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Zooplankton of the Open Baltic Sea: Extended Atlas

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Zooplankton of the Open Baltic Sea: Extended Atlas

By

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2009

In memory of Professor Ulrich Schiewer

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ABSTRACT

This is the Second Edition (improved and extended) of the third volume in a series of zooplankton atlases of the Baltic Sea. It describes zooplankton community of the open Baltic waters, which is a mixture of marine species and diverse representatives of brackish water and limnetic faunas. Brief information on morphology and ecology of zooplankters, picture key to higher invertebrate taxa and methodological recommendations for sampling, identification and counting of zooplankton are provided. Two checklists present more than 1030 names of micro-, meso- and macrozooplankton organisms. The volume is enhanced by 4 tables, 31 line drawing, and 462 colour photographs. The photographs of zooplankters are combined into 63 photo plates depicting most common species of Protozoa (Ciliata), Cnidaria, Ctenophora, Turbellaria, Rotifera, Phyllopoda, Copepoda, Chaetognatha and Copelata, as well as meroplanktonic larvae of Polychaeta, Mollusca, Cirripedia, Bryozoa and Echinodermata. The atlas is recommended for university students, zoologists and aquatic ecologists who are investigating and monitoring the pelagic ecosystem of the Baltic Sea.

PREFACE

This volume is the Second Edition (revised and extended by new information on ciliates) of the third book in a series of zooplankton atlases – an illustrated inventory of planktonic invertebrates inhabiting the open waters of the Baltic Sea. The two previous volumes deal with the estuarine mesozooplankton of the Baltic Sea (Part I, Rotifera: Telesh & Heerkloss, 2002; Part II, Crustacea: Telesh & Heerkloss, 2004).

The necessity of creating a zooplankton inventory of the Baltic Sea had been under discussion since the 1980-s when Dr. Gerda Behrends headed the Working Group (WG) "Zooplankton" within the association of the Baltic Marine Biologists (BMB). In the early 2000-s, the BMB WG No. 29 "Zooplankton Diversity" was established (convener – Dr. Irena Telesh) in order to continue and facilitate the biodiversity research in the Baltic Sea region. The present series of zooplankton atlases of the Baltic Sea was initiated and produced by the members of the BMB WG No. 29 "Zooplankton Diversity".

It is worth mentioning here that since 1998 Professor Ulrich Schiewer (1936-2007) from the University of Rostock, to whom we dedicate the present volume, enthusiastically supported this work.

This atlas provides information on the zooplankton of the open Baltic Sea, which is to great extent a mixture of marine species and diverse brackish water and limnetic faunas typical for the vast estuarine and coastal areas located mainly in the southern and north-eastern parts of the Baltic. The specific feature of this volume is the updated species list of meso- and macrozooplankton of the open Baltic (217 species), and the checklist of ciliates (814 species) inhabiting open waters of both, central and coastal areas of the Baltic Sea.

The atlas is illustrated by 31 line drawing and 462 colour photographs. The photographs are combined into 63 photo plates depicting most common holo- and meroplanktonic representatives of different invertebrate taxa: Protozoa (Ciliata), Cnidaria, Ctenophora, Turbellaria, Rotifera, Phyllopoda, Copepoda, Cirripedia, Polychaeta, Mollusca, Bryozoa, Echinodermata, Chaetognatha and Copelata.

A new feature of the present edition is the extended chapter "Ciliates of the Baltic Sea" which contributes substantially to both biodiversity and cyto-ecological research directions in the area. The chapter is illustrated by 197 original colour photographs that describe the diverse cell morphology of the free-living ciliated protists in the Baltic Sea.

Photographs of live ciliates were made with the help of the Research Microscope LEICA DM-2500 at magnifications 40x and 100x using the regimes of bright field (BF), differential interference contrast (DIC), or phase-contrast (PHC).

Original photographs of major other zooplankters were made with a CANON video system connected to OLYMPUS Research Microscope BH-2. In the illustrations, the magnification is indicated by a scalebar with a number which is its length in micrometers (µm). Alternatively, the size of a depicted organism is indicated in the legend.

This volume should be of practical use for students and technicians as well as for scientists. Suggestions about how to improve the project and cooperation to enhance it are most welcome.

1. INTRODUCTION

Since the last glaciation, the Baltic Sea has undergone several evolutionary stages between a huge marine bay and a large freshwater lake during which a number of different ecological systems developed and were successfully replaced in this area (Jansson, 1972).

Today the Baltic Sea is the largest brackish water area in the world characterized as a temperate shelf sea with permanent salinity stratification, a horizontal salinity gradient and low water turnover of 35 years. It is a shallow sea with a mean depth of 62.1 m, the greatest depth 459 m, an area of 415,266 km^2 (Baltic Proper itself is 211,069 km^2), a volume of ca. 22,000 km^3 (HELCOM, 2001; Wulff et al., 2001; Schiewer, 2008). The presence of shallow sills at the western inlets causes stable water stratification. The Baltic can be best compared to a stratified fiord with a rich supply of fresh water from the rivers.

Due to the humid climate, there is an estuarine circulation with outflow of low-saline water above the halocline and powerful periodic injections of North Sea water below the halocline which greatly affect the salinity of the deep water layers.

The location of the Baltic Sea in the northern high latitudes means a pronounced seasonality in temperature and light regime. The vegetation season lasts longer in the southern areas than in the north of the sea. Ice coverage is occasional in the southern Baltic. The average temperature of the surface waters in summer is 16° C, and a thermocline is formed at the depth of 10 to 30 m. The water below thermocline is usually colder than 7-8° C during the whole year. The water stratification delimits vertical exchange of water masses and pelagic organisms thus creating layers with different ecological characteristics.

In terms of salinity, the regime ranges between oligohaline (0.5 PSU) and mesohaline (18 PSU) conditions, with an average of 7-8 PSU in the major open Baltic waters (HELCOM, 2001). Climate change and decadal scale variability of these parameters modifies the hydrographic characteristics accordingly (BACC, 2008; Feistel et al., 2008).

Since the end of the XIX century many authors have tried to divide the different Baltic Sea areas with respect to hydrographical conditions, and their ideas have differed greatly in many cases (Ekman, 1931, and Wattenberg, 1949, cited after Ackefors, 1969).

In the present study we use the following names of the different sub-areas of the Baltic Sea and a classification which is largely based on the division of the sea made by Ackefors (1969):

- **Baltic Proper** – the area east of the Belt Sea and the Sound, limited at the north by the Åland Sea and the Archipelago Sea, at the east – by the Gulf of Finland;
- **Western Baltic Sea** includes Kiel Bight and Mecklenburg Bight;
- **Northern Baltic Sea** includes the Åland Sea, the Archipelago Sea, and the Gulf of Bothnia;
- **Southern Baltic Sea** – the area of Gdansk Basin;
- **Eastern Baltic Sea** is attributed to the Gulf of Riga and the Gulf of Finland.

This is a very general division of the Baltic Sea; moreover, it is defined to a great extent by the availability of published data on zooplankton species composition.

As any other attempt to classify the natural systems, including water bodies, the proposed subdivision of the Baltic Sea is largely **conventional** and the real borders between the areas mentioned above do not exist. This is especially true for the pelagic communities that may be driven by water masses to significant distances.

The shallowness and the consequently vast area occupied by the coastal ecosystems in the Baltic Sea (Schiewer, 2008) are the major reasons for the intensive mixture of the coastal and open-water plankton communities and for the penetration of brackish water, euryhaline and also freshwater species of zooplankton far into the open Baltic waters (Telesh et al., 2008a). Thus, the strict definition of the "open Baltic Sea" in respect to the pelagic fauna can hardly be given.

Due to peculiarities of the salinity regime, the pelagic ecosystem component in the Baltic Sea consists mainly of plankton communities dominated by euryhaline species. The organisms in the Baltic are well adapted to the brackish water environment, but only a few true brackish water species have developed here. The present species composition is a result of the selection process, where organisms with a high osmotic resistance have been able to survive.

Since the publication of the "species minimum curve" by Remane (1934, 1940), it has been generally accepted that "the number of species in the Baltic is small" (Jansson, 1972, p. 12). This conclusion was commonly applied to and supported mainly by the data on benthic macrofauna (Zenkewitch, 1963). Meanwhile, already in the 1960-s Hans Ackefors proposed that "**if the microfauna in the water and at the bottom are included the number of species will be much higher**" (Ackefors, 1969, p. 5). In other words, according to an exceptionally evocative affirmation of Jansson (1972), "the diversity is there but it is found on a microscale with a beautifully designed network of flows among many different kinds of bacterial decomposers" (p. 14).

Thus, already in the second half of the XX century scientists around the Baltic were admitting that the real diversity of microscopic invertebrates in plankton might happen to be much higher when special biodiversity investigations are performed. This idea was later supported by the results of the long-term zooplankton diversity and ecosystem functioning research in the eastern Baltic coastal waters which demonstrated high species richness of pelagic communities in major Baltic estuaries and other coastal ecosystems (for details see the review publications: Telesh, 1987, 1988, 2001, 2004, 2006a, 2006b; Telesh & Heerkloss, 2002; Telesh & Heerkloss, 2004; Telesh et al., 2008).

