

2. Order Acari - General introduction and key to the major groups

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Diagnosis

Small to minute in size (body length in general 0.2-2.0 mm, in extreme cases 0.1-30 mm). The ancestral arachnid tagmata, the prosoma and the opisthosoma, not apparent as main body divisions (2-1). Opisthosoma completely fused to prosoma; body secondarily divided into: (1) the gnathosoma bearing as appendages the chelicerae and palps, formed in different kinds by the palp coxae and further sclerotized elements of the palp segment; (2) the idiosoma formed by fusion of the leg-bearing region (called podosoma) and the opisthosoma. Often, leg-bearing segments grouped in two parts, with a large area (transversal furrow) separating the segments of legs I and II (propodosoma) from those of legs III and IV (metapodosoma). If metapodosoma and opisthosoma form a functional unit, then the proterosoma (gnathosoma + propodosoma) separated from the hysterosoma (metapodosoma + opisthosoma), thus forming a third level of body subdivision. Genital opening in anterior part of opisthosoma, anal opening in posterior part, both ventrally. Respiratory system evolved in different ways within the main taxa. Postembryonal development plesiotypically including five free-living stages (plus the non-hatching prelarva): larva with three pairs of legs only (2-3), three nymphal stages (2-2, 2-4 *g-h*) and the adult (2-4 *a-f*) with the typical arachnid pattern of four pairs of walking legs. Developmental stages relinquished or realized under particular conditions; e.g. in *Parasitengona* two of the nymphal stages transformed into pupa-like resting stages without appendages (2-2).

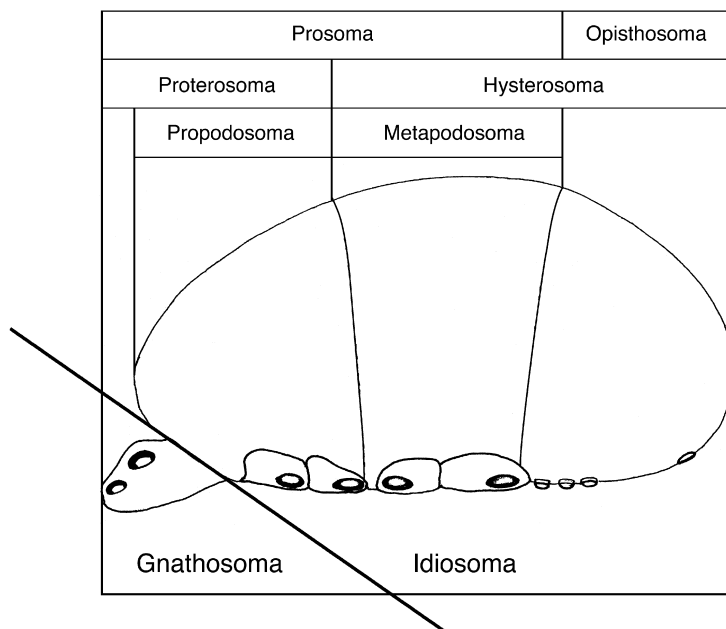


Fig. 2-1: Mite ground plan and basic terminology - for details see chapters „Diagnosis“ and „General introduction“.

Introduction

Like other Arachnida, mites are primarily terrestrial, and therefore aquatic mite taxa are derivatives from terrestrial ancestors. The small body size, name-giving for this group of animals (Greek: το ακαρί = the mite, from ακαρίαῖος = tiny), developed in several groups to an extreme extent. Species so small may appear „terrestrial“ only at a first glance: often they are bound to elevated moisture and important steps in mite phylogeny may have taken place in hygric microhabitats. Mites demonstrate a range of morphological and physiological adaptations not found in other orders of Chelicerata. They can be herbivores, carnivores or parasites; they disperse by active migration, passive transport or phoresy, in or without combination with parasitism; they reproduce bisexually or parthenogenetically, and they have radiated successfully in both terrestrial and aquatic habitats.

The suborder Anactinotrichida, with the Opilioacarida, Holothyrida, Gamasida (2-4 *a*) and Ixodida (ticks, 2-3 *a*), only includes a few hygrophilic species, mainly in the Gamasida, but not one aquatic taxon. In contrast, in the other suborder, the Actinotrichida, many different clades include hygrophilous species regularly found in aquatic fauna samples, and adaptations to submerged life evolved several times independently. An exploitation of freshwater and marine biotopes, with subsequent adaptive radiation, took place only in two clades: the Halacaroida, a morphologically homogenous monotypical superfamily with ecological gravitation in marine habitats, and the Hydrachnidia, a subtaxon of the phalanx Parasitengona including about 50 families whose representatives are restricted mostly to freshwater habitats. Both, halacarid mites and hydrachnid mites developed similar morphological and physiological adaptations to a submerged life (e.g. closed spiracles) but display different strategies in their life cycles: halacarid mites are quite homogeneous in morphology and particularly minute in size, and their eggs and early stages can be distributed passively both by water currents and attached to larger animals. Some, or all stages in the life cycle of their freshwater representatives are capable of colonizing extreme aquatic habitats such as high mountain pools with extreme changes in water level. Here they coexist with microcrustaceans, nematodes, oligochaetes, flight active insects, and hygrophilous representatives of „terrestrial“ mite groups, „true freshwater mites“ (Hydrachnidia) being absent. A prerequisite for the evolutionary success of the Hydrachnidia was probably the presence of a parasitic-phoretic stage in the life cycle of their stem group, the Parasitengona. Parasitism on flying insects enable the Hydrachnidia to migrate horizontally and thus to compensate for the disappearance of habitats due to summer drought or due to changes over a longer time scale. This is obviously necessary for long-term colonization success of inland waters.

Due to the particular radiation of these two mite groups (resulting in about 1000 known species of Halacaroida and 5000 of Hydrachnidia), the presence of representatives of other mite taxa in aquatic habitats has often been neglected. However, in many habitats such as mountain springs, temporary lowland pools or the hyporheic interstitial, species of Trombididae, Oribatida or Acaridida may be found to be dominant in terms of population density. Many of them display well developed adaptations to a submerged life, sometimes during all life stages.

Morphology and terminology

The basic morphological ground plan of the mite body is explained in Fig. 2-1. Several systems of morphological terminology have developed in terrestrial and aquatic acarological research, particularly in North American, West European and East European scientific traditions. Therefore, a detailed explanation of the morphological terminology and a

specific glossary for each of the mite groups treated in this work are provided in the appropriate chapters.

Sampling and sample processing

Collecting

Due to the minute size of mites, they require a special treatment both during field work and in the laboratory (e.g., BARR 1973, SMITH & COOK 1991, for methodological discussion with particular regard to spring habitats, see GERECKE & DI SABATINO in press). If standard techniques such as Surber sampling, dredging, hand netting or drift sampling are applied (for details see SCHWOERBEL 1994), adult Hydrachnidia can be collected adequately only if the mesh size does not exceed 250 μm . For collecting their larval stages as well as the smaller representatives of mite groups such as halacarids, a mesh size of 100 μm is necessary. Many limnological sampling methods have, however, two major disadvantages: first, they do not adequately represent species with a patchy distribution - species adapted to particular life conditions, accumulating in particular microhabitats at the edge of water bodies, may be easily overlooked or underestimated. Second, especially many species of Hydrachnidia as specialized predators and/or parasites, may be present in population densities which are lower by an order of magnitude compared with other elements of the meiofauna or macrofauna (GERECKE et al. 2005). Consequently, such species, often characteristic elements of the studied habitat, are hardly represented in collections applying area- or time-limited standard methods. For a representative investigation of the acarological diversity, intensive hand-netting is obligatory in all microhabitats. The sediment should be dispersed locally, tufts of plant material vigorously shaken and the material washed and filtered through a net. This method does not allow for quantification, but differences in the relative abundance between the species can be studied.

Since most Hydrachnidia (like many limnic insects) are aerial during particular phases, the use of Malaise traps and sweep nets helps to estimate population densities and both instruments are indispensable tools to investigate population development and phenology. Moreover, soil probing and hand collecting during the dry phases of temporary waters allows the detection of resistant stages and the estimation of survival strategies in such habitats. In the following, we give a more detailed survey on sampling techniques and their application in various habitat types:

Lentic habitats:

1. Dip-nets are often used by hand for sampling in the littoral vegetation, but this technique is also useful in scuba diving in vegetation deeper than 1-2 m.
2. Frame-nets operate like a dip-net but have a square, round or triangular opening. Collecting is accomplished by pulling a frame-net for a certain distance through the sediment to a depth of 2.0-2.5 cm. Each sample taken represents a certain area. Samples are passed through a sieve and are further sorted by eye (TEN WINKEL 1987). Vertical haul: nets are lowered to the bottom and hauled vertically (BAGGE et al. 1996).
3. Grabs are suitable for estimating the density of the organisms living on the bottom. Organisms swimming close to the bottom will also be collected (K. VIETS 1930).
4. Underwater traps can attract mites to a light source or to some kind of bait. Light traps may work on the base of electric (PIECZYNSKI 1962, 1969; BAGGE et al. 1996) or chemoluminescent light sources (BARR 1979a, see there for more practical details) placed in the centre of a polyethylene container with a wire-basket like entrance. Many Hydrachnidia are attracted in particular to yellow-orange light. However, the technique works selectively: a few species, e.g. *Hygrobatas nigromaculatus* and *H. trigonicus*, show no positive

phototaxis (DAVIDS et al. 1994). CONROY (1973) proposed an underwater trap working with bait instead of a light source.

