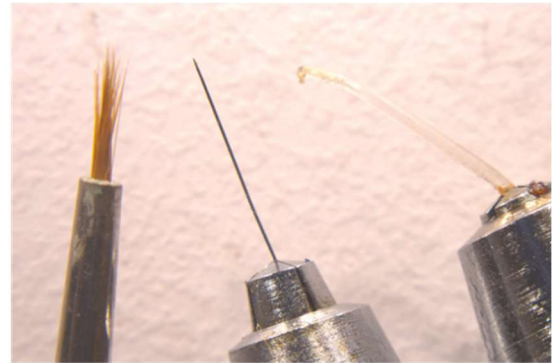

Figure 4. Close view of tool tips. From left to right: precision brush, mounted minuten pin, nylon wire fragment (fishing line) with tip flat crushed.

Transfer the specimens to 10% ethanol added with wetting agent. They may be left overnight. Another massage may help to expel organic remnants. Then, to 70% ethanol for at least one hour. This step may last several days as well. Finally, transfer them to 95% ethanol for 5-10 minutes.

After the final dehydration step, transfer the specimens to lavender oil for 5-10 minutes. It enables the transition to Canada balsam, because alcohol is not soluble into this latter. At this stage, it is recommended to spread the wings, legs and antennae. To facilitate the final mounting it is best first to prepare a small mounting block. This is done by marking the center of microscope slide with crossed lines from angles, providing a template. Use the template to locate the center of another slide, and put a drop of Canada balsam on it. The drop must be sufficient to avoid crushing the thrips. Transfer a single thrips specimen in the drop, dorsal side uppermost. Spread the legs and wings, and straighten the antennae by pressing on the basal segments with a fine needle. Then, place a clean 10-14mm diameter cover slip on the balsam drop, over the thrips, and gently press on so that the balsam spread over the whole circular surface. Exceeding balsam, if any, may be wiped out with lavender oil, but beware not to move the specimen. Then dry the slides until they are hard in an oven at about 40°C for several weeks. The quantity of balsam must be sufficient - after it has dried - to support the coverslip without distorting the specimen.

Semi-permanent mounting (long-term conservation of larvae)
An intermediate methodology is relevant for long-term conservation of larvae. Though not as durable as the permanent one using Canada balsam, it will preserve larvae in good conditions for years. In that process, transfer directly the larva into a drop of Hoyer or Faure fluid (Table 4), centered on a microscope slide. Then, place a clean 10-14mm diameter cover slip on the drop and press gently, as for permanent mounting. The difference here is that the circumference of the cover slip must be sealed hermetically. Several hardening fluid may be used for that purpose (*e.g.* nail polish).

Labelling
With the head of the thrips directed toward you (because microscope invert the image), the right hand label should indicate the host plant, followed by the country (in capital letters) and then the locality and date, with collector's name (and code number). The left hand label should indicate the sex, morph and genus and species names with author, with sufficient room left for any special notes to be added about that particular specimen e.g. measurements…

Morphology

Generalities

Thrips are, as a rule, tiny insects. Their size range from 0,5 to 15mm. They are all characterized by a synapomorphic (*i.e.* derived shared) trait, the presence of the sole left developed mandible. Consequently the ventral surface of head is asymmetrical. The maxillary stylets are coapted in a feeding tube emerging through mouth cone (Fig. 5). This tube is similar in all species, but its length is very variable: in some extreme cases, the stylets are longer than the whole body and thus retracted into coils within head at rest.
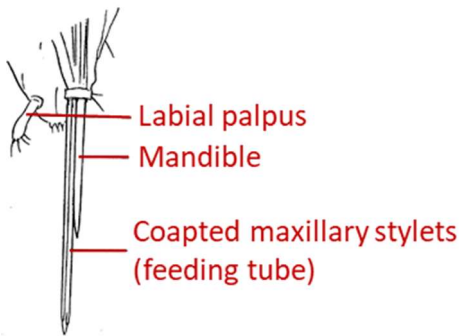
Figure 5. Lower head (profile). Detail of exerted mouthparts (after Palmer *et al.* 1989).

Adults bear a pair of antennae commonly with seven or eight segments, although the plesiomorphic (or ancestral) number is presumably nine, and various species have segments fused to produce lower numbers. Paired compound eyes are always present but reduced to a tenth of onomatidies in some species. Ocelli are also generally present though absent in many apterous adults.

Other body parts also bear features that are of great importance for generic and/or specific identification. The pronotum commonly has a specific regular number of major setae. The legs of adults lack typical insect tarsal claws, but each tarsus has an eversible bladder-like arolium. Finally, the abdomen is invariably composed of 10 segments.

Sexual dimorphism is common in the whole order. Besides the primary sexual organs, it is also often marked on wing development. Macropterous, brachypterous or apterous forms actually occur. But this intra-specific variability is not restricted to sexual dimorphism, since numerous species are variable for this aspect within a given sex. Morphological differences between forms is then usually not restricted to wing length (Fig. 6).

The present chapters focus mostly on Terebrantia morphology. Tubulifera are actually far less common in agricultural plant health activities. Both sub-orders are first distinguished by the structure of the 10th abdominal segment, especially in females, and the wings (Figs 7, 16).

Figure 6. *Kladothrips sterni* (Phlaeothripidae). Left: apterous female; right: winged female (Mound, 2018).

Suborders distinction

Tubulifera wings (when present) lack longitudinal veins, have a smooth surface without microtrichiae, and bear nonarticulating fringing cilia that insert directly into the wing membrane. The abdominal tergites of these species bear one or two pairs of sigmoid wing-holding setae, under which the wings lie flat on top of each other when at rest. Moreover, the 10th abdominal segment is tubular, with the anus terminal but the genital opening at the base of the tube, the female's ovipositor being an eversible, chute-like, structure (Fig. 17). In Terebrantia, forewings (when present) have two longitudinal veins, the wing surface is covered in microtrichia, and the fringing cilia are inserted into sockets. Wing-holding mechanisms vary considerably between species. The 10th abdominal segment is incomplete ventrally, and the ovipositor comprises four saw-like blades (Figs 17, 18).
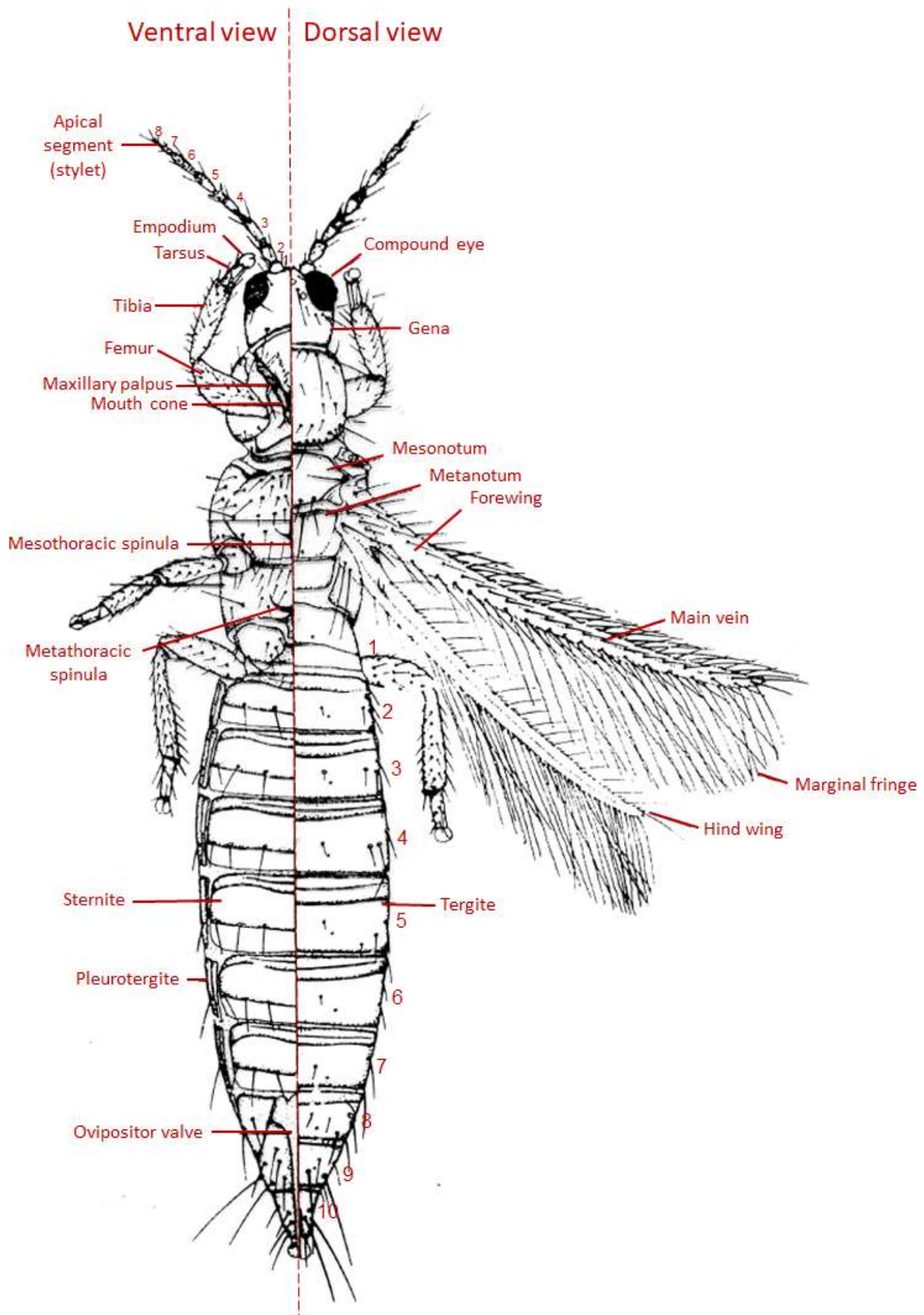
Figure 7. Terebrantia general morphology (*Frankliniella* sp.) (after Palmer *et al.* 1989).