However, until recently attempts to evaluate the total zooplankton diversity including the unicellular organisms in the open Baltic Sea have hardly been made. Zooplankton diversity in the Baltic Sea has been routinely described in terms of dominant species of certain groups (mainly copepods) and size fractions (mesozooplankton) that are identified and counted for monitoring purposes (see Chapter 2). Presently, geographical coverage of the Baltic Sea, as well as of other European seas is still incomplete for Protista, Rotifera and Brachiopoda (Costello et al., 2006).

Meanwhile it is widely accepted that assessment of zooplankton species diversity provides important information on the marine ecosystem structure, trophic webs and functions, and their natural and/or human induced alterations. In many zooplankton groups, major functional characteristics responsible for the animals' behaviour and interactions within the community are species-specific, therefore the importance of correct taxonomic identification of zooplankton, especially of key species, indicators of water quality, and non-indigenous species can hardly be overestimated.

On the basis of data published in the first edition of zooplankton atlas of the open Baltic Sea (Telesh et al., 2008b) we can conclude that the earlier existing conception of the low species diversity of planktonic communities in the Baltic Sea had resulted from the insufficient knowledge on the species composition of zooplankton, particularly its small-size fraction (Telesh, 2008; Mironova et al., 2009; Telesh & Skarlato, 2009).

It is commonly accepted that a marine zooplankton community is formed by the following size fractions: **picoplankton** (size of organisms 0.2-2.0 µm, mainly heterotrophic bacteria), **nanoplankton** (2.0-20.0 µm, heterotrophic nanoflagellates), **microplankton** (20-200 µm, ciliates and a large part of rotifer species), **mesozooplankton** (0.2-20.0 mm, larger rotifers, mainly planktonic crustaceans, meroplanktonic larvae of some benthic invertebrates, etc.), and **macrozooplankton** (organisms larger than 20 mm:

Cnidaria, Ctenophora, Chaetognatha, Mysidacea, Euphausiacea, Decapoda, Polychaeta and others) (Lenz, 2000).

The sub-division of zooplankton into micro- and mesozooplankton size classes used in this book is (to a certain extent) not a traditional one: we consider all rotifers as mesozooplankton, applying the term "microzooplankton" to planktonic ciliates. This is another conventionality determined by the fact that the authors want to draw special attention of the readers to the chapter "Ciliates of the Baltic Sea". It is the first attempt to provide updated illustrated information on biodiversity of these protists in the Baltic ecosystem. Unlike the dominant mesozooplankters, ciliates are usually not considered in the regional monitoring programs; nevertheless, they are good indicators of water quality. They play an important role in the zooplankton communities, and contribute significantly to energy fluxes (through the microbial loop, for example) and water purification in the Baltic Sea ecosystem. The chapter provides a checklist of 811 species of planktonic and benthic ciliates (which may also be numerous in plankton) that inhabit both open and coastal waters of the Baltic Sea. This part of the atlas is illustrated by the original photographs of live ciliates collected in the Gulf of Finland, eastern Baltic Sea and cultured in the laboratory for the purpose of precise species identification.

Aquatic ecologists know that species identification of zooplankton organisms is a tedious and time-consuming work which requires certain taxonomic skills, understanding of the general principles of species identification, and knowledge of taxonomically-important morphological characteristics of zooplankters from different groups. On a regular basis, species determination should be performed with the help of taxonomic identification keys. Additionally, illustrated atlas books with drawings and photos of live and preserved planktonic organisms can be also helpful; however, they can not substitute the classical taxonomic guides.

Nowadays, it is a common problem worldwide that professional taxonomists with the deep knowledge of the systematic of different groups of aquatic invertebrates become extinct (Costello et al., 2006). For the Baltic Sea region, taxonomic training of the professional staff in hydrobiological laboratories storing their results in joint databases is of exceptional importance for harmonising the methods and improving the skills necessary to identify the zooplankton species. These training courses for Baltic zooplankton identification are essential for acquiring and maintaining the quality assurance of the laboratories participating in the joint international monitoring programmes in the Baltic Sea region. Unfortunately, such taxonomic training courses are rare so far.

The authors hope that the new, extended zooplankton atlas of the open Baltic Sea together with the previously published atlas books on the Baltic estuarine and open-water zooplankton will contribute to the general knowledge of marine biodiversity in the region and make the species identification of zooplankton more easy. The atlas is recommended for university students, zoologists and aquatic ecologists who are investigating and monitoring the pelagic ecosystem of the Baltic Sea.

2. GENERAL CHARACTERISTICS OF ZOOPLANKTON OF THE BALTIC SEA

Zooplankton organisms range in size from few micrometers to meters. **Mesozooplankton** (0.2 – 20 mm) is the dominating group in the Baltic Sea in terms of biomass. It may constitute 76% (i.e. >1000 kg C/m²) of the average annual carbon mass, as it was measured in the western Gdańsk Bay during the 1980-s (Witek, 1995). The remaining 18 and 6% were contributions of protozoans and macrozooplankton, respectively. The percentage of the average annual production of mesozooplankton was 39%. Within the mesozooplankton fraction, copepods *Pseudocalanus* spp., *Temora longicornis*[1], *Acarti*a spp., rotifers *Synchaeta* spp., and cladoceran *Evadne nordmanni* are the most important taxa in both, biomass and production. The ctenophore *Pleurobrachia pileus*, the copepod *Eurytemora affinis* and rotifers *Keratella* spp. played a minor role, while the appendicularian *Fritillaria borealis*, Polychaeta larvae, the cladocerans *Bosmina* spp., *Podon* spp., the copepod *Centropages hamatus*, and Bivalvia larvae ranged in between (Fig. 2.1).

There are about forty mesozooplankton species that are regularly occurring in the Baltic Sea in significantly high abundances (Ackefors, 1981). Ten to twelve of them are dominating taxa. Their spatial occurrence is mainly explained by the salinity patterns. According to hydrographic regime with prevailing outflow of low saline water in the upper layer and temporary inflow of higher saline water below the halocline, species of relevant salinity preferences inhabit the western and the eastern parts and the open Baltic Sea, respectively.

Behrends et al. (1990) described presence of the dominant taxa in the Baltic Sea regions from Kattegat to the Gulf of Finland and the Bay of Bothnia, accordingly, in a semi-quantitative way. After conversion of this information into numbers, the changing occurrence, amount and rank of various taxa from west to northeast became visible (Table 2.1). Generally, the amount of species is larger in the transition to the marine or the limnetic environment than in the Baltic Proper (Remane & Schlieper, 1971).

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[1] Authors of the Latin species names are mentioned in the zooplankton checklists (Tables 4.2.1 and 5.2.1).