Lotic habitats:

1. Extracting and washing of submerged substrata (plants, detritus, stones). The material is at best transferred under water into a net and then put into a white tray filled up to a few cm of clear water. Regular collection of a constant amount of determinate material (e.g. a certain volume of mosses) may provide quantitative indications (BADER 1977). Larger, immovable structures such as trunks of trees and large rocks are brushed *in situ* and the material collected in a hand net.

2. Kick sampling: In a gravelly stream habitat a net is held vertically in the stream, with the mouth opening upstream. The collector stands upstream from the net and vigorously disturbs the substratum to a depth of 15-20 cm, so that detritus and animals are washed into the net. The net is then withdrawn from the water and the sediment collected distributed over the bottom of a white tray (BARR 1973).

4. Additional species with a patchy distribution may be detected by drift netting. In contrast to other stream invertebrates, Hydrachnidia drift with maximum abundance in daylight (SCHMIDT 1969).

5. DEWEZ & WAUTHY (1984) worked with an artificial substratum kept submerged for at least eight weeks. At the end of the experiment, mites were extracted and counted.

Interstitial habitats:

1. The „Karaman-Chappuis“ digging is in most cases the technique best suitable for collecting the biocoenosis in the hyporheic interstitial (CHAPPUIS 1942). A hole is dug in the exposed gravels of a stream, deep enough to penetrate the water table. The interstitial water percolating into the hole is removed using a small bucket or bowl and washed through a net or sieves to eliminate turbidity.

2. The use of a so called „Bou Rouch“ pump is an often applied alternative (VIGNA TAGLIANTI et al. 1969; BOULTON et al. 1992). This technique has the advantage of being applicable in the absence of gravel banks, pumping the sample directly from the interstitial below the bed of the stream. However, in general, in small streams without sand or gravel banks, the sediments below the running water do not provide habitats suitable for colonization by hyporheobiont specialists. In general, as compared with digging, pumping produces lower numbers of specimens and species and a lower amount of hygrophilous ripicoles, but a higher percentage of species confined to hyporheic habitats (GERECKE et al. 2005).

Semiterrestrial habitats:

For quantitative collecting hygic populations of Acaridida, Oribatida and terrestrial Parasitengona, it is recommended that substrate (algae, moss cushions, detritus) be brought into the laboratory. Mites can be easily extracted from the substrate using a heat-driven dynamic extraction method such as a Berlese- or MacFadyen-apparatus, or the mites can be sorted from the samples directly. However, these methods work selectively and are not suitable for extracting halacarids.

Treatment of samples

Due to their bright colours and/or active behaviour, most deutonymphs and adults of the Parasitengona may be sorted on the spot from the collected material if light is favourable and a flat white pan or tray is used. Mites become active a short time after the sample is poured into the pan and can easily be sorted by eye and picked up with a pipette. Samples with much detritus and silt should be washed carefully through a set of two sieves. By stir-

ring clean water through the upper sieve (mesh size about 1.5 mm.), the mites will accumulate in the lower sieve (mesh size 100 μ m). Typical lotic species do not move actively when the sample is removed from the habitat. However, under the influence of rising temperature and decreasing oxygen availability, Hydrachnidia leave the collected substrata to swim or crawl in the open water. Mites living in moss growing at the air-water interface or in cascades often cling tightly. To extract them, a clump of moss is picked apart in a tray of water, strand by strand, with a pair of forceps, carefully searching for mites, or the sample is stirred vigorously until the mites become detached. Sorting in the field is of particular importance if the remaining sample is to be preserved in a fixative such as formalin or alcohol (see below). However, not only representatives of the Halacaridae, Acaridida and many groups of Oribatida which are small in size and less agile, but also larvae and small nymphs of Parasitengona can be sorted only in the laboratory under a stereo microscope, preferably from the living material. Living samples are best transported at a low and stable temperature, bathed by, but not submerged in water (in order to avoid mechanical damage), and in contact with a large volume of air for oxygenation. Most mites are rather resistant against many kinds of stress during sampling and transport and are found still alive in the sample when many of the other invertebrates have died. Also in the laboratory, sorting should be processed through stacked sieves, the top one with a mesh size of 1000 μ m, and the bottom one with 60–100 μ m, being rinsed with a strong water jet.

In the laboratory, mites can be extracted from bottom mud by placing the sample for 10 minutes in a concentrated magnesium sulphate solution. The sample is stirred occasionally to allow animals to float to the top and the floating mites are subsequently decanted (EFFORD 1965). Further flotation methods, which make use of the specific weight of the animals work with solutions of sodium chloride, sugar or silicate. PROCTOR (2001) got good results with a kerosene-flotation technique. FAIRCHILD et al. (1987) were successful in extracting Hydrachnidia from moss samples by inducing them to respond to a temperature gradient. Preserved samples with little detritus can be stained with Bengal red.

Classification of undissected material

The mastery of dissection and slide mounting techniques (see below) is an important prerequisite not only for the inexperienced student. Confronted with material from little known habitat types or unstudied geographical areas, even a specialist who is an old hand at classifying mites is obliged to produce series of preparations in order to investigate diagnostic characters and their variability range. However, once a certain experience and familiarity with a local fauna is built up, most of the recorded mites may be recognized without preparative work. For instance, large parts of collections from habitats regularly investigated in the course of long term monitoring projects may be sorted under the stereo-microscope to species level. For the solution of problems requiring stronger magnifications (recognition of sexes, separation of similar sister species) it is in general sufficient to examine selected character states under the light microscope, temporarily placing the undissected mites on a concavity slide or, in order to bring the specimen into a particular position, on a slide prepared by the application of a viscous medium such as HOYER'S fluid (see below). In this manner classifying mites from aquatic habitats may become as routine as the study of other groups of benthic invertebrates.

However, specimens which are conspicuous e.g. due to particular dimensions, coloration patterns or aberrant characters merit special attention and should be dissected and slide mounted. Water mites tend to form monstrosities, often probably due to developmental disturbance at the proto- and tritonymphal resting stages, but there are also very rare, often enigmatic species of particular faunistic interest which appear only on rare occasions. Furthermore, selected specimens of the more problematic species should be regularly slide

mounted at regular intervals in order to control the correct taxonomic attribution. In any case, slides as well as material classified without slide mounting should be carefully labelled and conserved for later reinvestigations. For long term preservation in vials, both KOENIKE'S fluid (see below) or 96 % glycerine are suitable media (BARR 1973).

Rearing

The most satisfactory method of obtaining larval material for study is from eggs laid by identified females in the laboratory. Depending on the scope of the study, mites are isolated in couples or as single females in small vials or petri dishes together with substrata suitable for spermatophore and/or egg deposition (e.g. plant stems or pieces of filter paper). Temperature is best at a stable level similar to that of the species' natural habitat. As most species are adapted to starvation, in most cases feeding is not necessary for successful reproduction, at least if females are ovigerous at the start of the experiment. However, eggs should be kept separate from parents in order to avoid feeding by parents on their progeny. If the whole life cycle of a parasitengone species is to be studied, potential hosts are exposed to the freshly hatched mite larvae in small aquaterraria. Completing a parasitic phase demands much ingenuity and effort, no general rules can be given. For the resolution of questions concerning the host range of a selected species, it is recommended to interrupt the experiment when larvae have attached to their hosts and prepare them on microscopical slides (MARTIN 2000, 2003). A rather simple method for investigating parasite-host relations under natural conditions is the exposure of sediments which include both a mite population and their presumptive hosts in plastic bags in the field (GERECKE & MARTIN 2006). However a good knowledge of the fauna of the site is necessary because attribution of larvae and adults cannot be made with 100 % certainty. Bibliography concerning the wide field of specific requirements for successful reproduction is cited in the special part.

Preservation

Mites of the „terrestrial“ groups (acaridids, oribatids, terrestrial Parasitengona), also halacarids can be preserved in ethanol (75 %). Some drops of glycerine may be added to prevent the sample from drying out if the ethanol accidentally evaporates (see KRANTZ 1978). Formalin should be avoided, at least the animals should be transferred to ethanol as quickly as possible. For histological studies animals should be preserved with glutar aldehyde or Bouin's solution and subsequently be transferred to ethanol. If a sample dries up completely, water with a drop of washing-up liquid or some vinegar should be added. After soaking, the mites are transferred to ethanol. For Hydrachnidia required for dissection and light microscope studies, preservatives containing formalin or alcohol, are particularly disadvantageous. These substances fix the tissues in such a way that they cannot be properly cleared in either basic or acid corrosives. In general, clearing with lactic acid leads to acceptable results in all mite groups.

In the case of strongly sclerotized species like many oribatids, a preceding exposure to hot (90 °C) 10 % KOH is advantageous; combination of KOH followed by lactic acid may also allow clearing of mites preserved in formalin or alcohol. Water mite workers traditionally use as a preservative a glycerine - acetic acid - water mixture commonly referred to as „Koenike's fluid“. In the first days following fixation, this liquid may cause a shrinking of the animal and gas bubbles may fill parts of its body. A few days later, the animal extends again, the gas disappears and it becomes clear due to tissue breakdown. At the end of this process, all important characters of the skin as well as the sclerotized parts are clearly visible.