Adults

This chapter lists the detailed terminology of the structures used for the morphological identification of adult Terebrantia thrips. Illustrations are extracted from Mound and Kibby (1998) and zur Strassen (2003).

The general body, as described in generalities, is shown in Fig. 6. The different parts and the associated chaetotaxy enable family, genus and/or species identification. Details are thereafter provided, precising the structures affected by inter- or intra-specific variability. Note that setae are numbered. Generally, 1 is attributed to the center-most pair. Ocellar setae are the exception: the pair 1 is the most forward one, the inter-ocellar pair being thus numbered either 2 or 3 (Figure 8). For thorax and abdomen, setae are called either marginal (main setae near segment margins), or discal (secondary, usually smaller setae, on segment disc).

Head

Antennal segments bear sense organ that are characteristic for the family or the genus. Most important is the shape of these organs on segments 3 and 4 (Fig. 9). Typically, Thripidae are defined by the presence of long and slender sense cones that may be simple or forked (Fig. 10). Second, the number and position of ocellar setae vary between genera and species (Fig. 11). Other discriminant features may be observed in the length of stylets, of genae, number and length of post-ocular setae, shape of eyes, and forward protrusion of head between antennae...
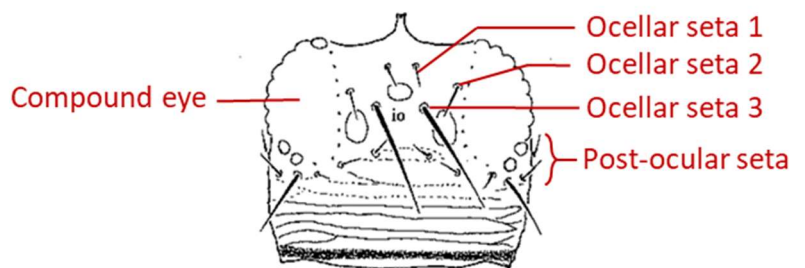

Figure 8. Head (dorsal). Chaetotaxy (*Frankliniella occidentalis*) (after zur Strassen, 2003).
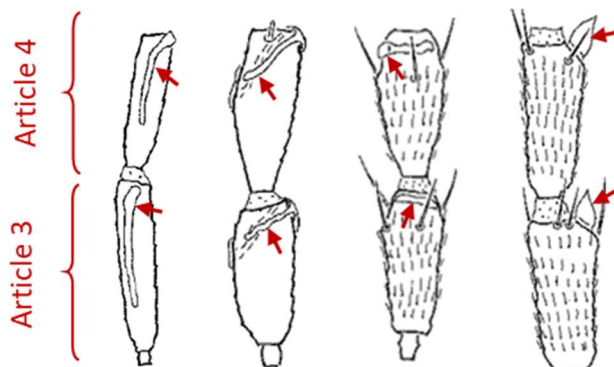

Figure 9. Antennal articles 3-4. Variation in shape of antennal sensoriae (arrows) in non-Thripidae families. From left to right: linear, elongate and longitudinal (*Aeolothrips propinquus* (Aeolothripidae)); linear, elongate and oblique (*Melanthrips paspalevi* (Aeolothripidae)); linear, elongate and transverse (*Ropotamothrips ressli* (Fauriellidae)); stout and cone-like (*Holarthrothrips tenuicornis* (Stenurothripidae)) (after zur Strassen, 2003).
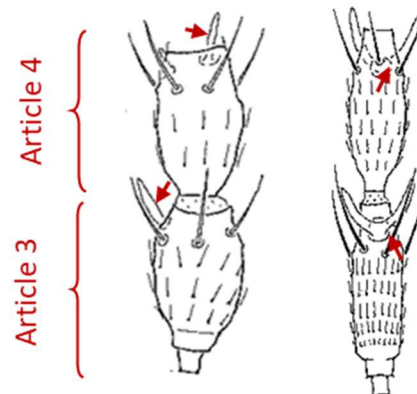

Figure 10. Thripidae antennal articles 3-4. Variation in shape of slender sense cone. Left: simple (*Bolacothrips jordani*); right: forked (*Thrips meridionalis*) (after zur Strassen, 2003).
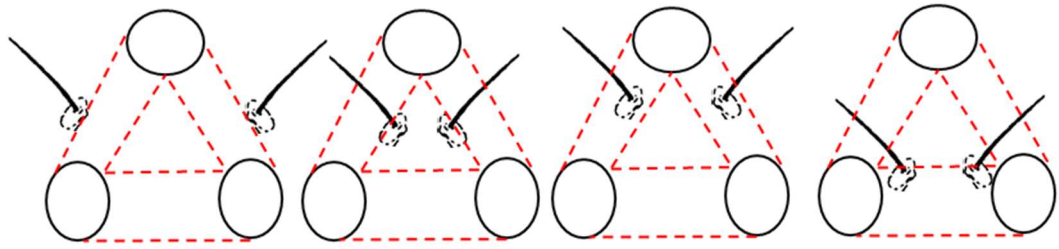
Figure 11. Schematic representation of ocelli showing variation in ocellar setae 3 position. From left to right: outside ocellar triangle; within ocellar triangle, on ocellar triangle margin, between posterior ocelli.

Thorax

The chaetotaxy of pronotum, most especially the number and length of antero- and postero-angular setae (Fig. 12), are of great importance. Pronotal sculpture is also sometimes used as discriminators. Further, the metanotum harbors a specific sculpture (Fig. 13). The mesonotum is rarely considered in the keys. On metanotum, species are also defined by the presence/absence of campaniform sensillae appearing under microscope as round pores. They may be isolated or paired, but this may be affected by intra-specific variability. Finally, the relative position of setae in regards of the anterior margin is sometimes a specific feature.
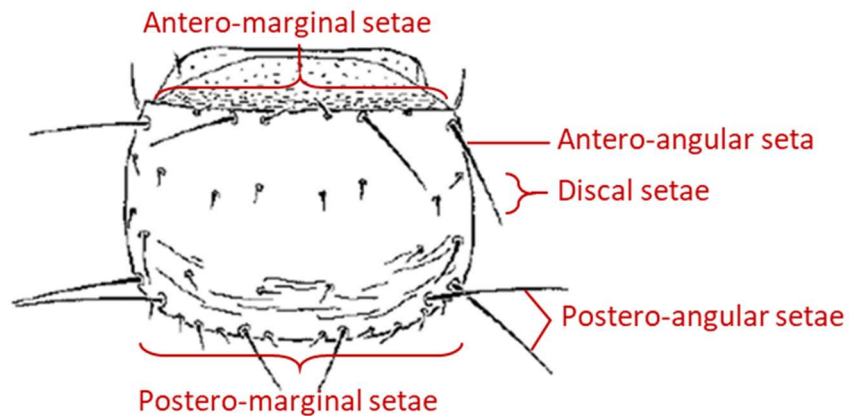

Figure 12. Pronotum (dorsal). Chaetotaxy (*Frankliniella intonsa*) (after zur Strassen, 2003).

Ventral features are also used, as is the shape and structure of the furcae (internal muscular attachment points) on meso- and metathorax (Fig. 14). A few genera exhibit a long lyre-shaped metathoracic furca. For most genera the trait to be observed is the presence/absence of a median spinula on both furcae.

On legs, one may observe the presence/absence of tarsal or tibial teeth, or tubercles (Fig. 15). Here again, this may vary within a given species (Fig. 6). Femurs and tibiae may bear setae of uncommon length and stoutness of generic importance.
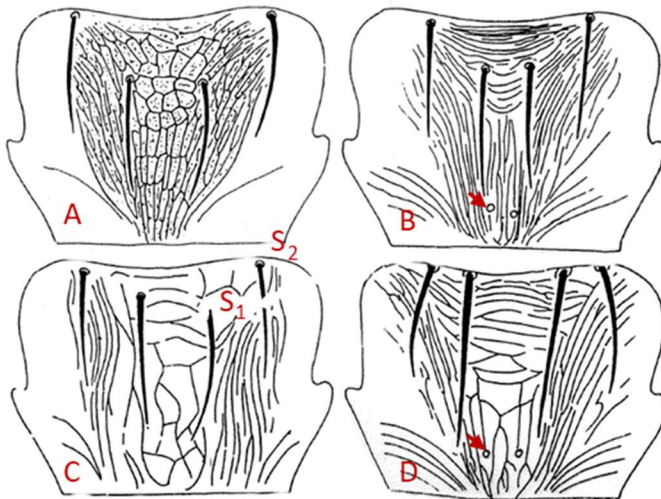