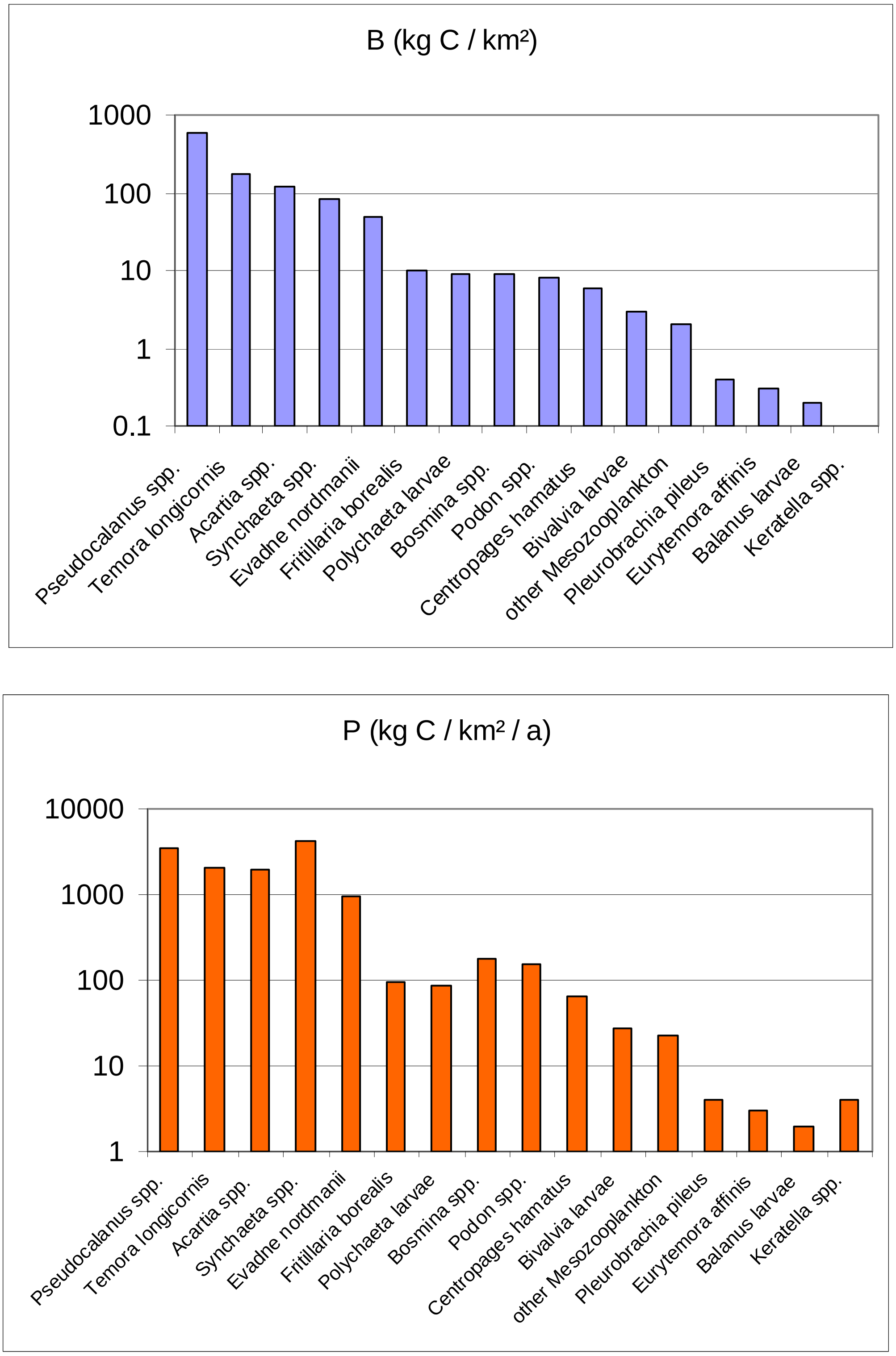


Figure 2.1. Importance of various zooplankton taxa in terms of biomass (above) and annual production (below) in Gdańsk Bay during the 1980-s (after Witek, 1995).

Additionally, there is a remarkable shift in the dominating taxonomic groups. Thus, *Paracalanus parvus*, *Pseudocalanus* spp. and *Oithona similis* dominantly occur in the entire water column of the western Kattegat, while *Calanus finmarchicus* and *Centropages typicus* occasionally appear there. Predominant cladocerans are the carnivorous *Evadne nordmanni*, *Podon* spp. and *Pleopsis polyphemoides* in this area. The brackish water filter feeding cladocerans from the genus *Bosmina* are dominant in the Baltic Proper during summer.

Partly in the eastern Kattegat and especially in the Sound, the zooplankton species composition demonstrates similarities to that in the near-surface water of the Arkona Sea, for example, by the occurrence of *Acartia* species, which is a result of the Baltic Sea water outflow. On the other hand, copepods *Acartia bifilosa* which tolerate a salinity of 0.30 PSU (Sewell, 1948), and *Eurytemora affinis*, which survive at 0.50 PSU (Busch & Brenning, 1992), are the key species in the Gulf of Finland and the Bothnian Sea. Finally, Behrends et al. (1990) described a two-layer distribution of zooplankton in the Bay of Bothnia. While the glacial relict copepods *Limnocalanus macrurus* inhabit the cooler and low-saline deep water, *Daphnia* species appear in the surface layers, in nearly fresh water conditions. *Centropages hamatus* is a subdominant; it occurs at maximum population densities from Kattegat to the Arkona Sea. The Baltic Proper is the area where *Acartia* species, *Temora longicornis* and *Bosmina* spp. (in summer) are dominating.

The **seasonality** is a pronounced reason for structural variability in plankton communities of temperate regions like the Baltic Sea. It is exposed in the reproduction cycles which are linked with the species' demands for food availability and for certain temperature. Rotifers typically dominate in May (*Synchaeta* spp.) and in August (*Keratella* spp.) when their parthenogenetic reproduction mode allows utilizing optimal food conditions within a short period of time. Cladocerans proceed in the same way. *Bosmina* spp. use a small temporal "window" in summer, when temperature rises above 15°C (Ackefors, 1969).

There are two species of appendicularians in the Baltic Sea, *Oikopleura dioica* and *Fritillaria borealis*. The first one prefers the higher salinity in the western Baltic Sea. Its reproduction maximum is in autumn while *F. borealis* inhabit all regions of the Baltic Proper, mainly in spring. Bivalvia larvae also peak in a bimodal way probably depending on different reproduction periods of various species which is likely, because two of four co-occurring species are more abundant (Ackefors, 1969) and have their reproduction from May to August (*Macoma baltica*) or from August to October (*Mytilus edulis*) (Hernroth & Ackefors, 1979).

Table 2.1.

Degree of dominance (from 1 – the lowest, to 7 – the highest) of the most common zooplankton taxa within the Baltic Sea regions from Kattegat to the Gulf of Finland and the Bay of Bothnia, respectively (modified from Postel, 1995).

Taxa	Baltic Sea regions										
	A	B	C	D	E	F	G	H	I	J	K
Paracalanus parvus	**1**	**2**		**5**[1]							
Pseudocalanus spp.	**2**	**1**	**1**	**2**	**2**	**2**	**1**	**1**	**4**	**3**	
Oithona similis	**3**	**4**	**4**	**1**	**4**						
Centropages hamatus	**4**	**3**	**2**	**3**	**3**	**4**					
Carnivorous cladocerans[2]	**5**	**5**			**5**[3]						
Meroplanktonic larvae	**6**			**4**[4]							
Calanus finmarchicus	**7**										
Centropages typicus	**7**										
Acartia spp.		**6**	**3**	**4**	**1**	**1**	**2**	**2**			
Oikopleura dioica			**5**[5]								
Temora longicornis						**3**	**3**	**3**			
Bosmina spp.						**5**[4]	**4**[4]	**4**[4]	**3**[4]	**2**[4]	
Evadne nordmanni							**5**				
Acartia tonsa				**5**[1]							
Acartia bifilosa									**1**	**1**	
Eurytemora affinis									**2**	**1**	
Limnocalanus macrurus									**4**	**3**	**1**
Synchaeta spp.									**5**[6]	**4**[6]	
Fritillaria borealis									**6**		
Pleurobrachia pileus									**6**		
Polychaeta (larvae)									**6**		
Keratella spp.										**4**[6]	
Daphnia spp.											**2**

A Shallow, western Kattegat
B Deeper, eastern Kattegat
C The Great Belt, Belt Sea
D Kiel Bay
E The Sound
F Arkona Sea
G Bornholm Sea
H Gotland Sea
I Gulf of Finland
J Aland Sea and Bothnian Sea
K Bay of Bothnia

[1] late summer / autumn
[2] *Evadne nordmanni, Podon* spp.
[3] not numerous
[4] in summer
[5] not every year
[6] in spring

The amount of co-occurring *Cardium* species and *Mya arenaria* is normally negligible (Ackefors, 1969). Polychaeta larvae are more abundant during the phytoplankton spring bloom than in the remaining time of the year. Finally, the seasonal patterns of the adult calanoid copepods density demonstrate one peak in March and another period of higher abundances during several months in summer and autumn.

Taking the key species with maximal abundance of several thousands individuals per cubic meter separately, the seasonal pattern of calanoids is more differentiated and explains the annual course of the total zooplankton abundance. *Pseudocalanus* spp. become mature in March, April and May; they are followed by *Acartia bifilosa* (May, July, August), *Eurytemora affinis* (July, August), *Temora longicornis* (July, August) and, finally, by *Acartia longiremis* (mainly August). *Pseudocalanus* spp. are probably responsible for the total zooplankton peak in May, while the majority of calanoids become adult in summer. This could be explained by different habitat temperatures. Meridianal shifts in seasonality are possible.

Decadal and multi-decadal variability in the atmospheric and consequently in the hydrographic regime also causes changes in mesozooplankton abundances and some times in species composition. Salinity and temperature changes are the main driving forces here. For example, the longer period of missing salt-water inflows and rising river runoff in the Northern Baltic Proper and the Gulf of Finland in the late 1980-s corresponded to the appearance of eight *Keratella* species and other rotifers (*Polyarthra* spp., *Kellicotia longispina*), as well as the cladocerans *Bythotrepes longimanus* (Postel et al., 1996). Consequently, the number of taxonomic groups increased. At the same time, the key species changed in the Central Baltic Proper. The former dominant halophilic representatives of the cold-water genus *Pseudocalanus* were substituted by the *Acartia* species. In the northern parts of the Baltic Proper, the former dominance of *Acartia* spp. was replaced by the brackish water species *Eurytemora affinis*. These results based on the HELCOM data set for the entire Baltic Sea were in accordance with the reports on the regional shifts published by Vuorinen and Ranta (1987), Viitasalo et al. (1990), Lumberg and Ojaveer (1991), Flinkman et al. (1998), Ojaveer et al. (1998), Vuorinen et al. (1998), Dippner et al. (2000, 2001) and Möllmann et al. (2000, 2003).