Molecular aspects which have gained increasing importance during the past years can be studied best on specimens preserved in special buffer solutions. Indications that molecular

DNA studies are possible also with material preserved in Koenike's fluid are based on a very small sample size and gave worse results compared with ethanol-fixed material (REY et al. 2004).

Contact with any type of preservative nearly always causes the loss of most of the colouration, namely in Parasitengona where most pigments are distributed in the internal organs or in lipids below the cuticle. Only colours located in, or originating from, sclerotized structures and some basic contrast patterns (e.g., the whitish contents of the excretory organ) may remain visible after years of preservation. Therefore, it is useful to document the colouration of interesting specimens before they are transferred to the preservative. A little studied, but promising alternative could be freeze-drying. However, specimens treated in this manner are mostly useless for further detailed microscopical investigation.

For the tiny Acaridida with their delicate cuticular structures only two methods of treatment are recommended: if the mites are alive, the specimens should be mounted directly in Hoyer's fluid; in doing so the mites stretch their legs and the natural shape can be preserved. In order to avoid compression of large specimens the coverslip should be supported at the edges by wax or modelling clay. For a more permanent preparation, after drying the coverslip should be ringed with Glyceel (BDH Chemicals Ltd, Poole, England) or Glyptal (not available in Europe, see TRAVIS 1968). If mites are not subject to immediate slide preparation they should be stored in ethanol (at least 70 %). Clearing agents are not necessary and should not be applied.

In contrast, many Oribatida have a dark and poorly transparent cuticle. Lactic acid (90 %) can be used to clear the body of the mites by maceration of the inner organs, which allows better examination of the cuticular characters. Temporary mounts in a cavity slide filled with glycerine or lactic acid (under a coverslip) is preferred. If the coverslip covers only a third or half of the cavity, the mite can be manipulated using mounted fine needles. Such a preparation allows the specimens to be turned and thus be observed from all perspectives.

Halacarids, again a group of rather tiny and often poorly sclerotized mites, can often be slide mounted without preceding treatment. Dark specimens may be cleared in pepsin or lactic acid for some minutes, exceptionally up to three days. If the body content of a specimen is very hard, it helps to apply heat up to 50 °C or to add acetic acid. If the animals are very fragile or the integument cleared too strongly, they can be stained with chlorazol black for microscopic examination.

Dissection

As mentioned above, dissection and preparation are not obligatory steps if the study of water mites is included into large scale limnological work. However, they are important for the documentation of little known or newly detected species and during studies on population variability. The dissection of body parts is best done in a drop of glycerol or KOENIKE'S fluid on a slide with a central cavity under a stereo microscope. Tools such as hooked minuten pins, fine needles (insect pins number ≥ 1) and micro-scalpels can be used. First the gnathosoma and the legs of one side are cut off with a sharpened needle and removed to the edge of the drop in such a way that the drop becomes enlarged and the objects are lying in a semicircle, so the legs cannot be mistaken one for the other. The body content is then gently squeezed out with a blunt needle. If the body content is too dark to allow transmission of light, or if there are important details to observe both on the dorsal and ventral surface, it is necessary to separate the dorsum from the venter. The mite is laid down on its side and cut with a micro scalpel into a ventral and a dorsal part. Next, the body contents can be removed. If the specimen has been preserved in Koenike's fluid it will be very easy to remove all soft parts leaving only the exoskeleton.

If the body parts need further clearing, e.g. in alcohol-preserved specimens, this can be done in lactic acid: the objects are placed in a drop of lactic acid on a corner of a slide and heated with a spirit flame. The flame must be moved along the edge of the drop till the lactic acid starts to smell. In this way the drop remains. A further possibility is the use of André's fluid (see below) which has the advantage that specimens may be left in it for several hours without damage to the integument. However, submersion of lightly sclerotized specimens for more than 48 hours tends to weaken everything, e.g. leg joints, and it obscures shields. A possible problem is that slightly sclerotized specimens may become so transparent that they are virtually impossible to find in the preparation dish. Another disadvantage is that any colour pattern disappears almost immediately. A basic corrosive, 10 % Potassium Hydroxide (KOH), is advised for use only with heavily sclerotized mites, KOH should be used at room temperature and the specimens left in the solution for only one to two hours (unsclerotized mites less than 15 minutes). A short contact with KOH does not destroy the colour pattern of the mite as is visible in preserved specimens.

Attention should be paid to the fact that representatives of different genera may need different treatment. For instance, in Hydrachnidia of the genus *Hygrobat* the gnathosoma is fused to the first coxae and cannot be isolated. Therefore, chelicerae can be removed only after cutting the idiosoma, pulling them out from inside the body. Species of *Eylais* can be determined best with the gnathosoma *in situ* (vs. visible from its ventral surface).

Mounting

The dissected parts of the mite should be mounted in a rather restricted area in the centre of the slide, so that they are easily found in the microscope preparation, at best following a stable scheme. If the idiosoma is dissected horizontally into two parts, dorsum and venter should be placed side by side, both with their external surface directed to the observer. If the idiosoma is not separated, the optimum position should be selected depending on the taxon in study. In many soft bodied species it is recommended to orientate the dorsal surface to the observer because in this way the often minute dorsal structures are visible, but also, by transparency, those of the venter. Detached legs are best located laterally from the idiosoma, the genital skeleton or the eggs posteriorly, the gnathosoma and its appendages anteriorly. It is important that both palps are orientated in the same direction to assure both lateral and medial views - naturally the same is true also if both legs of the same segment are dissected.

An overview of mounting media and their refraction indices for small arthropods can be found in KOOMEN & VON VAUPEL KLEIN (1995) and STOCK & VON VAUPEL KLEIN (1996). In recent years, in most acarological laboratories aqueous mounting media have replaced oil soluble resins such as balsam, primarily because specimens may be mounted without preliminary fixing operations as required for resin mounts. Furthermore, they have better optical properties. A disadvantage is that they are hygroscopic and often tend to liquefy or to dry up, depending on the humidity in the room where they are kept (UPTON 1993). The application of a sealant ring may slow down, but not completely interrupt, such processes.

Glycerine - jelly (a mixture of glycerine, gelatine, distilled water and a little carbolic acid) is probably the best medium for permanent preparations (K. VIETS 1923, 1936, HEVERS 1985). In museum collections, slides mounted in this medium have remained in good condition over many decades. The study objects retain shape without shrinking and also the fine integumental structures do not undergo alterations. The unique phenomena of deterioration frequently observed in older collection material are the forming of fine, dark droplets covering the body structures, or the precipitation of crystals. To avoid the potential risk of mustiness, preparations should be kept in a dry climate. An important advantage is

that mounts can be opened again by carefully heating, then cleaned and reorganized. Specimens preserved in an aqueous medium can be transferred directly to glycerine-jelly if it is liquefied by heating to about 50 °C. Prior to mounting cleared specimens the lactic acid or other clearing agents have to be removed. For each mounting session, only an adequate portion of glycerine jelly should be heated (heated too often, the medium becomes useless). A droplet is placed in the centre of a slide and extended to an area equal in surface to the coverslip. During the process of solidifying with lowering temperature, the dissected parts of the mite can be placed and orientated. Action may be taken on the cooling speed, e.g. placing the slide on artificial ice for accelerating, or keeping it warm if more time is needed for a complicated mounting procedure. However, after cooling down, the objects can be rearranged with warm needles. Air-bubbles and dust can be taken away with a micropipette. After hardening of the medium, the preparation is closed with a coverslip bearing a thin layer of liquefied medium on its bottom side. Excess of glycerine-jelly after placing the coverslip can be removed with a micropipette. It is important to keep the layer of mounting medium as thin as possible: a too large distance between the object and the coverslip would hamper the use of high microscope magnifications. An equally simple and effective suggestion was published by JÄNTSCH (2002 - for more details see there): mites are mounted on the coverslip instead of the slide. Due to their own weight, dissected parts sink down and are placed as close as possible to the future upper side. After hardening of the medium, the coverslip, still upside-down, is picked up with a thin drop of liquefied medium which hangs in the centre of a slide. When the preparation mounted in this way is kept upside down until completely hardened, gravitation will guarantee that the objects remain in the desired position. The mount can be sealed after half an hour with a rim of lacquer. Afterwards the slides must be labelled and should be stored in slide drawers or boxes so that the slides lie flat and, if possible, upside down.

A technique for making particularly perfect preparations and of particular aesthetic value, with the help of a turn-table is described by HEVERS (1985, see there for details).