Figure 13. Variations in sculpture in metanotum (dorsal). A: densely reticulate medially, laterally striate (*Thrips simplex*); B: densely striate, striations converging posteriorly (*Thrips palmi*); C: loosely striate medially, laterally striate (*Thrips nigropilosus*); D: antero-medially transversely striate, postero-medially reticulate, laterally striate (*Thrips hawaiiensis*). Arrows indicate the campaniform sensilla (if present), S_1 and S_2 indicate the bases of setae 1 et 2 to point out their relative position to the anterior margin (after zur Strassen, 2003).
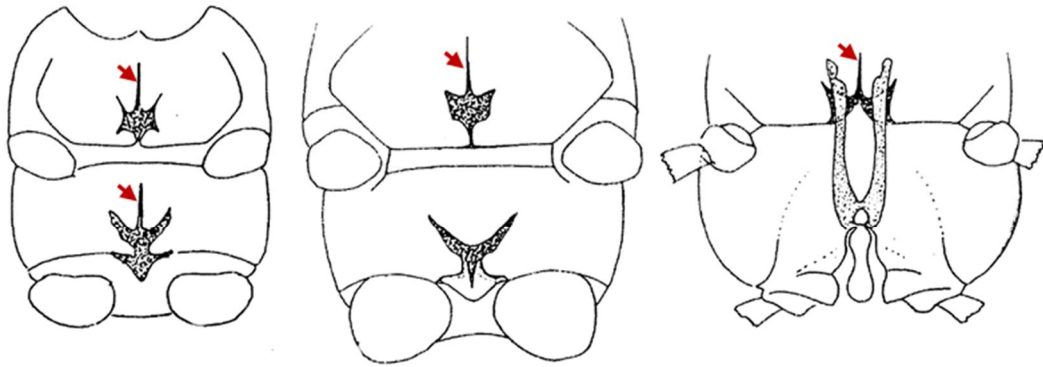
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Figure 14. Meso- and metasternum (ventral). Variations in shapes of furcae (darkened). From left to right: both furcae with spinula (arrows) (*Mycterothrips* sp.); metathoracic furca without spinula (*Ceratothripoides* sp.); metathoracic furca without spinula, elongate and lyre-shaped (*Dendrothrips* sp.) (after zur Strassen, 2003).

One of the most important structure for the identification of adult Thysanoptera is the forewing, fairly different between families and even subfamilies (Fig. 16). Forewings may be fully developed (macropterous), somewhat reduced (hemi-macropterous), reduced to stubs (brachypterous), or totally absent (apterous). This may be variable within a species and related to sexual dimorphism, or even within a given sex. It owns a postero-basal subdivision named clavus. Venation occurs only in Terebrantia: it is often hardly visible directly but may be deduced by the alignment of vein setae. Beside occasional short transverse veins, venation is reduced to one forked longitudinal vein that subdivides near basal third of the wing into a main (anterior) and a secondary veins. The precise number of setae on these veins is often an important feature, though it may slightly vary within a species. More, the presence/absence of a gap in the setae line is generically discriminant: for example *Frankliniella* spp. have a continuous line while in *Thrips* spp. one may distinguish a group of apical setae separated from the basal ones by a gap devoid of setae. Finally are sometimes helpful the general wing shape, as well as the length and shape of fringing setae along wing margins.
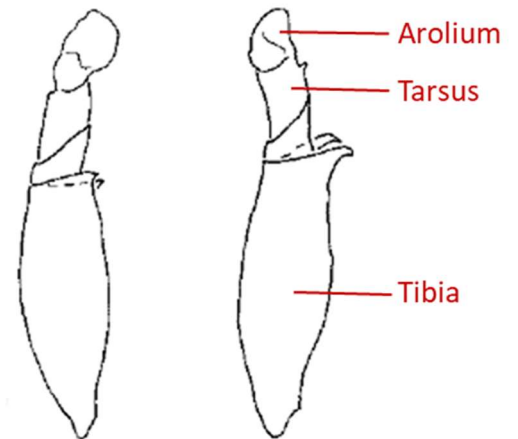

Figure 15. Variations in fore tibia and tarsus (left: *Odontothrips confusus*; right: *O. biuncus*) (after zur Strassen, 2003).
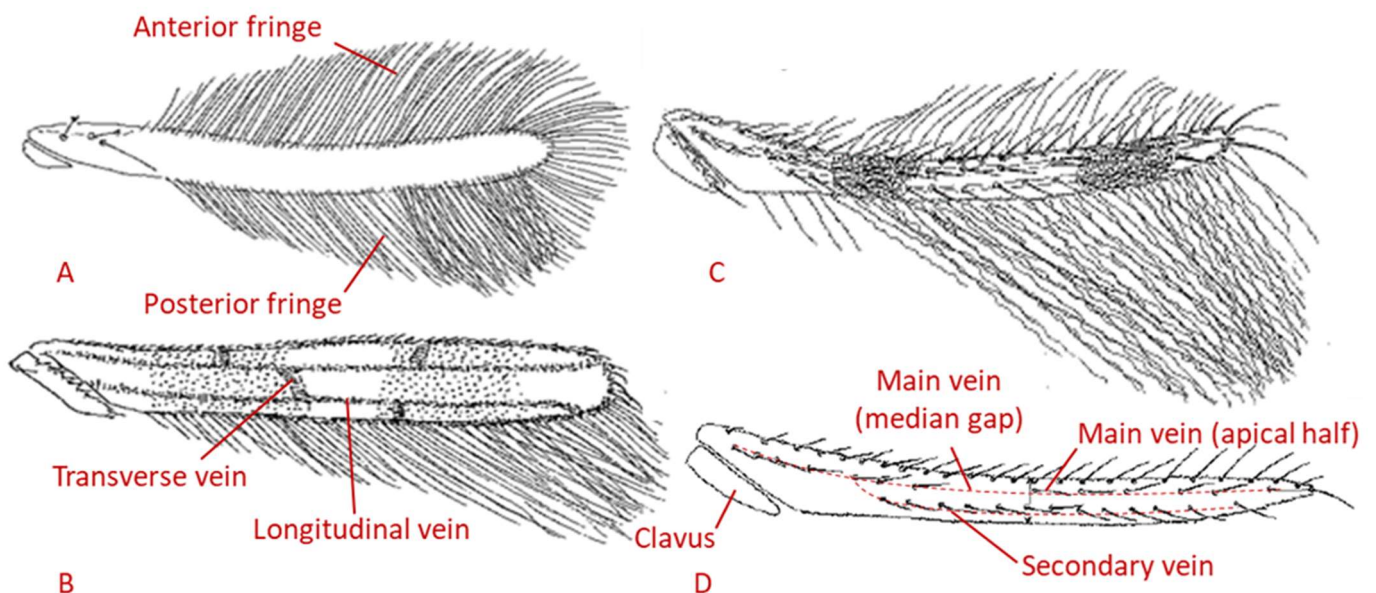

Figure 16. Variations in forewing. A: *Phlaeothrips coriaceus* (Tubulifera, Phlaeothripidae); B: *Aeolothrips intermedius* (Terebrantia, Aeolothripidae); C: *Hercinothrips bicinctus* (Terebrantia, Thripidae, Panchaeothripinae); D: *Thrips angusticeps* (Terebrantia, Thripidae, Thripinae) (after zur Strassen, 2003).

Abdomen

The first features to be observed on abdomen are the genitalia, useful for the sub-order (Fig. 17) and of course sex determinations. Terebrantia females have a sickle-shaped and toothed ovipositor, up- or down-curved (Fig. 18). Males usually harbor area porosa (porous secretory organs) on sternites, which may be of variable numbers and shape and enable specific identification (Fig. 19).

Several other abdominal structures are used in keys. First, the presence, location and numbers of setae, on sternites and tergites: the presence of discal setae on sternites enables for example an early segregation in *Thrips* spp. keys. Second, the presence or absence of microtrichiae. Pleurotergites, i.e. the lateral parts of tergites, are noticeable for this respect: for example,
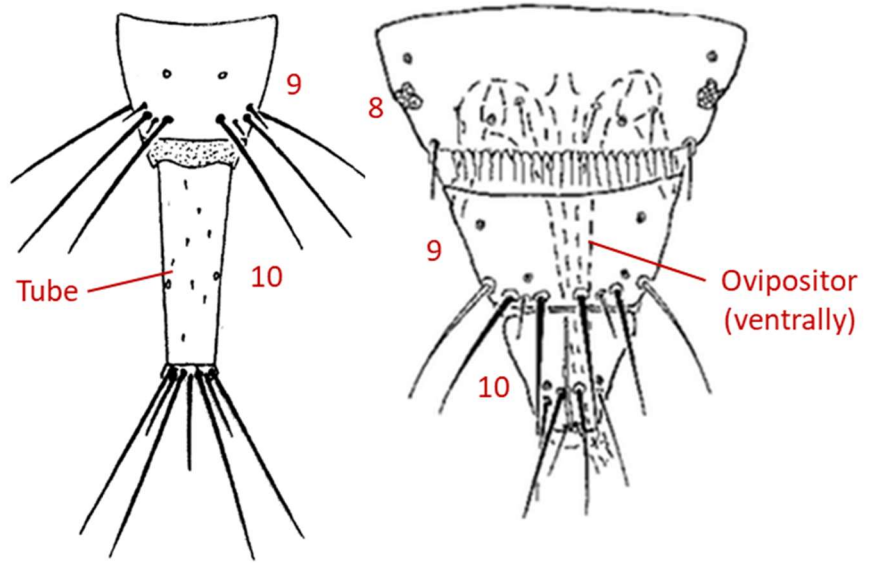

Figure 17. Detail of abdomen tip (dorsal) showing female genitalia. Left: Tubulifera (*Phlaeothrips* sp.); right: Terebrantia (*Anaphothrips* sp.) (after zur Strassen, 2003).