The role of the invasive species in the Baltic Sea increased during the recent decades. Currently, a number of cladoceran species from the Ponto-Caspian area (*Cercopagis pengoi, Evadne anonyx* and *Cornigerius maeoticus*), and a ctenophore from the American east coast (*Mnemiopsis leidyi*) are the examples of alien zooplankters in the Baltic Sea. As obligatory or facultative planktonic predators, they can impact the Baltic pelagic ecosystem. Therefore, such introductions have to be monitored very carefully. The fishhook water flea *C. pengoi* is regularly established now in the greater part of the Baltic Sea. It dominantly occurs in coastal waters, but it is also present in the open Baltic Sea (Uitto et al., 1999; Telesh & Ojaveer, 2002; Karasiova et al, 2004; Olszewska, 2006; Litvinchuk & Telesh, 2006). These carnivorous water flees feed on *Bosmina* spp. (Pollumäe & Väljataga, 2004; Gorokhova et al., 2005) and other planktonic filtrating crustaceans (Laxson et al., 2003), which are normally dominant in the central Baltic Sea during summer. Finally, due to elimination of other crustaceans by *C. pengoi*, panktivorous pelagic fishes feed on *Cercopagis* (Antsulevich & Välipakka, 2000).

These invaders are responsible for the prolongation of the food chain by one level which allows expecting additional energy losses during the general energy flow through the pelagic ecosystem of the Baltic Sea. This phenomenon can affect energy balance in general and the size of pelagic fish stocks in particular.

The influence of the most recent invader – the alien ctenophore *Mnemiopsis leidyi* on the pelagic food web of the Baltic Sea seems to be limited so far due to its low abundances in the Baltic Proper and the northern regions (Kube et al., 2007a, b; Lehtiniemi & Flinkman, 2007). Certain danger to the ecosystem might be expected from a spatial and temporal overlap between a potential mass occurrence of *M. leidyi* and cod eggs below the halocline of the Bornholm Basin (Haslob et al., 2007). This case requires further attention of the researchers (see also Chapter 5.1).

3. METHODS OF COLLECTING AND ANALYSING ZOOPLANKTON IN THE BALTIC SEA

3.1. Sampling: general aspects

Miscellaneous processes, like seasonality, daily vertical migration, swarming, etc. produce typical zooplankton distribution patterns (Haury et al., 1978). In reality, we observe a result of the combination of those processes, and skilful observation strategies need to be applied in order to distinguish them by choosing proper sampling duration (length) and measuring intervals (Sameoto et al., 2000). Appropriate **measuring intervals** are to be chosen considering the so called Nyquist sampling theorem (Nyquist, 1928). Following it, a signal must be measured in equal distances of more than two times within one period (or wave length) of the specific signal. Otherwise one produces aliased results as Figure 3.1 illustrates.

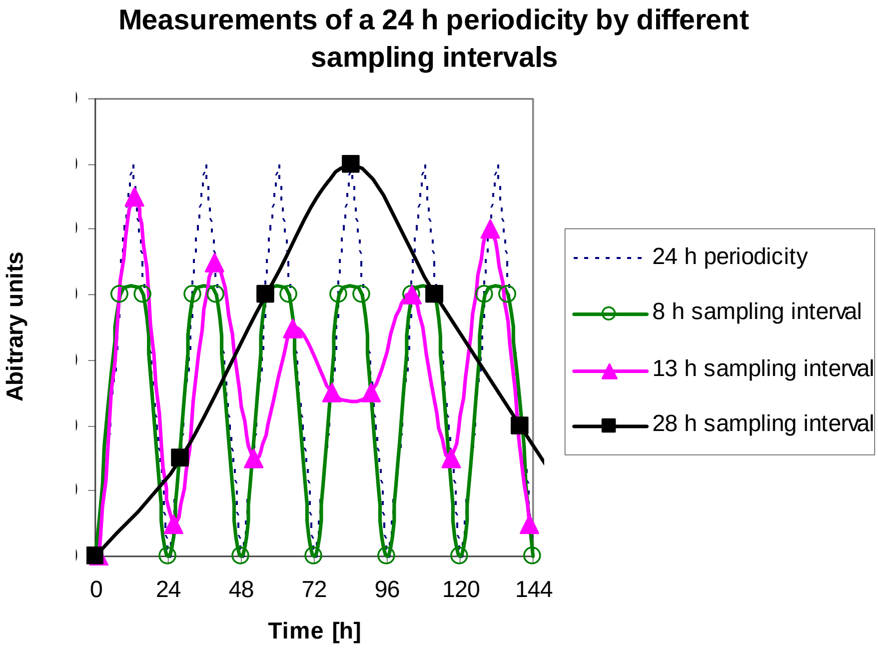


Figure 3.1.1. Examples of sampling intervals producing accurate (by 8 h) and biased (by 13 and 28 h) results of a 24 h periodicity.

Nearly accurate results will be obtained if a certain process will be measured at least three times within its periodicity or a wave length. For example, the twenty four hour periodicity of dial vertical migration demands an equidistant measuring interval of at least eight hours (Fig. 3.1.1). Longer distances, like thirteen or twenty-eight hours, produce apparent results of a thirty-six hour or even a seven day periodicity, for example. Continuous measurements would produce the most accurate outcome in both phase and amplitude. On the other hand, for exclusion of the dominant twenty four hour periodicity, for example due to daily vertical migration, one needs to measure the chosen parameter only during one phase of the process (at day or at night) or one needs to choose a twenty-four hour measuring distance. Vertically integrated sampling would also provide acceptable results.

For statistical reasons, to get the unbiased results, a period should be examined at least three times. That means a twenty-four hour periodicity demands a **measuring length** of three days.

Further, the **characteristics of the equipment**, i.e. the selectivity of a plankton sampler (type of net, mesh size, etc.) or the sensitivity of a sensor (e.g. depth recorder) influence the results accordingly. For example, the UNESCO Standard net WP-2 quantitatively selects organisms between 0.2 and 10 mm size (UNESCO, 1968). The limits are set by the mesh size of 200 μm on one hand and by the net opening area of 0.25 m^2 in combination with the towing speed of 45 m/min on the other. To obtain comparable results, one needs to use standardized techniques in this respect, too.

The "filter mechanisms" modifying "real patterns" into "observed patterns" determine mainly the accuracy but also the precision of the results (Fig. 3.1.2).

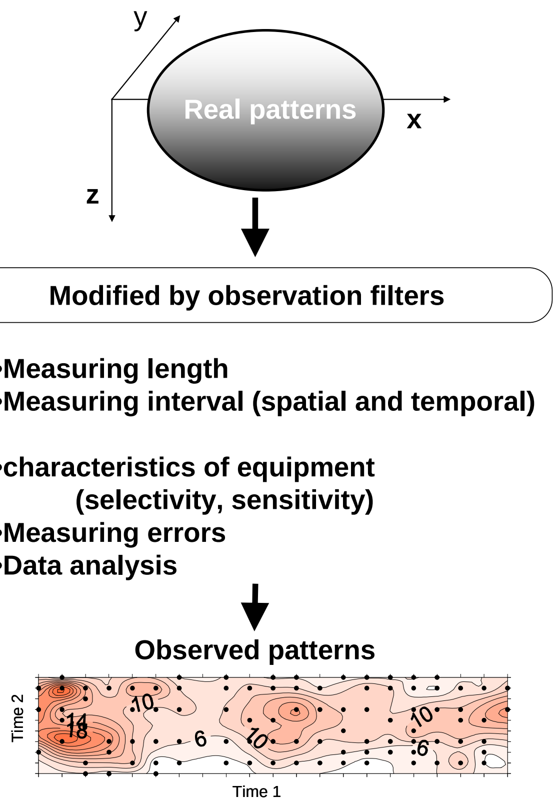


Figure 3.1.2. Factors modifying "real patterns" to "observed patterns" according to Postel (1983).

Measuring errors are to be divided into rough errors and absolute errors. The first category is not evaluated and therefore not taken into account. The second one splits into regular $\boldsymbol{\varepsilon_r}$ and irregular errors $\boldsymbol{\varepsilon_{ir}}$. Irregular errors will be determined by calculation of confidence limits. Examples are the results of sample splitting or of organisms counting in sub-samples. The regular errors include constant errors and systematic errors. A balance, which shows always a constant difference to the real weight, would be an example for a constant error, while an increasing weight of a dried sample due to an uptake of moisture with time is an example for a systematic error. Both are to be quantified and consequently provide part of a correction mode. The terminology bases on Junge (1981). According to the author, a measured value **a** differs from the real value **x** by $\boldsymbol{\varepsilon_r}$ and the scatter of $\boldsymbol{\varepsilon_{ir}}$, i.e.

$$x = (a +/- \varepsilon_r) \pm \varepsilon_{ir}$$

Finally, **methods of data analysis** may influence the outcome. For example, it is of importance to choose appropriate software settings, e.g. interpolation modes, when performing contouring mapping.