Hoyer's Fluid is a medium that has become popular because it is not only very easy to use, but it has particularly suitable optical properties (KRAUS 1984). It is viscous and tends to harden by evaporation at room temperature; as in the case of glycerine-jelly, the objects may be transferred directly from water or Koenike's fluid. The medium has the capacity of clearing the mounted material as time goes by. After slide-making, preparations can be completely hardened in about 48 hours by heating to about 50 °C. The possible problem that during this heating turbulence may cause displacement of the objects can be avoided if one or two days before mounting, slides are prepared with flat droplets of the medium (GERECKE 1991). For mounting, only small quantities of additional liquid medium are added to the surface of these hardened droplets. Here, the objects may be arranged perfectly in the locally liquefied surroundings and will not change their orientation during the following short process of drying. These preparations can be sealed with a coverslip immediately or after hardening and a final control. Such preparations may be opened again very easily by adding water (acceleration by warming). Hoyer's fluid is recommended if large numbers of specimens are to be investigated in a short time. Furthermore, Hoyer's fluid has been used with good results for tiny water mite larvae. Living or preserved larvae are placed directly into the media and a cover slip added. However, this medium should not be used for mounting valuable material such as type specimens or representatives of rare and uncommon species. Its properties in long-term conservation are unknown. Both liquefying or complete drying up have been observed, often accompanied by deformation of the objects by compression and/or torsion.

Polyvinyl lactophenol, another medium easy to handle, should also not be used for producing type material or voucher specimens, because the integument of the objects softens and it is almost impossible to remount specimens without deforming the integument.

Glycerol (sealed with a wax ring) is a useful medium for short and medium term preparations.

BARR (1973) and COOK (1974) advise and describe a double coverslip method. The idiosoma of mites mounted between two coverslips can be observed from both sides and therefore this method is economic with dissection time. In order to reduce the risk of breaking or loss of information (the double coverslip may detach from its base), the „coverslip sandwich“ may be introduced into appropriate frames (for example aluminium frames).

Formulae:

	Koenike's fluid (modified)	Alternative mixture (BARR 1973, COOK 1974):
Glacial Acetic Acid	2 parts by volume	1 part
Glycerine (96 %)	5 parts " "	5 parts
Distilled water	3 parts " "	4 parts

To avoid strong maceration, e.g. for *Eylais* spp., the use of the alternative mixture is recommended.

André's fluid (Acetic Corrosive)

50 ml	Glacial Acetic Acid
50 g	Chloral Hydrate
50 ml	Distilled water

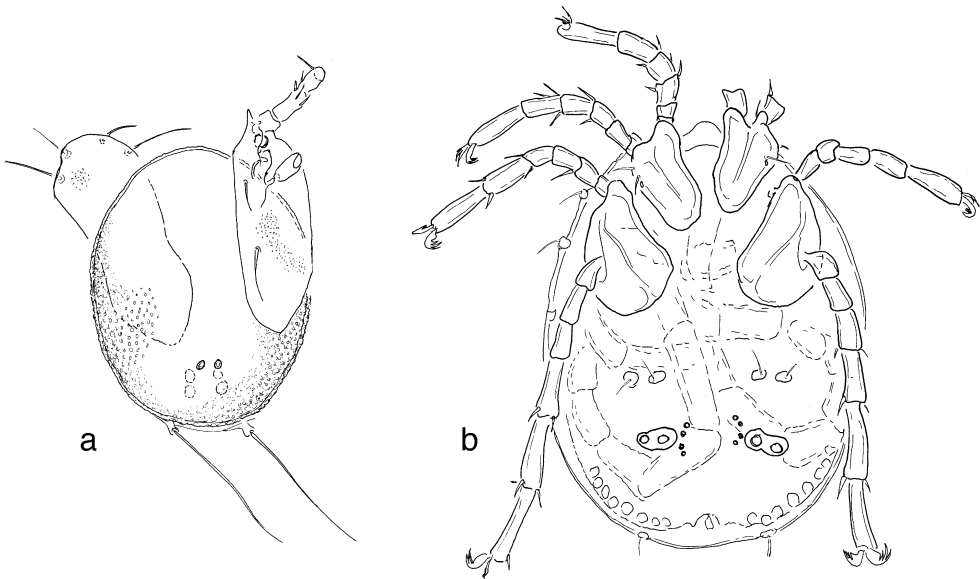


Fig. 2-2: Pupa-like resting stages of aturid mites (Hydrachnidia) in ventral view: a, protonymph, characterized by one pair of acetabula and detaching pieces of the larval exuvia (dorsal shield and left coxal plates, with the larval „ur stigma“ visible); b, tritonymph, characterized by four pairs of minute acetabula (in curved lines in the posterior part) and detaching pieces of the deutonymphal skin (coxae with leg remnants, „provisory genital organ“). Note the adult developing inside the nymphal integument, with folded legs and numerous acetabula arranged parallel to the posterior margin, typical for species of the subfamily Aturinae.

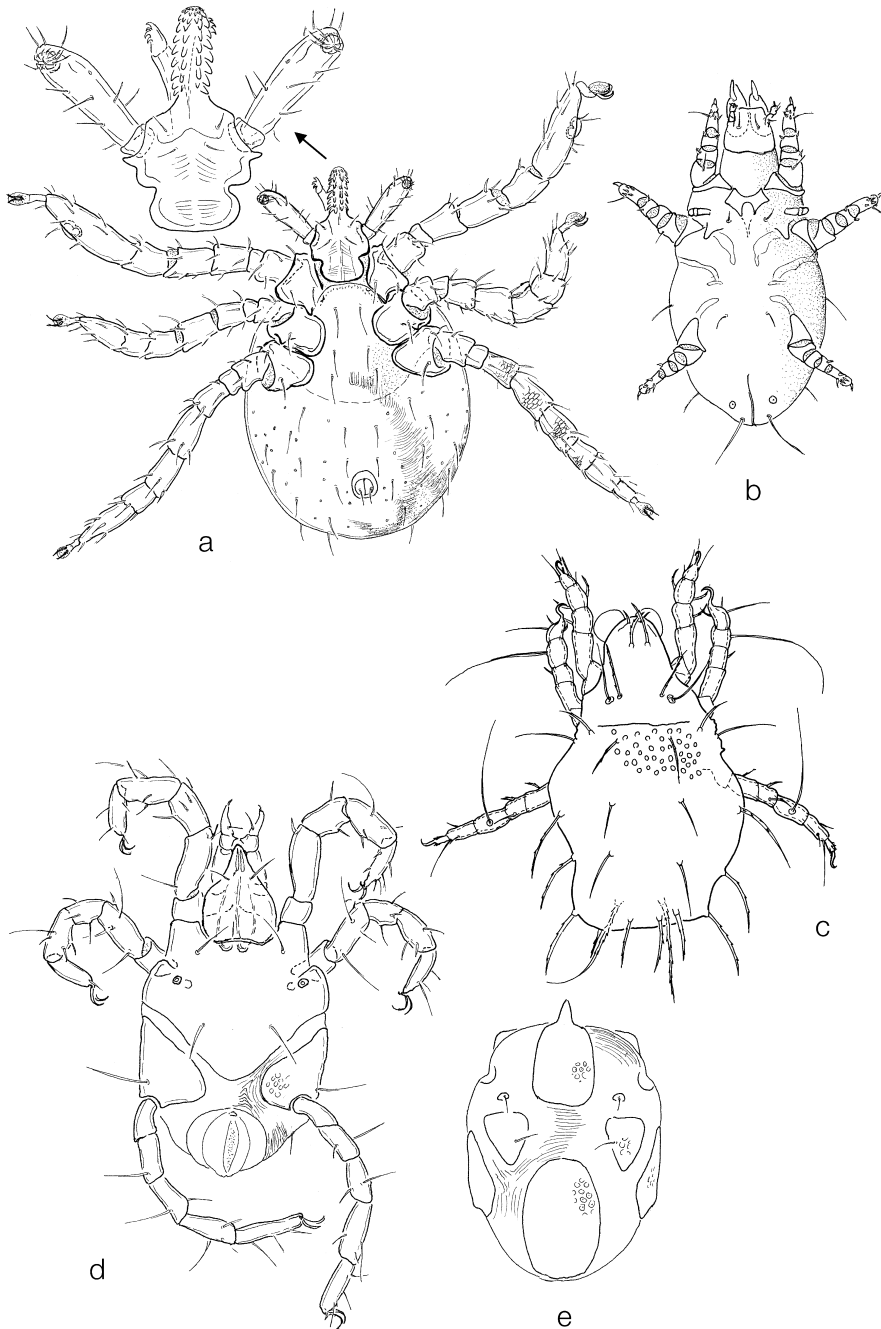
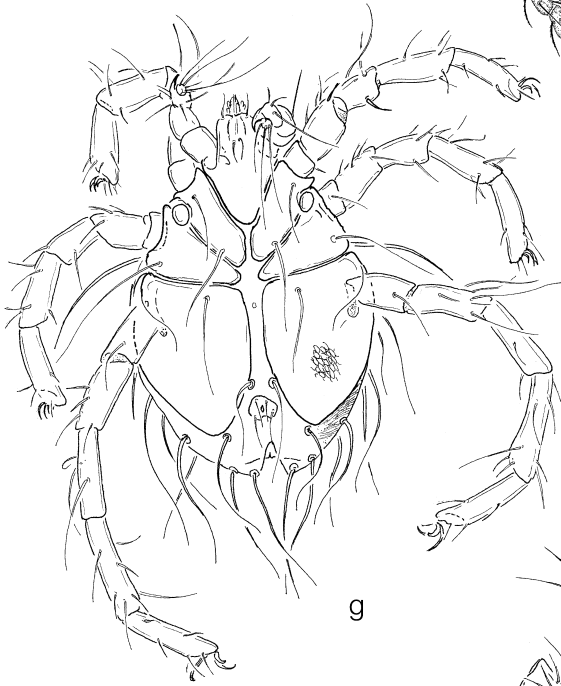
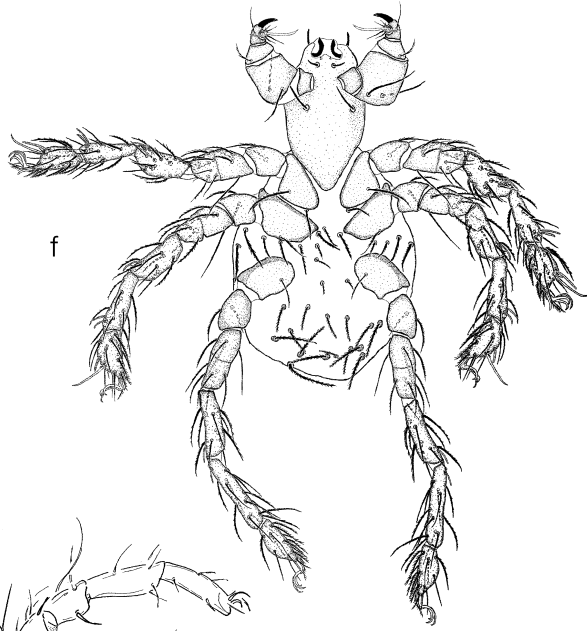