Thrips tabaci and a few other *Thrips* species are characterized by the presence of dense microtrichiae on pleurotergites, giving them a rough aspect under microscope (Fig. 20). Many species also have craspedae on tergites, i.e. a sclerotized expansion of their posterior margin (Fig. 21). The tergite 8 is of peculiar importance to check. It harbors several specific structures, as a posterior comb of variable aspect, large spiracles, and often (in Thripidae), a pair of ctenidia that are most
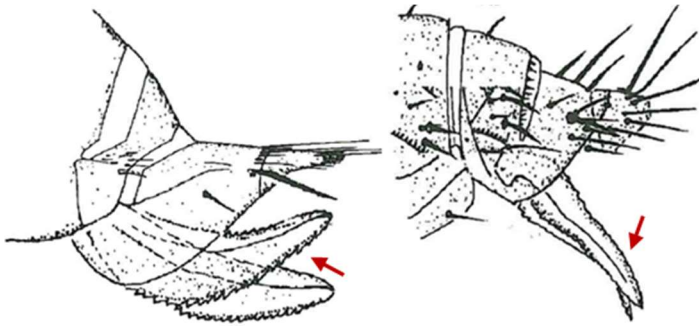
Figure 18. Ovipositor (arrows) in Terbrantia (profile). Left: Aeolothripidae; right: Thripidae (Mound & Kibby, 1998).

Figure 19. Male sternites detail, showing areae porosae. A: *Thrips vulgatissimus* (sternites 3-7); B: *Thrips validus* (sternite 5); C: *Thrips dilatus* (sternite 5); D: *Thrips oneillae* (sternite 3); *Euchaetothrips kroli*(sternites 3-5); F: *Idolimothrips paradoxus* (sternite 4); G: *Ceratothrips ericae* (sternite 5); H: *Hydatothrips boerhaaviae* (sternites 6-7) (after zur Strassen 2003).

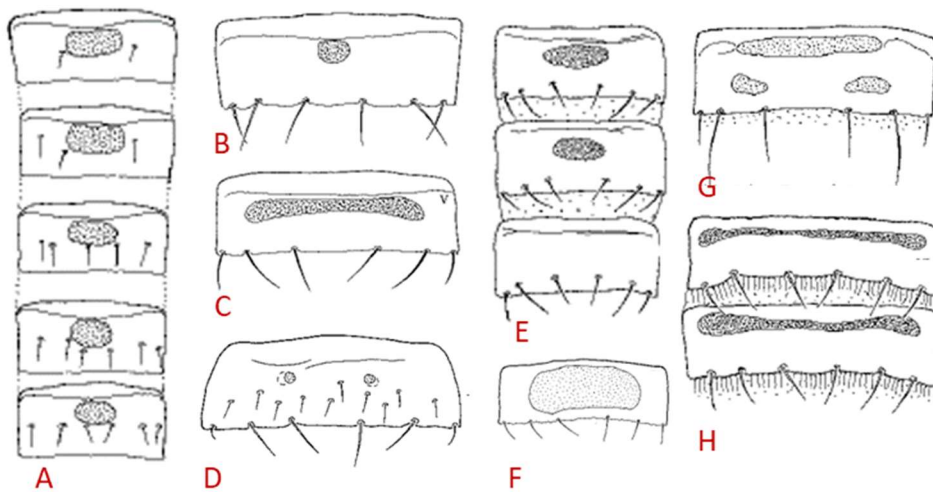often also present on previous tergites (Fig. 22). When present, the relative position of this ctenidium in regards of the spiracle is a critical feature to differentiate *Thrips* from *Frankliniella.* Finally, some other features are commonly considered, like the presence and position of campaniform sensillae on tergite 9, the number of lateral setae on tergite 2...

Figure 20. Detail of lateral part of abdominal segment showing pleurotergite (arrow). Left: pleurotergite without microtrichiae (*Thrips minutissimus*), right: with dense cover of microtrichiae (*Thrips tabaci*) (after zur Strassen 2003).
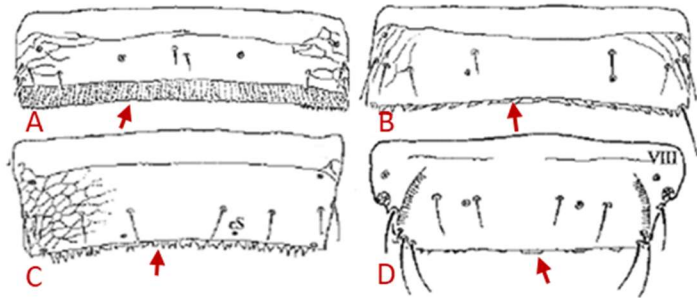

Figure 21. Detail of tergites showing variations in craspeda (arrows) expansion. A: *Parascolothrips priesneri* (tergite 5); B: *Stenchaetothrips biformis* (tergite 4); C: *Hemianaphothrips articulosus* (tergite 7); D: *Thrips sambuci* (tergite 8) (after zur Strassen, 2003).

Color

Color is often of limited utility in thrips determination, because it is intra-specifically variable within a given range, and because the total range of colors found in Thysanoptera is rather limited: the commonest color palette range from pale yellow to testaceous brown, sometimes added with grey, white, black or red. More, the lightening of specimens with KOH for slide-mounting removes most of natural coloration, making them merely appears as more or less dark during microscope examination. However, a general pale or dark habitus is often helpful because it is easy to notice, and thus often used in key. More, the color of given parts, especially the antennae and tergites, is commonly used as a final specific criterion.
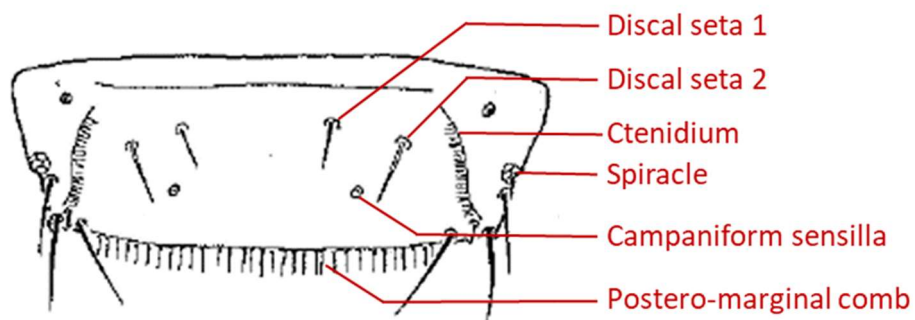

Figure 22. Detail of tergite 8 (dorsal) (*Thrips palmi*) (after zur Strassen, 2003).

Larvae

The identification of immature stages is pretty more difficult than adults. Vierbergen (2010) is one of the few publications dedicated to this purpose, and by far the most complete in European area. It covers 160 species of Thripidae reported in Europe, including invasive ones. Thripidae larvae I and non-Thripidae larvae II are identifiable to the family level, pupal stages are not.

When identifying a larva, the first thing to ensure is thus that the specimen is actually a second instar larva of Thripidae (Fig. 23). Larvae do not have wing pads, and slenderer antennae than pupae. Family and sub-family are readily determined by the examination of apical antennal segments (Figs 25D, 26). The first and second larval instars are firstly differentiated by chaetotaxy: larva I has six pairs of setae on pronotum, and a single one on abdominal sternites, while larva II has seven and three, respectively (Fig. 24).
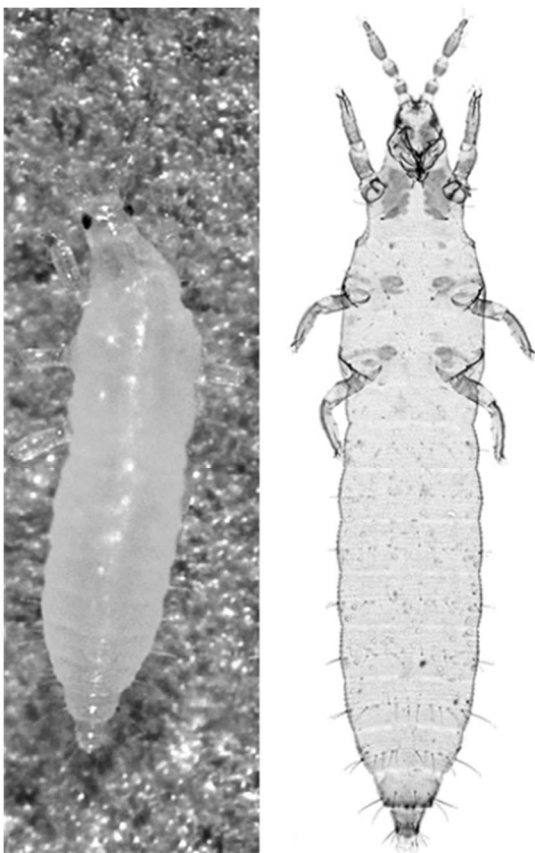

Figure 23. Second instar larva of *Frankliniella occidentalis* (Vierbergen *et al.* 2010).

Discriminant features in Thripidae larva II

The specific identification of second instar larvae strongly rely on details that are hardly visible under direct light. It therefore requires microscope equipped with phase contrast, or, better, interference contrast with polarized light.