These and other aspects need to be considered when elaborating the appropriate sampling strategies, planning the sampling surveys, or evaluating the outcomes.

3.2. Sampling of meso- and macrozooplankton

For measuring the total amount of plankton, a set of equipment is necessary. Protozoan sampling demands water bottles (see below); however, larger organisms are included only occasionally and in a non-representative manner in such samples. They will be caught by plankton nets with different mesh sizes and geometries. Mesozooplankton in the sea is sampled best by the already mentioned WP-2 UNESCO Standard net (UNESCO, 1968). It is a closing net suitable for vertical tows and stratified sampling. Considering the smaller mesozooplankton in the Baltic Sea, this net (Figure 3.2.1a) is recommended for the HELCOM Monitoring and Assessment programme with a mesh size of 100 µm (HELCOM, 1988, 2005). In shallow areas, the use of horizontally or oblique towed instruments of a similar shape is suitable, like Bongo or Multiple nets (Figures 3.2.1b, c).

Collecting macrozooplankton demands nets with larger opening areas and mesh sizes (Fig. 3.2.1 d). For details regarding the different net characteristics see Wiebe and Benfield (2003) and references therein.

Figure 3.2.1: **a,** The WP-2 UNESCO Standard net being deployed aboard the R/V A. v. Humboldt; **b,** Twenty cm and 60 cm Bongo nets ready for deployment from the R/V Johan Hjort; **c,** The Multinet rigged for horizontal towing from aboard the R/V A. v. Humboldt; **d,** Deployment of a CalCOFI net from the R/V A. v. Humboldt. All photos stem from an ICES/GLOBEC Sea-going workshop for intercalibration of plankton samplers at Storfjorden, Norway, June 1993 (ICES, 2002).

Each of the recommended gear collects a certain fraction of plankton. Occasionally collected organisms could be excluded by sieving in a separatory column with graded sieves as proposed by UNESCO (1968) and illustrated in Figure 3.2.2. The separated fractions theoretically contain the organisms that are most ideally collected by each of the samplers. The sum of the concentrations of the separate results should give the best estimate of the "total plankton concentration". Examples for such strategies are published by Witek and Krajewska-Soltys (1989), Quinones et al. (2003), and Postel et al. (2007).

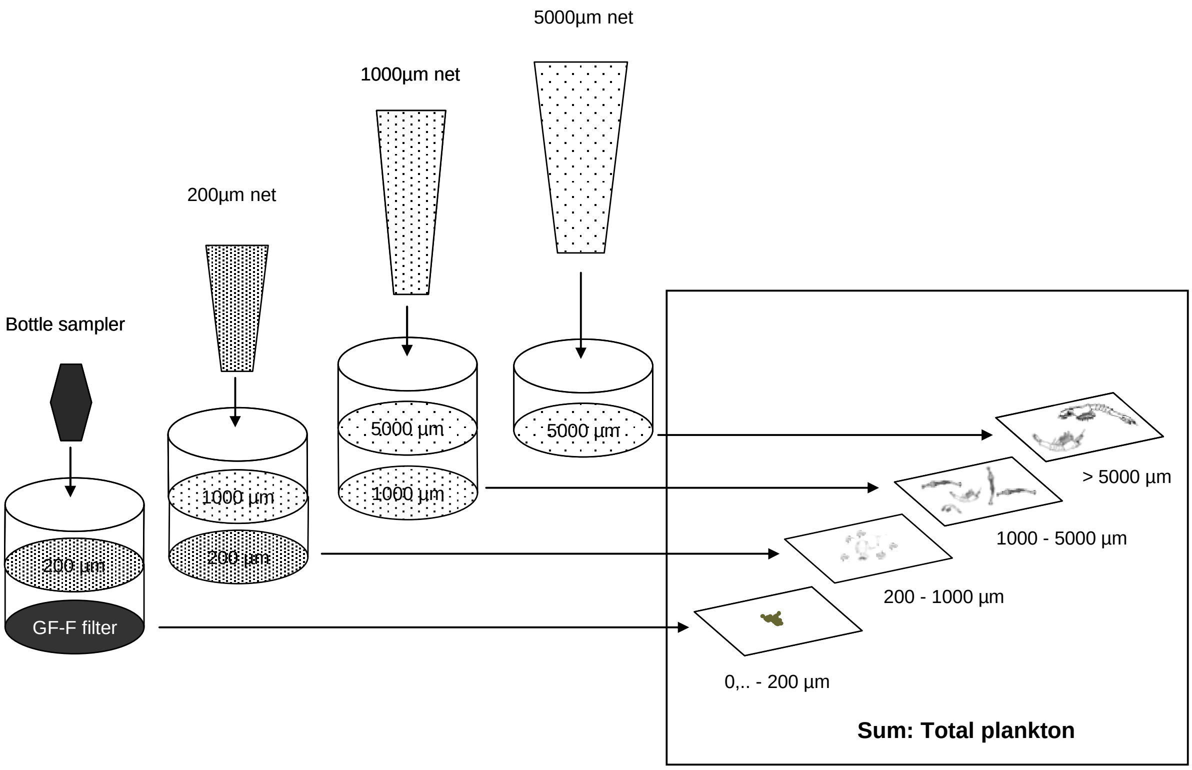


Figure 3.2.2. Scheme for eliminating the overlapping upper size fractions from samples collected with different mesh sizes. Ideally, the sum should be the total amount of plankton (UNESCO, 1968).

If several laboratories contribute to a joint data bank, standard operation procedures for sampling and analysing techniques are required to be used in each. For the HELCOM Monitoring and Assessment programme, a vertical sampling is recommended considering the actual hydrographic stratification.

The amount of filtered water should be determined by a flow meter. These instruments should optimally work within the towing velocity used. For example, the T.S.K. flow meter (The Tsurumi-Seiki Co., Ltd., Yokohama, Japan) is suitable for the WP-2 net. It should be placed in the half of radius. It is the position which provides nearly 100% filtration efficiency (Fig. 3.2.3).

Figure 3.2.3. UNESCO standard net WP-2 equipped with the T.S.K. flow meter on R/V Prof. Albrecht Penck, February 2007.

The number of flow meter rotations multiplied by the calibration factor gives the length of the filtered water column. It provides the filtered amount of water by multiplying it with the net opening area. The towing distance (which should be always noted for the flow meter control) could be also used for this purpose in a first approximation.

Wire angels will be considered by a trigonometric correction. The necessary length z_1 of wire out for reaching the depth level z will be calculated as z divided by the cosine of the wire angle α (Fig. 3.2.4).

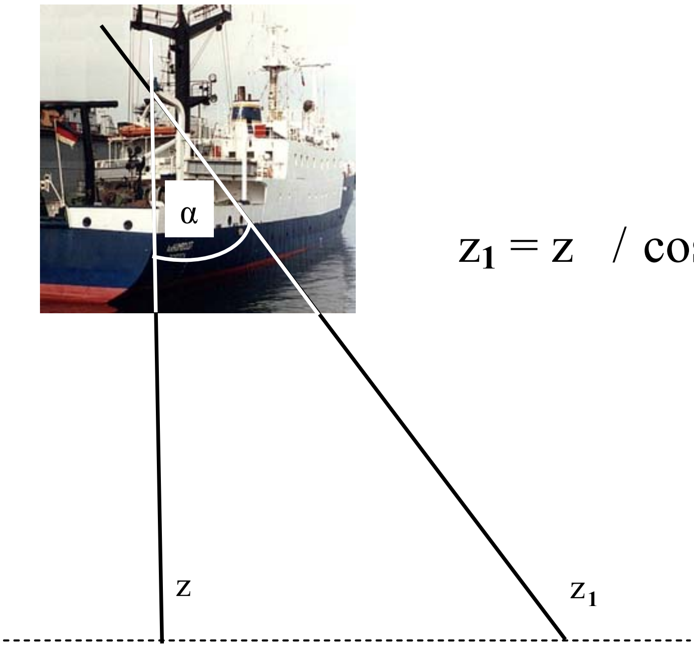


Figure 3.2.4. Scheme of trigonometric wire angle correction.

Finally, the net must be rinsed with seawater from outside. The sample concentrated in the cod end will be transferred to a sample jar and preserved by buffered formalin with a final concentration of 4%. Labeling of the sample jar inside and outside by station number, date, time, and sampling depth interval is important.

3.3. Sampling and study of microzooplankton

In order to study planktonic ciliates, most hydrobiologists collect water in routine, oceanographic sampling bottles (e.g. Niskin bottles). Firstly, it minimizes the cell disruption due to the turbulence and pressure, caused by pumps and plankton nets (Gifford & Caron, 2000). Moreover, plankton nets collect only a large fraction of ciliates (such as Tintinnids, in pioneering studies), but let nano-ciliates (<20 μm) pass through.