Fig. 2-3: Mite larvae (a-b ventral view): a, Ixodida; b, Acaridida (*Schwiebea* sp.); c, Oribatida, dorsal view (*Hydrozetes lemnae*); d-e, Halacarida (*Lobohalacarus weberi*); d, ventral, e, dorsal view (without legs); f, terrestrial Parasitengona (*Abrolophus quisquiliarius*); g, Hydrachnidia (*Sperchon squamosus*); h, Hydrachnidia, dorsal view of gnathosoma (*Stygothrombium* sp., only the basal leg segments given).



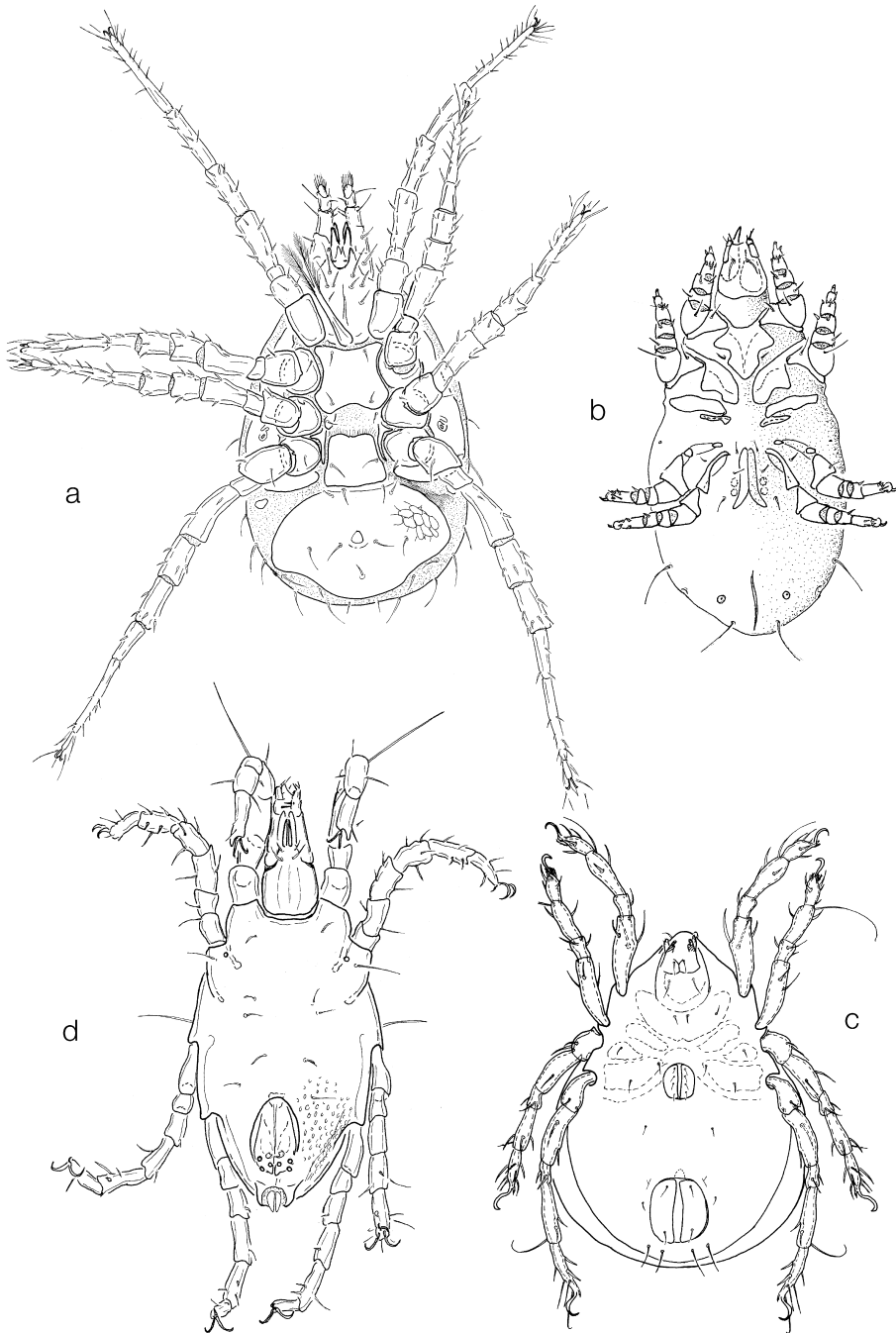
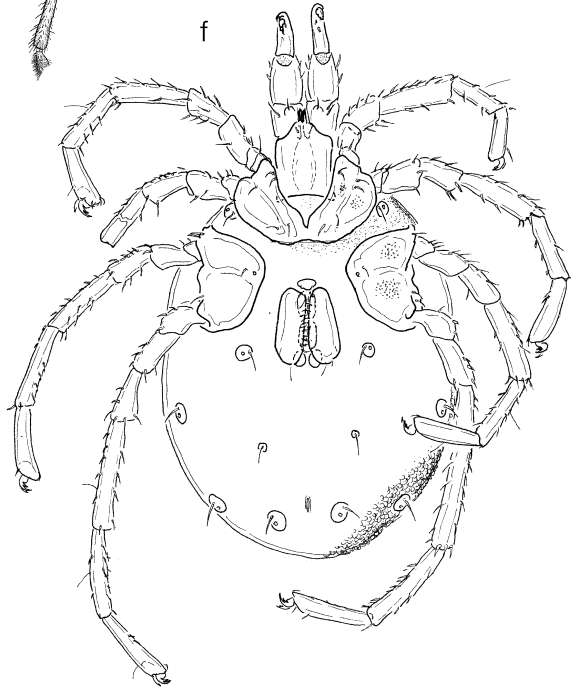
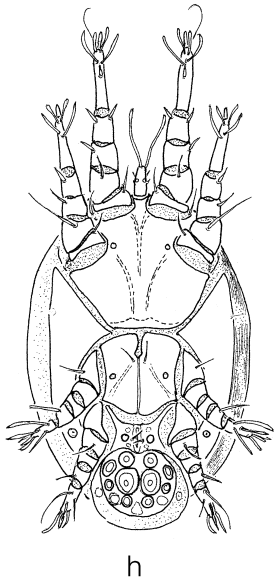
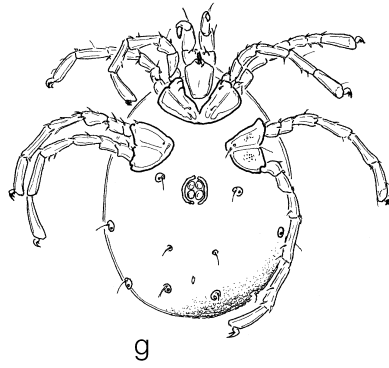
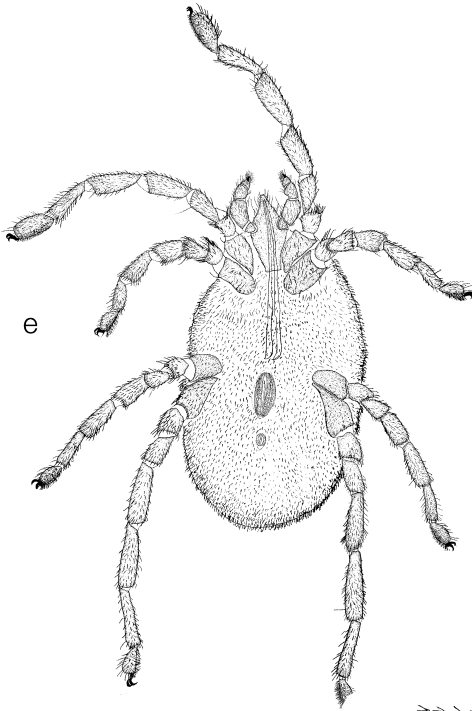


Fig. 2-4: Mites (a-f adults, ventral view): a, Gamasida; b, Astigmata (*Schwiebea* sp.); c, Oribatida (*Hydrozetes lemnae*); d, Halacaridae (*Lobohalacarus weberi*); e, terrestrial Parasitengona (*Abrolophus quisquiliarius*); f, Hydrachnidia (*Sperchon mutilus*); g-h, deutonymphs; g Hydrachnidia (*Sperchon mutilus* - given at the same magnification as adult in f); h, Acaridida (*Schwiebea* sp.).