Larvae offer less diagnostic characters than adults for identification, though this may also be artifactual since they received less attention from taxonomists. Chaetotaxy is of first importance. On abdomen, setae are numbered from middle outwards, as in adults. On head and thorax, the orientation is less visible and the numbering follows a fixed order (Fig. 25A). Setae may have various shapes, from acutely pointed to apically expanded (Fig. 26). Their length, absolute or relative, is also a commonly used cue.

Another critical discriminant feature is the examination of plates that covers the body, especially on abdomen. These may be either small, rounded, appearing as scattered dark spots, or rectangular and arranged into transverse rows (Fig. 27). Furthermore, the rectangular plates may bear, or not, apical microtrichiae. If so, they appear as pointed (Fig. 29). The presence of microtrichiae on plates is sometimes easier to assess in profile view, by observing the plates located on lateral margins of tergites.

The tergites 2 and 8 harbor or not a pair of spiracles (Fig. 25B). Their presence and shape are characteristic. Finally, the tergite 9 is of peculiar importance here (Fig. 25C). It bears a pair of campaniform sensillae whose distance from each other is compared to the distance between S_1 setae. The posterior margin of that tergite also bears teeth of variable stoutness. Also, it is more or less sclerotized, sclerotization appearing under microscope as a transverse darkening (Fig. 30).

Larva I
Pronotum: 2x6 setae
Tergites: 2x3 setae
Sternites: 2x1 setae

Larva II
Pronotum: 2x7 setae
Tergites: 2x3 setae
Sternites: 2x3 setae

Pre-pupa
Antennae extended
Wing pads short

Pupa
Antennae folded back
Wing pads longer

Figure 24. Immature stages in Terebrantia (after Mound & Walker 1982).
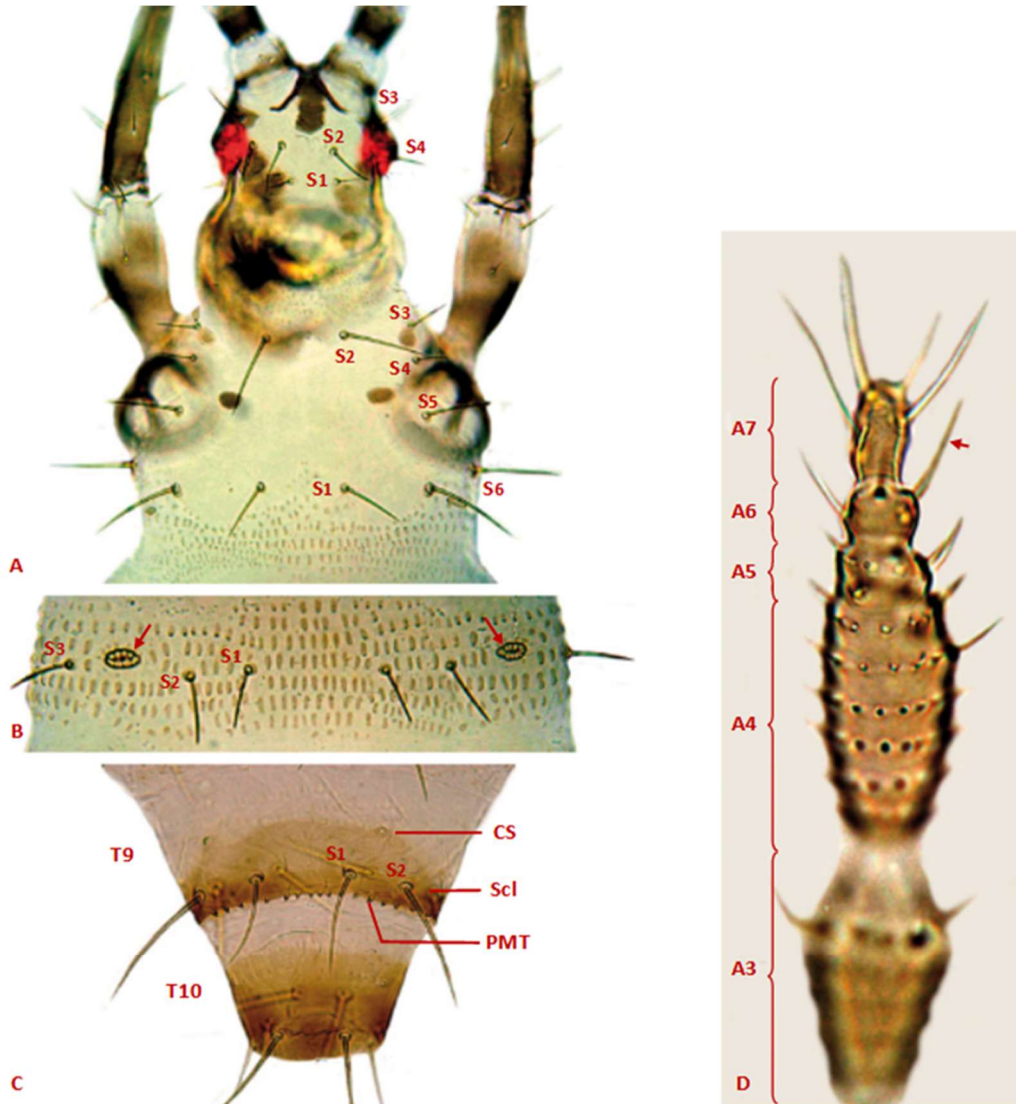
Figure 25. Chaetotaxy and indication of morphological structure of importance in larvae II identification. Sx: setae; Tx: tergites; Ax: antennal segments. A: chaetotaxy of head and pronotum (*Thrips vulgatissimus*); B: detail of tergite 2 showing spiracles (arrows); C: cle (4); chaetotaxy of abdominal tergites 9 and 10, showing campaniform sensillae (CS), posterior sclerotization of tergite 9 (Scl) and postero-marginal teeth (PMT) (*Pezothrips dianthi*); D: antennal segments 3-7 showing sense cone (arrow) (after Vierbergen *et al.* 2010).

Figure 26. Detail of antenna, showing the elongation and relative lengths of apical articles. Left: article 5 as long as 4 (Haplothrips gowdei, *Phlaeothripidae*); middle: article 5 less than half length of 4, apical article more than 7x longer than basally wide (*Heliothrips haemorrhoidalis*, Thripidae Panchaetothripinae); right: article 5 less than half length of 4, apical article less than 4x longer than basally wide (*Pseudodendrothrips mori*, Thripidae Thripinae) (after Vierbergen *et al.* 2010, and Ulitzka 2022).
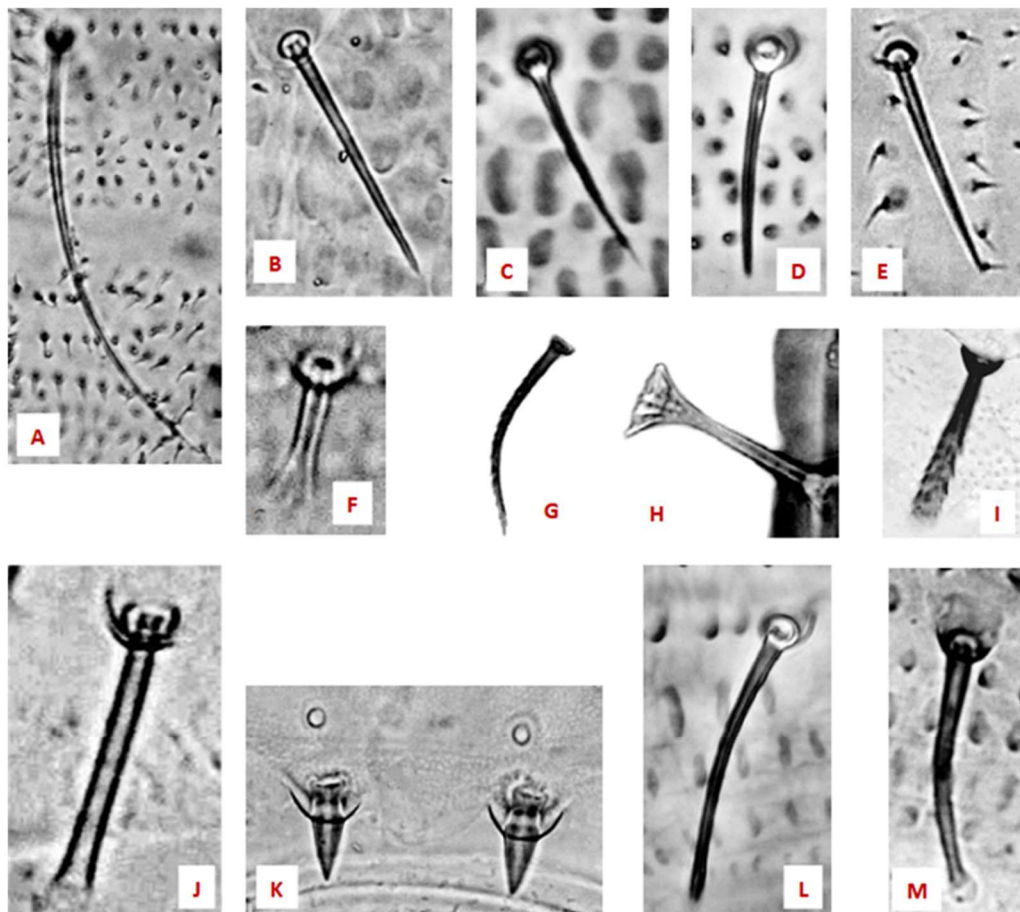
Figure 27. Types of setae (interference microscopy). A: flagelliform (*Euchaetothrips kroli*); B: pointed (*Taeniothrips inconsequens*); C: acute (*Thrips angusticeps*); D: blunt (*Thrips conferticornis*); E: acute (*Thrips major*); F: expanded (*Anaphothrips obscurus*); G: fringed (*Echinothrips americanus*); H: strongly expanded or crateriform (*Parthenothrips dracenae*); I: clavate (*Neohydatothrips gracilicornis*); J: slightly expanded and knobbed (*Dendrothrips ornatus*); K: conical (*Oxythrips ajugae*); L: blunt (*Thrips verbasci*); M: knobbed (*Tmetothrips subapterus*) (Vierbergen *et al.* 2010).