Immediately after sampling, subsamples should be preserved. The most common fixatives are acid Lugol's solution (2-10%), borax neutralized formalin (1-4%), or glutaraldehyde (1-2%). For the precise quantifying of ciliates, Lugol's solution is better than many aldehyde-based fixatives (Stoecker et al., 1994). The disadvantage of Lugol's solution is in masking the chlorophyll fluorescence, which may be needed to recognize mixotrophic ciliates (Gifford & Caron, 2000). Preservation in glutaraldehyde is used mainly for epifluorescence and electronic microscopy. After preservation, samples can be stored in a cool, dark place for several months.

Compound microscopes are commonly used to quantify and identify planktonic ciliates, as well as other microplanktonic organisms. Unfortunately, identification of ciliates is often difficult. The specialized technique of silver-staining, with protargol, reveals diagnostic features of ciliates (e.g. the nuclei and infraciliature) and is normally used for taxonomic descriptions. For more information about protargol staining, see Montagnes and Lynn (1987, 1993) or the review by Foissner (1991). Some fine diagnostic features of ciliates could be clearly recognized by scanning electronic microscopy.

The abundance of ciliates can be obtained in special settling chambers by counting under a microscope (Gifford & Caron, 2000). Lugol's fixation enhances the sinking of cells and also stains them a dark brown colour, that simplifies counting of ciliates. Another way to obtain planktonic ciliates' abundance is epifluorescence microscopy. The sample is stained with a fluorochrome (for example, primulin), a fluorescent dye that binds to nuclei acids. Then, it is excited by UV-light and can be observed with epifluorescence microscopy, using an inverted microscope. The detailed procedure of sample preparation is described in Gifford and Caron (2000), Strüder-Kypke et al. (2003).

All fixatives do not necessarily preserve the cell shape and size of live specimens, it is important to note that fixatives shrink cells. So, investigators will use a conversion factor (for aloricate ciliates, which are preserved with 2% Lugol's - 190 fg C μm^{-3}) (Putt & Stoecker, 1989). Biomass of planktonic ciliates can be determined by multiplying the species abundance by the individual mass of ciliates. Calculation of the volume and mass of the cell may be performed by establishing the similarity of the ciliate cell to different geometric figures.

Additional methodological information on ciliates can be obtained from manuals for microzooplankton sampling, fixation, and staining (e.g. Gifford & Caron, 2000; Strüder-Kypke et al., 2003; http://www.liv.ac.uk/ciliate/intro.htm).

3.4. Identification and counting of meso- and macrozooplankton

Species identification and counting are the basis of any community analysis. These procedures are time-consuming and require considerable professional experience. This fact often restricts the number of samples that can be analysed with an acceptable effort within a reasonable time span. Attempts to overcome these difficulties by the automatic counting methods may help to solve the problem of under-sampling (Wiebe & Benfield, 2003). However, application of the automatic methods is limited to uniform samples (e.g. cultures in laboratories), to certain size-class specific analyses, or to a coarse separation of organisms from larger taxonomic groups with significant differences in general body morphology. Coupling of such procedures with computerised image analysis may be helpful; however, it is still linked with sophisticated technical equipment and a need of special software.

Routinely, for monitoring purposes counting is performed for the dominant organisms from easily identifiable taxonomic groups, and their developmental stages. More taxonomic skills are required for the identification of certain species. The species names should be used according to the *International Code of Zoological Nomenclature* (http://www.iczn.org). Information on the validity of names and actual taxonomic classification can be given, for example, after the Integrated Taxonomic Information System (http://www.itis.gov) and The European Register of Marine Species (http://www.marbef.org/data/erms.php).

The laboratory procedure of sorting mesozooplankton starts with removing the carcinogenic formalin from the sample by its filtration through meshes smaller than the mesh size of the sampling gear. The filtrated preservative is used again after the analysis for any further storage. The organisms are suspended in filtered tap water or distilled water for analysis. The procedure should be carried out under a fume-hood. The dilution of the total sample will be chosen according to experience to reach an appropriate concentration for the analysis. The sample is often so densely concentrated that it demands sub-sampling into aliquots. Therefore, 1/32 ± 1/8 of the sample were analysed, for example, within the monitoring programme of the Leibniz Institute of Baltic Sea Research in 2005 (Wasmund et al., 2006). Technically, the volume of the total sample, measured in a graduated cylinder, is noted as the reference amount. The sample is then poured into a beaker to allow a thorough mixing until the organisms are distributed randomly before taking an aliquot. Repeated sub-sampling by the Stempel pipette (Hensen, 1887) produces a coefficient of variation of 7-9%, applying a bulb pipette – of 14-15%, and a Folsom splitter – of 5-18%. The variability between total counts amounts to 0.3–2.5% (Guelpen et al., 1982). The use of pipettes is 5 to 8 times faster than the splitter technique. Its limitation lies in the size of zooplankters in case they are larger than the pipette's diameter. The Kott splitter (Kott, 1953) is more convenient in comparison to the Folsom splitter (Sell & Evans, 1982; Griffiths et al., 1984). The Kott splitter produces 8 sub-samples at the same time, while the Folsom device splits samples into halves and increases the error from one step to another (Behrends & Korshenko, pers. comm.).

For routine sorting of larger zooplankton, a dissecting stereo-microscope will be used. It makes manipulation of the specimen during the identification procedure possible. For the smaller mesozooplankton of the Baltic Sea, such as rotifers, cladocerans, copepods and their developmental stages, an inverted microscope accomplishes the same. It allows routine survey with the 50x magnification and the analysis of details with a magnification factor of 80x to 125x as well. For more specific investigations, like the examination of the fifth leg of copepods, a compound microscope with achromatic condenser and 10x to 70x objectives is the preferred instrument. For details of rotifer morphology, 100x oil immersion objective is necessary.

An inverted microscope needs an open counting chamber with high transparency like the Mini-Bogorov chamber (modified after Arndt, 1985). Closed types of counting chambers are preferably used in microzooplankton studies. The trays are provided with sections to allow a better orientation and to avoid a repeated counting of the same organism. One counting strip is fully covered with the 50x magnification. The Mini-Bogorov chamber (Fig. 3.4.1) is easy to produce in a workshop. It has the following dimensions: the length, width, and height are 40, 70, and 8 mm, respectively. The counting paths are 6 mm deep, their width amounts to 3 mm, the section walls are 1 mm wide, and their height is 4.5 mm. The sides and walls are tapered sloping at top. The tray is made of clear plastic and needs to be polished to high quality (Postel et al., 2000). The table of the microscope has to be adapted to carry the tray (Fig. 3.4.1). The Mini-Bogorov tray is filled with a known aliquot (e.g. 0.5 or 1 ml which has to be considered for calculation of abundance) and finally made up to the top (10 ml) with filtered tap water or distilled water. The surface must be level to avoid any reflections. Therefore, the outer walls are 1.5 mm higher, than those of the counting paths.

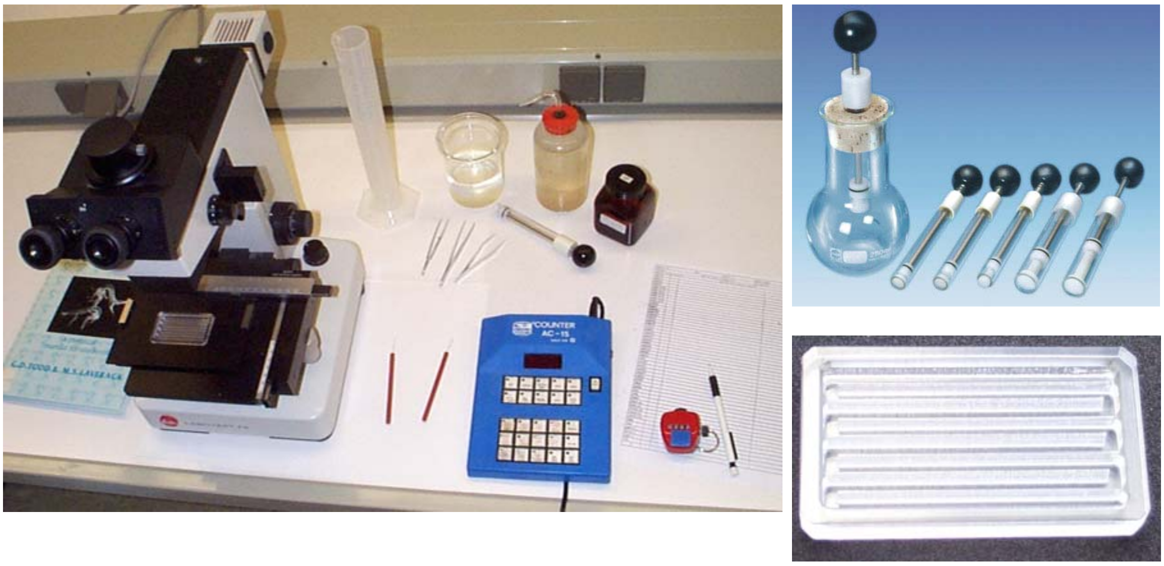


Figure 3.4.1. Working place for counting and identification of smaller mesozooplankton with an inverted microscope (Labovert, Leica Microsystems GmbH, Wetzlar, Germany) and accessory equipments like Stempel pipette (Hydrobios GmbH, Kiel, Germany), and Mini-Bogorov chamber (Postel et al., 2000, modified after Arndt, 1985).