Hoyer's fluid

50 g	Distilled water
30 g	Gum arabic, clear crystals
200 g	Chloral Hydrate
20 g	Glycerine

The ingredients should be mixed in the order listed above. Each substance should be fully dissolved and mixed before the next is added hence each step may take several days. The procedure is accelerated by heating in a water bath (KRAUS 1984).

Lundblad's Solution

To get rid of oil droplets prior to mounting in glycerine-jelly, LUNDBLAD used this solution:

112,5 g	Phenol
125 g	Chloral Hydrate
12.5 ml	Distilled water

Use for about 5 minutes. It is necessary to follow the process under the microscope because specimens and appendages become more-or-less colourless (work in a cavity slide).

The major groups of mites found in aquatic habitats

Key to life-cycle stages

- 1 Mouth opening and mouth parts, body sclerites, eyes and legs absent (but exuviae of appendages of the preceding stage may be attached to the outer surface; also, sclerotized structures and eyes of the next stage may be seen through the integument, 2-2); animals spherical or egg-shaped, covered by a thick, often sculptured integument; the only external organ is a smooth or slightly sclerotized field generally with one or two pairs of minute, cup-shaped osmoregulatory organs („acetabula“, absent in Erythraeioidea) located in the centre of the venter (future genital area); a longish zone with a smooth surface, may indicate the future mouth-part area („pharyngeal organ“).
 **resting proto- and tritonymphs of Parasitengona**
- Body with appendages, eyes often present; sclerites present or absent. 2
- 2 Three pairs of legs **larvae**
 („mite-larvae-like“ wingless insects are generally distinguished by the presence of articulated antennae, in particular cases, also by mouthparts transformed into a sucking apparatus, or by the presence of appendages on the ventral abdomen modified for jumping).
- Four pairs of legs. **active nymphs and adults**

Key to larvae

- 1 Mouth opening on an elongated projection ventrally densely covered by barbed hooks (2-3 a) or a bifurcated appendage („tritosternum“) located at the ventral base of the gnathosoma (as given in 2-4 a for an adult); coxae of legs movable, spherical or cylinder-shaped.
 **Ixodida and Gamasida**
 (terrestrial, accidentally in aquatic habitats, not treated in this work)
- Mouth opening not on an elongated projection densely covered by barbed hooks, no bifurcated appendage („tritosternum“) located at the ventral base of the gnathosoma; coxae of legs fused to the body surface in the form of coxal plates, not movable. 2
- 2 Palps with one or two movable segments only (2-3 b); one pair of rod-like Claparède organs (2-3 b) or ring-shaped osmoregulatory organs (3-34) between leg I and II; prodorsum never bearing trichobothria. **Acaridida** (page 38)
- Palps with more than two movable segments (e.g. 2-3 f, 2-3 g – but note that Oribatid larvae may have their palps retracted into their rostrum); osmoregulatory organs between leg I and II rod-like („Claparède organs“), cup-shaped (2-3 g) or invisible (2-3 f), generally paired, rarely supernumerary; trichobothria present or absent. 3

- 3 Gnathosoma (including palps and chelicerae) more or less covered by the rostrum of the prodorsum, not or hardly visible in dorsal view (2-3 *c*); prodorsum with or without trichobothria in dorsolateral position; anal opening in the form of a longitudinal slit; often species with a remarkable contrast in colour between the appendages (dark) and body (clear). **Oribatida** (page 89)
- Palps and chelicerae visible in dorsal view (2-3 *h*), not covered by a prodorsal rostrum, anal opening in the form of a round pore (2-3 *g*), a longitudinal slit (2-3 *d*), or absent (2-3 *f*); colour various. 4
- 4 Gnathosoma snout-like (cone-shaped, equally tapering from the base to the tip, as given in 2-5 *a* for an adult); palp tarsus distally with two conspicuous, long setae; palp tibia without claw; chelicerae elongate, distally with small movable digit; two pairs of trichobothria; active at terrestrial conditions. **Bdellidae** (**Eupodina**)
(several species present in semi-aquatic habitats, not treated in this work)
- Gnathosoma not snout-like as described above; palp tarsus various, but without two conspicuous, long distal setae; palp tibia with or without claw; chelicerae various; trichobothria present or absent 5
- 5 Idiosoma dorsally covered by one single hemispherical shield (as given in 2-5 *c* for an adult); empodium of legs branched into adhesive setules; palp tibia with claw; trichobothria absent from idiosoma, body red in colour, specimens often found floating on water surface. **Homocaligidae** (**Eleutherengona**)
(restricted to aquatic habitats, in Central Europe only *Homocaligus scapularis* (C.L.KOCH, 1838), not treated in this work)
- Dorsal shield, if present, plain or only little curved; empodium of legs claw-like or absent; palp tibia with or without claw; trichobothria present or absent. 6
- 6 Idiosoma with only three pairs of ventral setae (2-3 *d* - no anal setae) and at most seven pairs of dorsal setae; palp four-segmented; legs with five movable segments; free living. **Halacaroida** (page 113)
- Idiosoma with more than four pairs of ventral setae (2-3 *f-g* - at least one on each coxa and two pairs of anal setae) and more than seven pairs of dorsal setae; number of palp segments various, often five; legs with five or six movable segments; mostly parasitic on arthropods, but may be found during the preparasitic phase in high population densities in sediment samples. 7
- 7 Anal opening present or absent (2-3 *f*); without a sclerite or flanked by a pair of sclerites. Osmoregulatory organs never stalked, coxa II with one or more setae; legs I/II short or long (occasionally more than double the length of the idiosoma); number of setae on palp genu various: one (**Trombidioidea**), one to two (**Erythraeoidea**) or many (**Calypstomatidae**). **terrestrial and hygric Parasitengona** (page 158)
- Anal opening present, usually within an undivided sclerite (2-3 *g*); if flanked by paired sclerites (**Stygothrombidioidea**) then osmoregulatory organs stalked and reaching well beyond the lateral body sides; if anal sclerite absent (**Hydryphantoidea**: *Wandesia*) then coxa II without setae; length of legs I/II never more than double the length of the idiosoma; palp genu with two setae. **aquatic Parasitengona: Hydrachnidia** (page 241)

Key to nymphs and adults

In the following key, we use adult character states also found in the related nymphal stages. In general, besides the genital area being less developed and with the gonopore closed, nymphs differ from adults in their smaller size, lower numbers of setae on the appendages, lower numbers of osmoregulatory organs („acetabula“, in Acaridida called „genital papillae“ or „ring organs“) and relatively large areas of unsclerotized integument (compare 2-4 *f-g*).

- 1 Gnathosoma with minute palps consisting of one to two movable segments (2-4 *b*), or completely reduced; if a gnathosoma and mouth opening are absent, mouthparts replaced by a longish appendage („palposoma“, 3–13) with two long distal sensory setae („solenidia“) or by a group of sensory setae between the insertions of the first legs; furthermore, nymphs (3-13) with a reduced gnathosoma bear an adhesive organ or a clasping apparatus with cuticular plates ventrally at the posterior end; stigmata and trichobothria always absent; only one claw on the pretarsus; males ventrally with an external copulatory organ (aedeagus). **Acaridida** (page 38)