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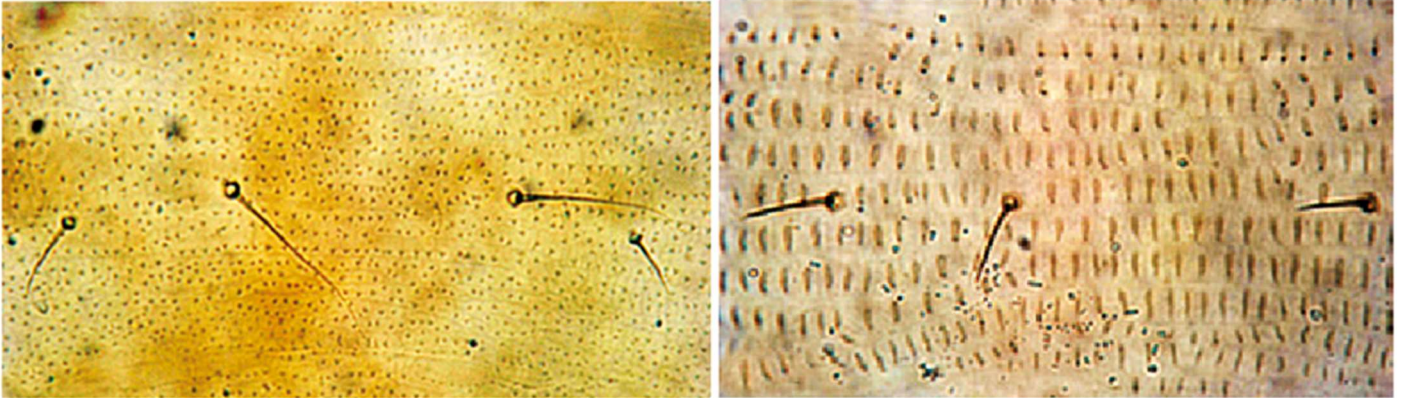
Figure 28. Detail of tergite surface showing plaques structures. Left: scattered small plaques (Dendrothripinae, Sericothripinae and part of Thripinae); right: with transverse rows of plaques (most Thripinae) (after Vierbergen *et al.* 2010).
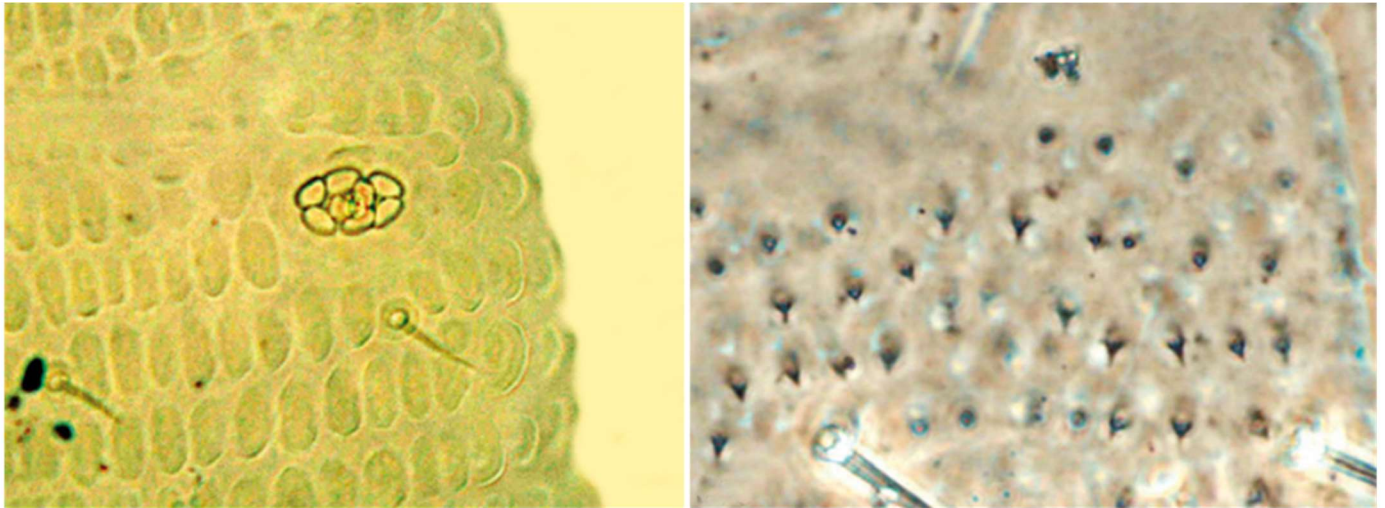
Figure 29. Detail of plates on abdominal tergites. Left: plates without apical microtrichiae (*Anaphothrips euphorbiae*); right: plates with apical microtrichiae (*Frankliniella intonsa*) (after Vierbergen *et al.* 2010).

Figure 30. Detail of tergites 9-10 showing variations in posterior sclerotization. Left: sclerotization totally absent (*Thrips palmi*); right: sclerotization strong (*Frankliniella pallida*) (after Vierbergen *et al.* 2010).

Thrips palmi Karny, 1925

A complete identification method for *T. palmi* may be found in the related EPPO standard (EPPO 2018) (Appendix). Here is additionally provided a diagnostic procedure for the morphological identification of second instar larvae (Fig. 31), pinpointing the features used in Vierbergen et al. (2010).

Table 5. Diagnostic features used for the identification of *T. palmi* larvae

Feature	Diagnostic level	Figure(s)
Abdominal sternites IV-VIII with 3 pairs of setae; pronotum with 7 pairs of setae	Larva II	32, 36
Antenna with segment 5 about 4x shorter than segment 4	Family	33
Antennal segment 7 2-3x times longer than wide, segment 5 wider than long and broadly attached to segment 4	Subfamily	33
Abdominal tergites 4-7 with transverse rows of plates	Subfamily	34
Abdominal tergite 10 with S1 setae bristle-like	Genus	32
Abdominal segment 9 with posterior margin barely toothed	Genus	32
Abdominal segment 9 with distance between campaniform sensillae slightly less than 1.5x distance between S1 setae	Genus	32
Abdominal segment 2 with spiracle distinct	Genus	35
Pronotum without plates, and with setae acutely pointed	Genus	36
Abdominal tergite 9 fully pale, without any sclerotization on apical margin	Species	32
Abdominal tergites 3-6 with 6-8 rows of plates, 1-2 of them lacking microtrichiae posteriorly to setae	Species	34

Practical guide for the identification of *Thrips palmi* second instar larvae

The following procedure enables to go through Vierbergen *et al.*'s key. We recommend to first get familiarized with the key before a most straight-forward attempt for *T. palmi* detection.

When examining a larva, the first point to ensure is that it is a Thripidae Thripinae. This is made by the examination of antennae (Table 5; Fig. 33). Then, that it is a second instar larva by examining the setae on pronotum and abdominal segments (Table 5; Figs 32, 36). Actually, it is sometimes difficult to number precisely the setae on pronotum: because procoxae also bear setae, because setae are sometimes broken, and because the larva I bears only one less. A good tip is thus to focus on the abdomen: a total of 12 setae are theoretically present on both tergite and sternite, they are readily countable by transparency. More, there is a great difference with the eight that are present on larva I, so that the discrimination is still possible even if some setae are broken.

Then, the examination of tergite 9 is very informative. *Thrips palmi* owns tergite without any sclerotization on posterior margin (Fig. 32), whereas this margin is distinctly darkened in most other commonly intercepted species, including all *Frankliniella* and other (though not all) *Thrips* spp. If the tergite is entirely pale, the other generic features (Table 5) have then to be checked (Figs 32, 35).

At this step, the specific identification relies mostly on the examination of abdominal plates. In *T. palmi*, there are 6-8 distinct transverse rows of plates on each tergite, some of which being devoid of microtrichiae. All the plates in front of the major S setae bear microtrichiae, they hence appear pointed apically. From setae level to the posterior margin, at least two rows of plate also have microtrichiae, but the 1-2 following ones lack such. This feature is often easier to see on larval sides, by observing the protruding plates in profile view. It is also easier to use the relief aspect provided by the interference contrast (Fig. 34).

Morphologically close species (larva II)

Other species that are most commonly found on intercepted shipment are readily differentiable with the features listed above. *Frankliniella* spp. harbor strong sclerotization and usually large teeth on posterior margin of tergite 9. *Thrips tabaci* lack visible spiracle on tergite 2 and also has a strongly sclerotized margin on tergite 9.