Some organisms, for example, cladocerans, tend to float in the surface film. An addition of detergents or cetyl alcohol [$CH_3(CH_2)_{14}CH_2OH$] (Desmarias, 1997) reduces their surface tension and promotes sinking to bottom. This makes it easier to focus on all animals in the same way. Other sorting media are glycerol and propylene glycol, or lactic acid used for clearing tissues of small crustaceans (Omori & Ikeda, 1984). Contamination of a zooplankton sample by large quantities of phytoplankton makes the analysis more difficult. In this case staining of animals by adding Eosin Y is a helpful tool. A few drops are enough for a 100 ml sample volume. Several hours should be allowed for staining (Edmondson, 1971).

Lund et al. (1958), Cassie (1971) and others have considered the statistical aspects of counting errors, which allow the necessary amount of organisms for counting to be established. The required accuracy of results depends on the purpose of the work. To detect differences between total zooplankton abundance in space or time of 100%, an accuracy of 50% is adequate "and any time spent in making more accurate estimates is largely wasted" (Lund et al., 1958). Generally, an error of ±20% is acceptable. If all organisms are randomly distributed, following the Poisson distribution, the accuracy of a sample and the precision of a single count depends only on the number of specimens counted (Cassie, 1971). The 95% confidence limits ($C.L._{95}$) are calculated from the number of counts (n) and the significance level of the Piosson distribution at the 5% probability error of 1.96:

$$C.L._{95}\,[\%] = \pm\, 1.96\ (100/\sqrt{n})$$

In practice, one or more counting chambers (aliquots) with the same concentration should be analysed until 100 specimens of the most abundant taxonomic groups are reached in a sample (HELCOM, 2005).

The estimations of abundances of the remaining (less common) groups are of lower precision. If the counting procedure is continued until 100 specimens of the other groups are reached, neglecting the more abundant groups, the different sub-sample sizes must be considered in the successive calculations. Finally, the remaining part of the total sample can be surveyed for rare species.

The number of individuals per unit volume of water is defined as abundance. Its calculation (individuals/m^3) needs to consider the number of counts (n), the fraction of the sample counted (k), i.e. the proportion of total volume to sub-sample volume(s), and the amount of water filtered by the sampling net (m^3):

$$\text{Ind./ } m^3 = (n \bullet k) / m^3$$

The need for inter-calibration between joint observation programmes of different laboratories should be emphasised. For example, performing the so-called "ring-test" eight laboratories around the Baltic Sea analysed parts of the same sample (Leppänen et al., 1990). From 15 taxonomic zooplankton groups, 10 were analysed with differences being expected by subtracting the counting error and an error due to the splitting technique when partitioning the total sample. Reasons for the remaining deviations were an insufficient number of organisms counted, the non-random distribution of larger gelatinous individuals in the sample, and taxonomic uncertainties regarding the identification of certain species and developmental stages (nauplii) of copepods.

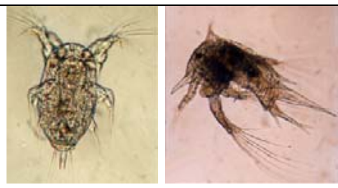
3.5. Biomass determination

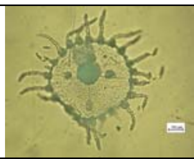
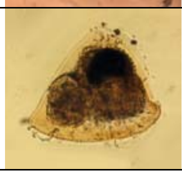
Information about numbers of organisms in a certain volume of water does not provide knowledge about their body mass which is essential for the analyses of trophic webs, energy balance calculations and zooplankton productivity estimation. The calculation of biomass is a way to overcome this problem if suitable individual biomass factors or proper morphometric approaches are applied (for reviews see: Table 4.12 in Postel et al., 2000; Telesh & Heerkloss, 2002, 2004). Such biomass determination is zooplankton specific, in contrast to quantifying the biovolume or other sum biomass parameters of the entire sample by volumetric or other procedures (for details see Postel et al., 2000). The advantage of determining zooplankton biomass using the conversion factors or length/mass correlations is in clear results that can not be falsified by phytoplankton and detritus.

For zooplankton monitoring purposes in the Baltic Sea area, biomass factors were recommended according to Hernroth (1985). This compilation includes individual wet mass of six copepod and three cladoceran taxa basing on volume calculations by morphometric approaches, for example, of Chojnacki and Jankowski (1982), Chojnacki (1983), and Chojnacki (1986) and the successive conversion to wet mass. The compilation was supplemented by literature data for rotifers, chaetognaths, appendicularians and some copepods. Seasonal and regional differences were considered; therefore the amount of data was sufficient. Coarse conversion factors were used to reach comparability. Rough factors may produce significant errors when multiplied by large individual numbers. Therefore, factors and length to mass ratios based on direct measurements should be preferably used. There are some of those available from the Northern Baltic Sea (Kankaala & Johansson, 1986; Kankaala, 1987; Tanskanen, 1994); they are based on the kryo-conservation technique (Latja & Salonen, 1978; Salonen, 1979).

Currently, the Monitoring and Assessment Group of the Helsinki Commission (HELCOM MONAS) is going to include length to carbon ratios and individual carbon factors for the major mesozooplankton species as a standard procedure in the Manual for Marine Monitoring (http://www.helcom.fi/groups/monas/CombineManual/AnnexesC/en_GB/annex7/) basing on the rapid kryo-conservation technique (c.f. Postel et al., 2007).

3.6. Picture key for major zooplankton taxa

No	Character	Taxon	Examples
1	Unicellular	**Ciliata** *(Page 35)*	
1a	Multicellular		2
2	With obvious legs/antennae		3
2a	Without obvious legs/antennae		4
3	Large compound eye; body with carapace of bivalve appearance, segmentation unclear; large antennae	**Cladocera** *(Page 131)*	
3a	Eye small; body elongated or cylindrical, with clear segmentation; antennae usually large	**Copepoda** *(Page 136)*	
3b	Single minute eye spot; body small, unsegmented; with 3 pairs of appendages	**Nauplia** of **Copepoda** *(Page 140)* or **Cirripedia** *(Page 224)*	
4	Body cylindrical or sack-shaped, covered with cuticle or lorica, usually <200 µm; head with ciliated corona	**Rotifera** *(Page 128)*	
4a	Body > 200 µm		5
5	Body segmented, with parapodia and prominent bundles of chaetae	**Polychaeta**, larvae *(Page 146)*	
5a	Body not segmented; without spines		6

No	Character	Taxon	Examples
6	Body oval, elongated or arrow-like		7
6a	Other body shape		8
7	Oval or elongated trunk; long tail with notochord	**Appendicularia** *(Page 144)*	 
7a	Large arrow-like body (15-45 mm) with paired lateral fins	**Chaetognatha** *(Page 142)*	
8	Medusa-like		9
8a	Other shape		10
9	Medusa-like	**Cnidaria** *(Page 123)*	 
9a	Biradially-symmetric comb-jelly-like	**Ctenophora** *(Page 126)*	 
10	Snail-like	**Gastropoda,** larvae *(Page 222)*	
10a	Bivalve-like	**Bivalvia,** larvae *(Page 222)*	
10b	With large projections	**Echinodermata,** larvae *(Page 230)*	
10c	Bell-like	**Bryozoa,** larvae *(Page 230)*	

4. CILIATES OF THE BALTIC SEA

4.1. Brief characteristics of planktonic ciliates of the Baltic Sea

General information

Planktonic ciliates play pivotal roles in the transfer and recycling of carbon and other nutrients in the seas. It is now well established that ciliates constitute an essential food link in marine environments since they remove a significant part of primary and bacterial production and assume ‘trophic repackaging’ of picoplankton particles (0.2–2.0 µm) otherwise inaccessible to mesozooplankton. Picoplankton organisms (heterotrophic bacteria, cyanobacteria, phytoplankton of cell size 0.2–2.0 µm) are the major consumers of dissolved organic matter; their biomass and production is the largest within the total planktonic community. Ciliates, which are among the most important consumers of this production, play a key role as an intermediate stage in the transformation of organic matter from picoplankton to higher trophic levels.

Firstly, ciliates have high growth and metabolic rates. Secondly, they belong to a size range easily removed by metazoans, therefore they constitute a significant part of the food biomass in the sea. Planktonic ciliates make an essential contribution to the nutrition of copepods, rotifers, other invertebrates and fish larvae. Along with phototrophic microorganisms, viruses, heterotrophic bacteria, and microscopic fungi, ciliates participate in a ‘microbial loop’, which is an integral part of the planktonic food webs. Furthermore, the importance of planktonic ciliates as indicators of eutrophication and water pollution has been emphasized.