- Gnathosoma well developed, palps with three to five movable segments (2-4 *a*, *c-g*); ventral body surface without any adhesive organ or clasping apparatus; with or without stigmata and trichobothria; one or two claws on the pretarsus; males only in exceptional cases with an external copulatory organ. 2
- 2 Coxae of legs movable, spherical or cylindrical - thus, legs with seven movable segments (2-4 *a*); a pair of stigmata on sclerotized plates laterally near the third or fourth legs (2-4 *a*); leg pretarsus always with well developed paired claws and an unpaired membranous pulvillus; chelicera with well developed pincers. 3
- Coxae of legs fused to the ventral body area in the form of sclerotized plates on which the leg trochanters insert - thus, legs maximally with six movable segments (2-4 *c-g*); stigmata, if present, in the anterior area of the idiosoma or in the gnathosoma, often not clearly visible; leg claws and chelicera of variable shape. 4
- 3 Gnathosoma with an elongated extension (“hypostome”) on its ventral side densely covered by barbed hooks (as given in 2-3 *a* for the larva); stigmata on a porose plate laterally near coxa IV; palp tarsus without a fork-like seta. **Ixodida**
(ticks - occasionally found in aquatic environments frequented by cattle as drinking places; however, purely terrestrial, not treated in this work).
- Gnathosoma without a hooked, elongated stiletto-like extension; stigmata on a plate near the third legs and associated with a longish sclerite more or less extending anteriorly („peritreme“ = tracheal groove, 2-4 *a*); palp tarsus at the inner margin with a fork-like seta. . . . **Gamasida**
(several hygrophilous species regularly found in samples of riverine substrata from all kinds of inland waters, and also in cave waters; however, no aquatic species known; not treated in this work).
- 4 Chelicerae and pedipalps normally not visible in a dorsal view, protected by a projection („rostrum“) of the prodorsum (4-1); with or without trichobothria dorsolaterally on the prodorsum; (in some nonaquatic families, posterior dorsum segmented by transverse sclerotized plates); adults characterized by genital and anal openings similar in dimensions, both protected by “window shutter”-like paired plates (2-4 *c*), usually heavily sclerotized, with a yellow to dark brown cuticula (sometimes with a mirror-like reflecting surface, sometimes whitish due to reflecting plas-tron structures); nymphs characterized by a genital field without sclerotized plates, in the form of longitudinal slits, mostly with a soft integument and unpigmented, but sometimes bearing distinctly contrasting brownish or reddish plates and appendages. **Oribatida** (page 89)
- Chelicerae and palps generally visible in a dorsal view, not protected by a projection („rostrum“) of the prodorsum (2-4 *i*, exceptions are made in species of *Isobactrus*, halacarids with the gnathosomal base not visible in a dorsal view, of *Calyptostoma*, terrestrial Parasitengona, as well as a few Hydrachnidia taxa with gnathosoma and its appendages being retractable into the idiosoma); generally with extended membranous areas (2-4 *e-g*, *i*) or, if heavily sclerotized, plate surfaces not darkened, at most opaque due to dense porosity (2-4 *d*, 7-18 *b*, 7-19); colour variable, from shiny white to intensely red, blue, green or brown; dark coloration, if present, always originating from internal organs, not from sclerite pigmentation; dorsum variable, but never with several transverse dorsal shields; adults with a genital pore far larger than the anal opening, “window shutter”-like plates may be present flanking the gonopore, but never the anal pore (2-4 *f*); if nymphs have appendages with dark colour, sclerotized structures similar in colour are generally found on the idiosoma. 5
- 5 Idiosoma dorsally covered with a single hemispherical shield (2-5 *c*), empodium consisting of one to five adhesive hairs (2-5 *b*); palp tibia with claw; trichobothria and genital acetabula absent on idiosoma, body of red coloration, specimens often found floating on water surface.
. **Homocaligidae** (Eleutherengona)
(restricted to aquatic habitats, in Central Europe only *Homocaligus scapularis* (C.L. KOCH, 1838), not treated in this work)
- Idiosoma dorsally without a single hemispherical shield, empodium absent or claw-like; palp tibia with or without claw; trichobothria and genital acetabula present or absent. 6
- 6 Palp tarsus distally with two conspicuous, long setae, gnathosoma snout-like (2-5 *a*); chelicerae elongate, distally with small movable digit; palp tibia without claw; two pairs of trichobothria; reddish body coloration, active at terrestrial conditions. **Bdellidae** (Eupodina)
(several species present in semi-aquatic habitats, not treated in this work)

- Palp tarsus various, but without two conspicuous, long setae distally, gnathosoma not snout-like; chelicerae various; palp tibia with or without claw; trichobothria present or absent. 7
- 7 Leg and idiosoma surface with a dense cover of setae („hypertrichous“, 2-4 e); osmoregulatory organs („acetabula“) present or absent; if present always arranged in the genital area, at most three pairs; idiosoma normally with a soft integument, often red in colour; legs always lacking swimming setae. **Trombidia (terrestrial Parasitengona)** (page 158)
- Leg and idiosoma surface generally with setae arranged in distinct patterns (2-4 d, f, g, i); idiosoma never densely covered with setae; osmoregulatory organs, integument and colour various; legs may bear swimming setae (2-4 h). 8
- 8 Palp bearing at most four movable segments (5-1 c); osmoregulatory organs („acetabula“) in three pairs in the genital field, minute, knob-shaped; two pairs of legs directed anteriorly and two pairs posteriorly (2-4 d); mostly minute in dimension and with little apparent coloration, idiosoma in general with four dorsal and four ventral plates in a cruciform arrangement (5-1 a-b, but fusion of these plates possible: 2-4 d); without regularly arranged idiosomal glands associated with sensillae; legs without swimming hairs (vs. fine hairs extending in length over more than 50 % of the next, following segment).. **Halacaroida** (page 113)
- Palp typically with five segments (7-14 - see there 7-14 f for a striking exception); osmoregulatory organs („acetabula“) in several taxa not restricted to the genital field, often enlarged or numerous

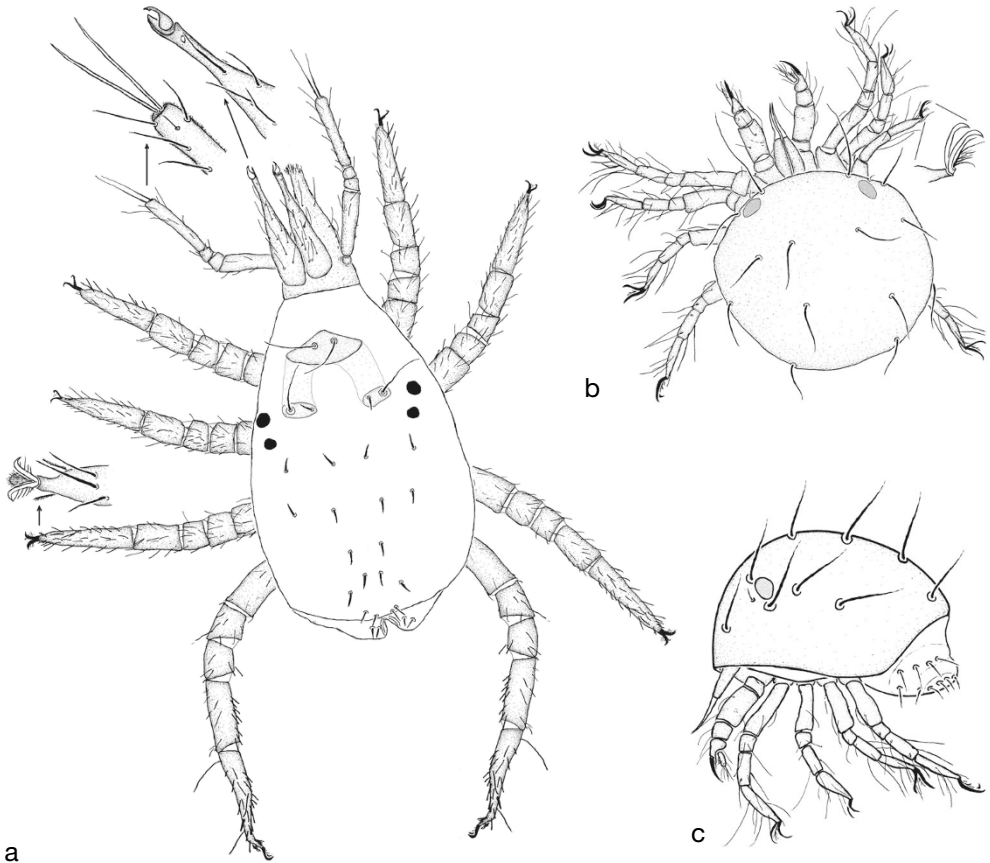


Fig. 2-5: Mites occasionally found in water: a, Bdellidae, Eupodina (*Molgus* sp. - details: tips of leg, palp and chelicera); b-c, Homocaligidae, Eleutherengona (*Homocaligus scapularis*), b, dorsal view. (detail: leg claws with adhesive hairs), c, lateral view.

(7-15); in general, legs in a circular arrangement or restricted to the anteriormost part of the idiosoma (2-4 f- g, i - exceptions are found in members of the Hydrovolzioidea, 7-12 c, with leg arrangement similar to Halacaroida); dimensions, coloration, and idiosoma sclerites variable, with numerous pairs of regularly arranged idiosomal glands associated with sensillae (7-1); legs variable in shape, in many taxa with swimming setae (2-4 h).
 **Hydrachnidia (aquatic Parasitengona)** (page 241)