Other more or less common *Thrips* spp. have no sclerotization on tergite 9: *T. major*, *T. flavus*, *T. parvispinus*… If the tergite 9 is totally pale, it is therefore of critical importance to check the number of plate rows and the presence/absence of microtrichiae. To the current knowledge, the only other species with a similar pattern of microtrichiae is *T. juniperinus* : it owns 15 rows of plates on each tergite and is moreover restricted to *Juniperus communis.* However, the precise assessment of the presence of microtrichiae may be difficult. Thus, if the tergite 9 is fully pale, it is highly recommended to check also the other specific features mentioned in Vierbergen *et al.* (2010).

Figure 31. *Thrips palmi*, second instar larva (x100). Habitus.

Figure 32. *Thrips palmi*, second instar larva (x800, phase contrast). Detail of tergites 8-10. On segment 8, a total of 12 setae are visible: three pairs on tergites and three pairs on sternite by transparency. On segment 9, the posterior margin is not at all sclerotized, the postero-marginal teeth are hardly developed, and the distance between campaniform sensillae is slightly less than 1,5x the distance between S1 setae. On tergite 10, setae are slender and acute.

Figure 33. *Thrips palmi*, second instar larva (x600, phase contrast). Detail of antennae showing the elongations of antennal segments 4, 5 and 7.
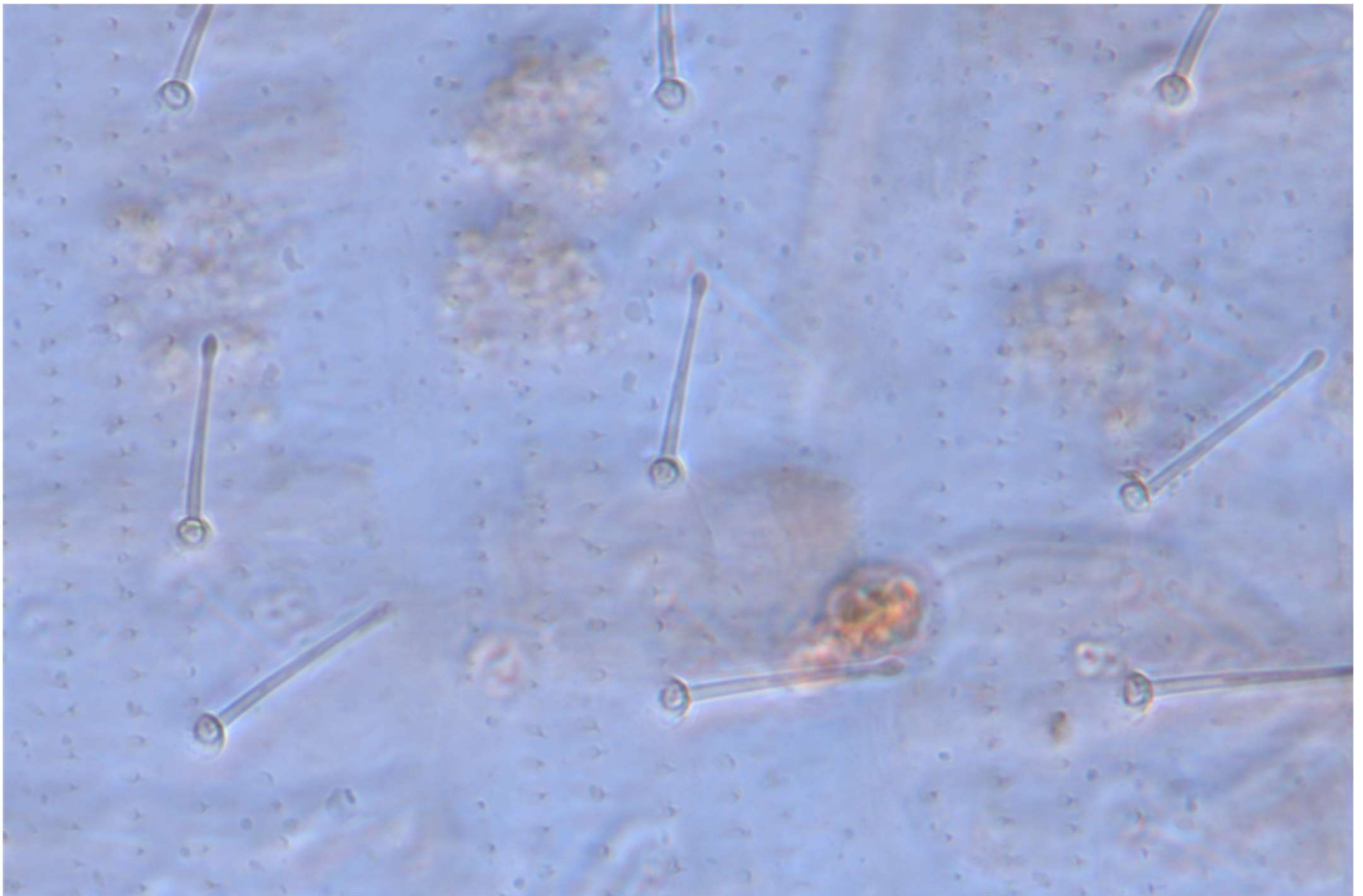
Figure 34. *Thrips palmi*, second instar larva (x800; top: phase contrast; bottom: interference contrast). Detail of tergites 5-7 showing seven rows of plates (numbered) from either sides of setae S. Rows 1-5 bear microtrichiae, 6-7 are weaker and without microtrichiae.

Figure 35. *Thrips palmi*, second instar larva (x600, phase contrast). Detail of tergite 2 showing spiracle (arrow).

Figure 36. *Thrips palmi*, second instar larva (x400, phase contrast). Detail of head and pronotum showing the absence of plates and the acute setae on pronotum.
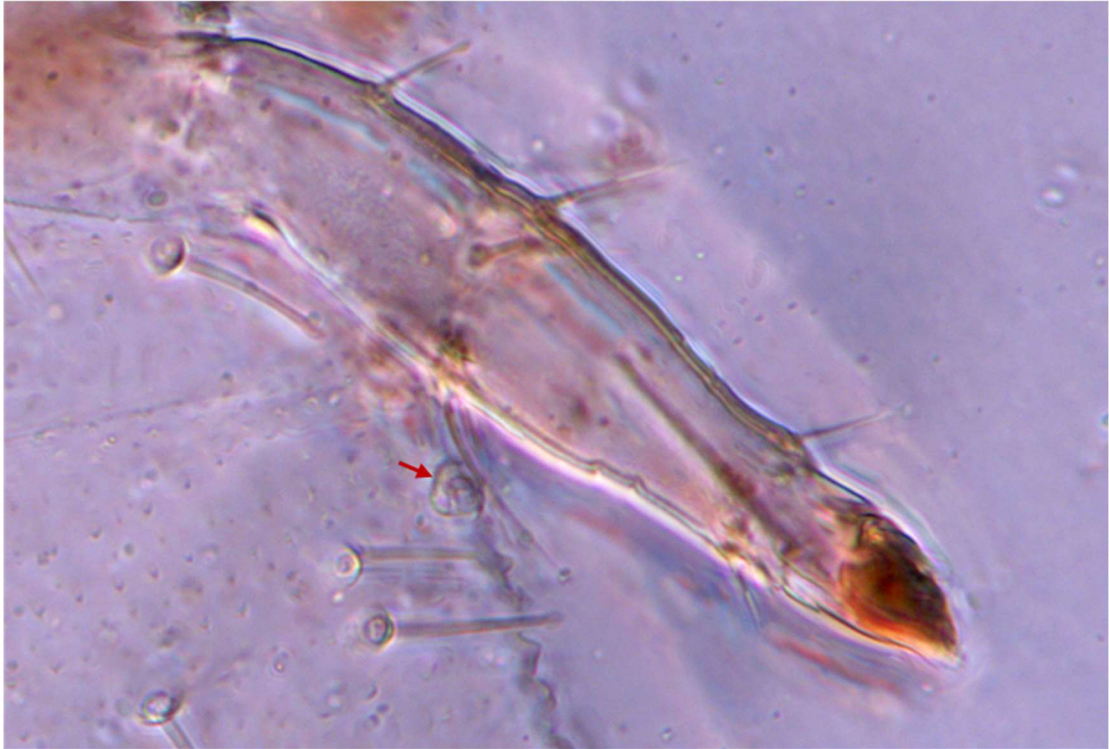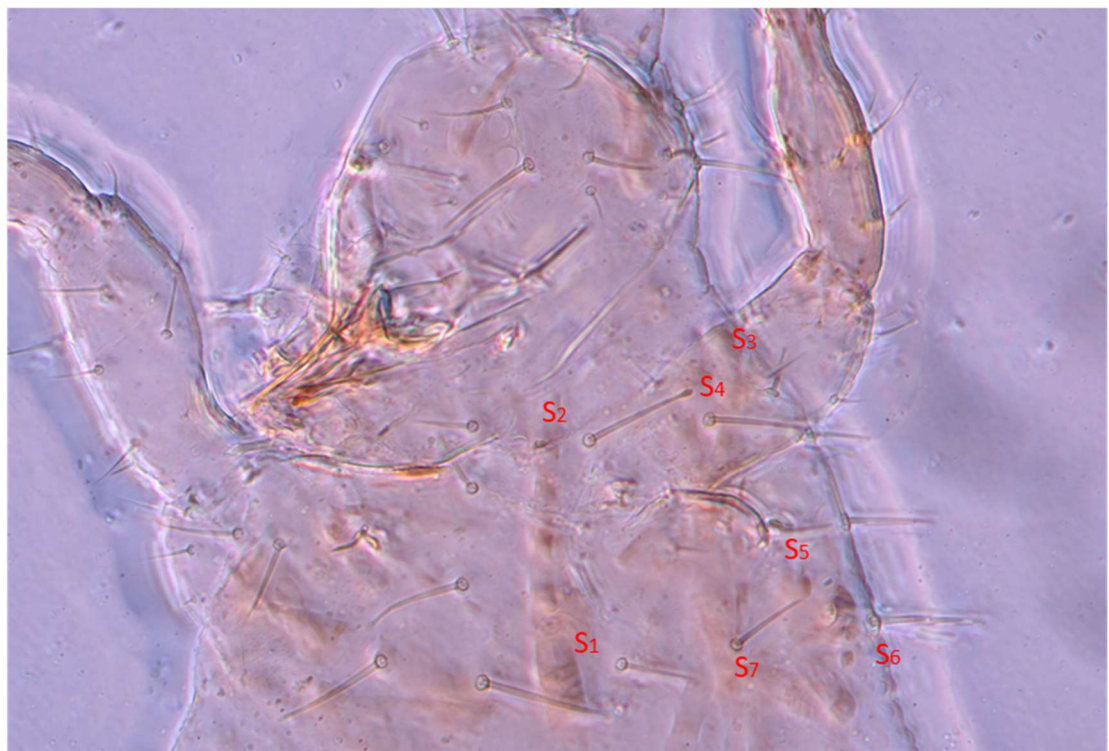
Bibliographical resources for thrips identification