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General Glossary Acari

(further terms used only in particular groups of mites are listed in the glossaries of the respective chapters)

acetabulum	Osmoregulatory organ of actinotrichid mites: Plesiotypically situated in the genital field, three pairs in adults, two pairs in nymphs, in larvae and some juvenile Oribatida only one pair (called „ur stigma“ or „Claparède organ“); in larvae of Hydrachnidia and in all stages of Acaridida in the interspace between leg I and II; secondarily, acetabula can be numerous and spread all over the idiosoma (= genital papillae, [German] Genitalnapf)
ambulacrum	clinging organ at the tips of legs, made up of an unpaired (often reduced) empodium and paired or unpaired claws; claws may be provided with clawlets (claw-like subdivisions of claws) and a claw blade (knife-like extension of the inner claw surface); in some taxa, the empodium may be claw like and occasionally the only functional claw.
anal opening	posterior opening of the digestive system. The rectum in some mites may be separated from the anterior intestine and then functions as an excretory system, often also called „anal pore“ or „excretory pore“. (= anus, [German] Exkretionsporus)
anus	see „anal opening“
apodeme	internal extension of a cuticular sclerite, in general functioning as muscle attachment site
apoderma	see „resting stage“
calyptostase	see „resting stage“
camerostome	see „gnathosoma“
capitulum	see „gnathosoma“

chelicera	appendage of the first prosomal segment of Chelicerata, in mites at least basally embedded into the gnathosoma; plesiotypically three-segmented, with the two distal segments acting as scissors („chelate“), but in many taxa two-segmented, with the third segment reduced to a membranous extension, or all segments fused to a stillete-like penetrating appendage (= [err.] „maxilla“)
Claparède organ	see „acetabulum“
claw, clawlet, claw blade	see „ambulacrum“
condyle	heavily sclerotized articulation point between leg segments
cornea	see „lateral eyes“
coxa	basal leg segment, in all mite taxa treated here (Actinotrichida) fused to the idiosoma surface and functioning as a basal plate on which the trochanter inserts as the first movable leg segment; neighbouring coxae may fuse and form „coxal plates“ („epimeral plates“), or all coxae may be fused into a continuous (→) „ventral shield“ (= „epimeron“)
coxal plate	see „coxa“
cuticle	as in all arthropods, consisting of an internal procuticula with „pore channels“ (rarely differentiated in endo- and exocuticula), and an external epicuticula; a cerotegument (cement- or wax layer hampering dehydration) is present in representatives of terrestrial mite groups - probably for this reason, aquatic Oribatida evolved particular respiratory surfaces (singular: „plastron“); in many Hydrachnidia, when pore channels become enlarged in order to allow respiratory activity of tracheoles, the procuticula has a reticular structure
dorsal furrow	see „dorsal shield“
dorsal shield	large sclerotized shield covering the whole dorsal idiosoma; in most cases separated from the (→) „ventral shield“ by a ring shaped membranous area called „dorsal furrow“
epimeron	see „coxae“
excretory pore	see „anal opening“
eye capsule	see „lateral eyes“
femur	see „leg“, „palp“
frontale	plesiotypically a sclerotized ring in the anterodorsal area of idiosoma surrounding an unpaired, weakly pigmented „frontal eye“ (occasionally called „median eye“ or „frontal organ“); in many clades, this eye being reduced, a more or less extended sclerite („frontal plate“, „frontal shield“) is formed in this area, occasionally including also surrounding (→) dorsalia.
frontal eye, fr. organ	see „frontale“
frontal plate, fr. shield	see „frontale“
genital bay	see „genital field“
genital field	area surrounding the opening of the reproductive system („gonopore“), plesiotypically on the anterior ventral surface of the opisthosoma, with anterior („pregenital“) and posterior („postgenital“) sclerites delimiting the slit-shaped gonopore; occasionally, a depressed area surrounding the male gonopore is called „genital pit“
genu	see „leg“, „palp“
glandularium	defence organ typical for, and a synapomorphy of, Hydrachnidia (see there)
gnathosoma	anterior body section, formed by the fusion of palp coxae, an unpaired sternal sclerite, and a dorsal extension, bearing as appendages the cheli-

	cerae and palps; partly, a caudal extension of the gnathosomal base, extending medially between the anterior coxal plates is called „anchoral process“; the bay formed by the medial margins of first coxae which embrace the gnathosomal base is called „gnathosomal bay“; if the attachment site for the gnathosomal base at the anterior opening of the idiosoma is formed as a collar-like sclerotized ring or tube, it is called „camerostome“; if the anterior part of the gnathosoma (anterior from the palp insertions) is narrowed and projecting, it is called „rostrum“. (= capitulum; infracapitulum [referring to the gnathosoma without appendages]; [German, err.] Maxillarorgan)
gnathosomal bay	see „gnathosoma“
gonopore	see „genital field“
hypostome	see „infracapitulum“
hysterosoma	posterior secondary body section, probably formed by segments of legs III and IV and the opisthosoma (in Oribatida often covered with (→) notogaster plate); anterior body section (→) proterosoma
idiosoma	body section posterior to gnathosoma, including segments of legs and all segments following caudally, as well as elements belonging to the mouth part-bearing segments I and II (→ „frontal eye, lateral eyes“)
infracapitulum	referring to the gnathosomal base (= gnathosoma without appendages); the sclerotized ventral part of this structure is called „subcapitulum“, its median longitudinal channel „hypostome“
integument	see „cuticle“
lateral eyes	paired light sensitive organs at the anterior edge of the idiosoma, generally consisting of paired lenses („corneae“) and retinulae, often enclosed in a sclerotized eye capsule
lateralia	see „plate“
leg	appendage of podosoma segments, often numbered „I-L, II-L, III-L, IV-L“, in most mite species treated here, consisting of a coxal plate fused to the idiosomal surface and originally six movable segments („trochanter, basifemur, telofemur, genu, tibia, tarsus“, often numbered: I-IV-L-1-6); tarsus with (→) „ambulacrum“; water mites with particular adaptations for swimming bear „swimming setae“ (see chapter Hydrachnidia)
lyrifissure	see „slit organ“
median eye	see „frontale“
metapodosoma	the posterior part of the podosoma, bearing legs III and IV, separated from the propodosoma, by the (→) „transversal furrow“
notogaster plate	dorsal shield of posterior body part in Oribatida
opisthosoma	in the ground plan of chelicerata a posterior tagma behind the last leg-bearing segment of the „prosoma“; in mites secondarily fused with the four leg-bearing segments of the (→) „prosoma“ forming the (→) „idiosoma“
palp	appendage of segment II of all chelicerata, generally involved in the uptake of food (grasping, tasting), occasionally also in spermatophore deposition or transfer; in actinotrichid mites plesiotypically consisting of five free segments („trochanter, femur, genu, tibia, tarsus“, often simply numbered as „P-1“ to „P-5“); pincer-like palps are called „chelate“ if P-5 is acting against a pointed dorsal projection or „antagonistic bristle“ of P-4, or „subchelate“ (= „uncate“) if P-5 is acting against a ventrodistal projection of P-4. (= pedipalp)
pedipalp	see „palp“

pharynx	anterior, dilatable part of the digestive tract, in mites located in the gnathosoma
plastron	non-compressible thin air layer on hydrophobic surface areas (→ respiratory system)
plate	idiosoma sclerite of minor (then sometimes called „platelets“) or medium dimensions, generally functioning as muscle attachment sites and plesiotypically arranged in a characteristic pattern reflecting the original body segmentation („dorsalia“, lateralia, (→) „frontale“); if numerous plates fuse to larger structures, covering extended parts of the body, they are called „shields“
podosoma	body area of the leg-bearing segments
pore channels	see „cuticle“
prodorsum	anterior dorsal part of idiosoma, probably the dorsal plate of the „propodosoma“ with legs I and II
propodosoma	the anterior part of the podosoma, bearing legs I and II (posterior part of the podosoma: metapodosoma)
prosoma	in the ground plan of chelicerata a tagma including segments I to VI (mouth part and leg segments); in mites its anterior part forming a separate body division (→) „gnathosoma“, its posterior part including the segments of the legs („podosoma“) may be fused with the (→) „opisthosoma“ forming the (→) „idiosoma“
proterosoma	anterior secondary body section, probably formed by (→) gnathosoma and segments of legs I and II (in Oribatida often covered with (→) prodorsum); posterior body section (→) hysterosoma
resting stage	inactive prelarva („schadonophan“) of all mites, and protonymph („nymphophan“) and tritonymph („teleiophan“) of Parasitengona; the term „apoderma“ is used in such quiescent stages for the internal cuticle, not exposed to the surrounding environment. (= „calyptostase“)
respiratory system	in mites consisting of tracheae and tracheoles plesiotypically communicating with the environment via „stigmata“; in water dwelling or hygric mites, these stigmata are closed and the unsclerotized parts of integument take over respiratory function, often facilitated by porosity of sclerites and development of fine peripheral „tracheoles“; occasionally completely reduced
rostrum	a narrowed anterior projection, (1) of the (→) „gnathosoma“ (apically bearing the mouth opening); or (2) of the (→) „prodorsum“ (in Oribatida, dorsally covering the gnathosomal base)
shield	see „plate“, „dorsal shield“, „ventral shield“
slit organ	mechanoreceptive organ typically found in all arachnids, consisting of an innervated slit-shaped opening in the sclerotized cuticle; organs composed of several slits are called „lyriform organs“ or „lyrifissures“, a term sometimes incorrectly used also for the single slits found in mites; in the ground pattern of Hydrachnidia, four pairs are found on the dorsum, and one in the posteroventral region; in other mite groups, different patterns are found
stigma	see „respiratory system“
subcapitulum	see „infracapitulum“
suture	strongly sclerotized line visible on a plate or shield originating from the fusion of several separate sclerotized elements, indicating their former contours
tarsus	see „leg“, „palp“
tibia	see „leg“, „palp“

tracheae, tracheoles	see „respiratory apparatus“
transversal furrow	area between the segments of leg-II and leg-III, often distinctly separating the anterior leg segments („propodosoma“) from the posterior ones („metapodosoma“) by a membranous intersegmental zone
trochanter	see „leg“, „palp“
ur stigma	see „acetabulum“
ventral shield	a continuous sclerotized plate including coxae, genital field, and ventralia, occasionally also the anal opening; in Hydrachnidia generally found in combination with a (→) dorsal shield. In higher Oribatida, this term is used for a plate surrounding the anal and genital openings, generally fused with the coxae

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