Three publications are fundamental when identifying intercepted thrips in Europa. They largely contribute to the content of the present document. First, Mound & Kibby (1998) is a practical handbook dedicated to the identification of adult pest thrips. Some extracts are provided in Appendix. Second, Vierbergen *et al.* 2010 is indispensable for the identification of Thripidae larvae. And third, zur Strassen (2003) is an exhaustive key for the Terebrantia in Europe and neighboring countries.

Many resources are otherwise available besides these cornerstones. A list of them is, devoted to the identification of Terebrantia, is presented in table 6. They are sorted by taxonomical then geographical coverages, and the references are detailed in the "identification keys" section of the References chapter.

Finally, a serial of online matrix keys are available on the Lucid website[2]. The keys have been built for the identification of Thysanoptera of a given region (Thysanoptera Britannica, Thrips of California…) or thrips of economic importance (Pest thrips of the world…). They also provide useful illustrations and detailed descriptions of the included species.

Table 6. Publications for the identification of Terebrantia, sorted taxonomical and geographical coverages (last updated October 2022).

Family	Subfamily	Coverage	Geographical area	Reference
Merothripidae		Phylogeny	World	Pereyra 2011
		Key to genera & species	World	Mound & O'Neill 1974
Melanthripidae		Phylogeny, key to genera	World	de Borbon 2009
		Dorythrips species	World	de Borbon 2009
Aeolothripidae		All species	Japan	Masumoto 2019
		All species	Australia	Mound 1967
		Aeolothrips species	North America	Bailey 1951
		Aeolothrips species	Iran	Alavi & Minaei 2018
		Dactuliothrips species	World	Pereyra & de Borbon 2013
		Desmothrips species	World	Pereyra & Mound 2010
		Erythrothrips species	World	Mound & Marullo 1993
		Franklinothrips species	World	Mound & Reynaud 2005
Fauriellidae		Key to genera & species	World	Mound *et al.* 1980
		Key to genera	World	Mound 1998
Stenurothripidae		*Holarthrothrips* species	World	Bhatti 1986
Heterothripidae		*Heterothrips* species	North America	Bailey & Cott 1955
		Heterothrips species	Brazil	Pereyra & Cavalleri 2012
		Heterothrips species	Argentina	de Borbon 2010
		Heterothrips species	Central America	Retana-Salazar 2009
Thripidae	-	Key to genera	Argentina	de Borbon 2009
	Panchaeto-thripinae	Key to genera & species	World	Wilson 1975
		Key to genera & species	China	Mirab-Balou *et al.* 2017
	Dendro-thripinae	Key to genera	World	Mound & Tree 2016
		Key to genera & species	Australia	Mound 2016
		Key to genera & species	Iran	Alavi *et al.* 2014

[2] https://www.lucidcentral.org/editors-pick-animal-and-plant-identification-keys/thrips-keys/

	Asprothrips species	China	Tong et al. 2016
	Dendrothrips species	World	zur Strassen 1968
	Dendrothrips species	India	Bhatti 1971
	Dendrothrips species	Japan	Noguchi & Masumoto 2019
	Pseudodendrothrips species	World	Mound & Tree 2007
Serico-thripinae	Phylogeny	World	Lima & Mound 2016
	Key to genera & species	South America	Lima & Mound 2016
	Key to genera & species	Australia	Mound & Tree 2009
	Hydatothrips species	East & South Asia	Wang 2007
	Hydatothrips species	China	Mirab-Balou et al. 2013b
	Hydatothrips species	East & South Asia	Wang 2007
	Hydatothrips species	India	Bhatti 1973
	Neoydatothrips species	Iran	Minaei 2016
	Neoydatothrips species	India	Tyagi & Kumar 2016
	Neoydatothrips species	Japan	Masumoto & Minoura 2014
	Neoydatothrips species	China	Mirab-Balou et al. 2013b
	Neoydatothrips species	East & South Asia	Wang 2007
Thripinae	Key to genera	SE Asia	Mound & Ng 2009
	Key to genera	Iran	Mirab-Balou et al. 2013a
	Quarantine species	Japan	Matsumoto 2010
	Phylogeny		Bhatti 1978
	Anaphothrips gen-group genera	World	Masumoto & Okajima 2017
	Anaphothrips gen-group genera & species	Australia	Mound & Masumoto 2009
	Anaphothrips gen-group genera & species	Iran	Mirab-Balou et al. 2012
	Anaphothrips gen-group genera & species	Central America	Retana-Salazar 2007
	Anaphothrips species	China	Cui et al. 2017
	Anaphothrips species	Iran	Mirab-Balou et al. 2014a
	Anaphothrips species	North America	Nakahara & Footit 1995
	Anaphothrips species	Japan	Kudo 1989
	Anaphothrips species	Australia	Pitkin 1978
	Anascirtothrips species	Japan	Masumoto & Okajima 2007
	Ceratothripoides species	World	Mound & Nickle 2009
	Chaetanaphothrips species	World	Pitkin 1977
	Chaetanaphothrips species (part)	Asia	Nonaka & Okajima 1992
	Chaetanaphothrips species	Japan	Kudo 1985
	Chirothrips gen-group species	North America	Nakahara 2012
	Chirothrips gen-group revision	World	Bhatti 1990
	Chirothrips species	World	Zur Strassen 1960
	Chirothrips species	Iran	Minaei & Mound 2010
	Chirothrips species	Australia	Mound & Palmer 1972
	Chirothrips species	North America	Andre 1939
	Ctenothrips species	World	Xie et al. 2011
	Exothrips species	World	Bhatti 1975

Frankliniella gen-group species	China	Mirab-Balou *et al.* 2014b
Frankliniella tritici spp-group	Central America	Retana-Salazar *et al.* 2014
Frankliniella minuta spp-group	Central America	Retana-Salazar *et al.* 2010
Frankliniella curiosa spp-group	Mexico	Johansen 1998
Frankliniella minuta spp-group	Americas	Sakimura & O'Neill 1979
Frankliniella species	Peru	Ortiz 1978
Frankliniella species	Argentina & Chile	de Borbon & Ines Zamar 2018
Frankliniella species	Brazil	Cavalleri & Mound 2012
Frankliniella species	Brazil	Lima & Myasato 2017
Frankliniella intercepted species	World	Nickle 2004
Frankliniella intercepted species	World	Vierbergen 1995
Megalurothrips gen-group species	Iran	Mirab-Balou & Chen 2011
Megalurothrips species	World	Palmer 1987
Odontothripiella species	Australia	Pitkin 1972
Odonthothrips species	France	Bournier 1990
Odonthothrips species	China	Dang *et al.* 2010
Odonthothrips species	World	Pitkin 1972
Pezothrips species	World	Masumoto & Okajima 2020
Rhamphothrips genus-group genera	World	Bhatti & Mound 1992
Rhamphothrips revision	World	Bhatti 1978
Scirtothrips genus-group genera	World	Ng & Mound 2015
Scirtothrips species	Malaysia	Ng *et al.* 2014
Scirtothrips species	Africa	Mound & Stiller 2011
Scirtothrips species	Japan	Masumoto & Okajima 2019
Scirtothrips species	Australia	Hoddle & Mound 2003
Scirtothrips pest species	World	Mound & Palmer 1981
Taeniothrips species	World	Mound *et al.* 2012
Thrips species	Brazil	Lima *et al.* 2018
Thrips species	Japan	Masumoto 2019
Thrips species	China	Zhang *et al.* 2011
Thrips species	Afrotropics	Mound 2010
Thrips species	Malaysia	Mound & Azidah 2009
Tenchaetothrips species	India	Bhatti 1982
Trichromothrips species	Japan	Masumoto & Okajima 2005

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