History of research

Pioneering studies of planktonic ciliates in the Baltic Sea began in the late 1940-s (Biernacka, 1948). Most of them dealt with Tintinnids because of their relatively large size (Biernacka, 1948, 1952; Hedin, 1974, 1975). There were also researches of other groups of ciliates (Bock, 1960; Biernacka, 1963). Investigations of benthic ciliates began earlier, in the late 1920-s (Sauerbrey, 1928; Kahl, 1930-1935). Contemporary investigations of Baltic ciliates deal with both, benthic communities (Klinkenberg & Schumann, 1994; Dietrich & Arndt, 2000; Garstecki et al., 2000), and planktonic ciliates (Smetacek, 1981; Boikova, 1989; Arndt, 1991; Kivi & Setala, 1995; Uitto et al., 1997; Witek, 1998; Setala & Kivi, 2003; Johansson et al., 2004; Samuelsson et al., 2006; Beusekom et al., 2007). Benthic and pelagic communities of ciliates showed little taxonomic overlap (Garstecki et al., 2000). However, benthic ciliates (genus *Euplotes, Aspidisca, Tracheloraphis* etc.) may be found in the water column because of bottom hashing; their role considerably increases in coastal waters and during choppiness (Khlebovich, 1987; Klinkenberg & Shumann, 1994).

Morphology, species richness, dominants

As a rule, planktonic ciliates are smaller than benthic (usually 20-200 µm). They have a strong adoral zone of membranells (AZM), and their somatic cilia, quite on the contrary, are reduced for fewer wisps (Fig. 4.1.1 – 4.1.4). They generally have a typical jumping character of movement.

Altogether, 814 species of ciliates are known for the Baltic Sea, and only 166 of them are truly planktonic (Table 4.2.1).

The dominant group of the Baltic ciliated plankton is small aloricate Oligotrichida (genera *Strombidium, Strobilidium, Lohmaniella*) (Smetacek, 1981; Boikova, 1989; Klinkenberg & Shumann, 1994; Kivi & Setala, 1995; Garstecki et al., 2000; Setala & Kivi, 2003; Johansson et al., 2004; Beusekom et al., 2007). Tintinnids (ciliates with lorica) form another important group of planktonic ciliates (Khlebovich, 1987; Boikova, 1989; Kivi & Setala, 1995; Johansson et al., 2004). Hymenostomatida (mainly small scuticociliates *Cyclidium, Cristigera, Balanion*) and Litostomatea (genus *Mesodinium, Didinium, Monodinium)* are also rather abundant in the water column (Garstecki et al., 2000; Johansson et al., 2004; Samuelsson et al., 2006). Almost the same groups (genera *Strombidium, Mesodinium)* are dominants in the Baltic Sea ice (Granskog et al., 2006). The most species-rich groups among Baltic planktonic ciliates are the aloricate genus *Strombidium* and the loricate genus *Tintinnopsis* (Smetacek, 1981).

Seasonality

Planktonic species of ciliates are distinguished from each other by their ecology (temperature, salinity, food preferences), therefore, the structure of dominant groups varies greatly in different seasons.

According to Johansson et al. (2004), major part of planktonic species revealed in the northern Baltic were seasonal, but some occurred year-round. Tintinnids were found mainly during autumn when the total diversity of ciliates was the highest. In the Bornholm Basin during spring and early summer, *Myrionecta rubra* (a phototrophic ciliate) dominated in the protozooplankton (biomass about 0.2–0.3 mg C/l), whereas during late summer *Helicostomella subulata* and *Strombidium* sp. gained importance with biomass up to 130 mg C/1. In late summer, a second ciliatoplankton peak developed, caused by *H. subulata* (Beusekom et al., 2007). In the northern Baltic Sea, the most abundant ciliates during the summer were oligotrichids from the genera *Strombidium, Strobilidium, Lohmaniella* and *Tintinnopsis* (Kivi & Setala, 1995). In shallow inlets of the southern Baltic, the abundance peak in July was due to a mass development of small scuticociliates (genus *Cyclidium*) and oligotrichids (Garstecki et al., 2000).

Sessilid ciliates also can be numerous in the Baltic plankton, especially in late autumn and early winter (Mamaeva, 1987; Witek, 1998; Johansson et al., 2004). Apparently at this time the peak of abundance of large filamentous cyanobacteria and intensive water mixing provide particles for sessilid ciliates to grow on.

Specificity of assemblages

The proximity of bottom and the availability of hard substrates greatly influence the ciliates' community composition. Near the bottom the ciliate community changes from dominance of the open water *Balanion* to *Euplotes*, which is known to occur in epibenthos (Samuelsson et al., 2006). There are several possible reasons for the decrease of the open water ciliates. It can be caused by an indirect effect due to changes in the prey community, or the excess surface may have influenced their swimming behaviour negatively (Samuelsson et al., 2006). Similar results were obtained by Klinkenberg and Shumann (1994). This study showed that in the bottom layers larger benthic and particle-associated ciliates (e.g. *Euplotes, Oxytricha, Blepharisma)* had developed whereas in the supernatant relatively small ciliates like *Strombidium, Strobilidium, Mesodinium, Halteria* and *Askenasia* had remained present. In the Gdańsk Basin, the deep-water ciliate community composed of *Prorodon*-like ciliates and *Metacystis* sp. also differed from the community of the epipelagic layer (Witek, 1998). Similarly, ciliate population structure in the anoxic depths of the central Baltic Sea (below 120 m) completely differed from those in the upper layers (Detmer et al., 1993). Substancially lesser number of ciliate species was found at this oxic/anoxic interface. Among them several specimens of the genus *Metopus* were recognised (Detmer et al., 1993). The deep-water associations in the Bornholm Basin were composed of larger-sized ciliate species if compared with the upper water layers (Setala & Kivi, 2003).

Indicators

There are several indicator species among the Baltic ciliates (Khlebovich, 1987; Boikova, 1989). For example, the presence of *Tintinnidium fluviatile* in some parts of the Neva Bay means that those waters are oligosaprobic. Such species as *Tintinnopsis cratera* and *Strombidium mirabile* (from the Neva Bay) are also indicators of clean water. Ciliates *Dexiostoma campylum* (Plate 4.3.6), *Colpoda steini*, *Coleps hirtus* (Plate 4.3.1), *Halteria grandinella* (Plate 4.3.13) are, on the contrary, indicators of polluted water (Khlebovich, 1987). The bloom of the autotrophic ciliates *Myrionecta rubra* is the evidence of eutrophication. It should be noted that in the central Bornholm Basin this ciliate dominated during spring and summer reaching maximum biomass of about 0.2 – 0.3 mg C/l (Beusekom et al., 2007).

Feeding modes and strategies

Different feeding modes can be distinguished in ciliates. Most ciliates are heterotrophic organisms, but there are some exceptions. Some planktonic ciliates (especially oligotrichids) are capable of mixotrophic feeding. *Myrionecta rubra* is noted for obligate autotrophy: it contains cryptophycean endosymbionts, which are capable of photosynthesis. In the south-western Gdańsk Basin, the potential annual production of *M. rubra* comprised 6 to 9% of the total primary production. These ciliates were the main contributors to the total biomass of autotrophs also in the Bothnian Bay (Jaanus et al., 2007).

Heterotrophic ciliates can be attributed to groups with different feeding types: microphagous (feeding on bacteria, detritus), phytophagous (feeding on phytoplankton), and predatory carnivorous ciliates (feeding on other ciliates and small metazoans). In the Baltic Sea, ciliates with all feeding modes are present: microphagous (genera *Balanion*, *Cyclidium, Mesodinium*), phytophagous (*Strombidium, Strobilidium*), and carnivorous (*Didinium*).

Each ciliate species shows a specific food particle size preference pattern. Most ciliates are known to prefer prey size 2 to 10 µm (Kivi & Setala, 1995; Samuelsson et al., 2006). The overall ciliate food size spectrum covers the most abundant food items in the Baltic summer plankton. Most species of the Baltic planktonic ciliates effectively ingest nanoflagellate-size food; a minority showed effective grazing on the smallest particles, suggesting a possible ability to utilize bacteria-size prey (Kivi & Setala, 1995).

Among the ciliates, two different feeding strategies appear to be valid: specialistic and generalistic, where the ciliates either concentrate on feeding on a narrow size range of food organisms (*Tintinnopsis lobiancoi, Strombidium conicum* and *Strobilidium* sp.), or use food particles of a wide size range, with little or no preferences within this range (*Lohmaniella oviformis, Strobilidium spiralis, Strombidium* sp.*, Tintinnidium fluviatile, Tintinnopsis beroidea*) (Kivi & Setala, 1995).

Abundance, biomass, distribution

As in the case of community structure, the abundance and biomass of Baltic ciliates show a high degree of spatial and season variability. Spatial variability of ciliate abundance consists of distinctions between different regions of the Baltic Sea, between coastal and open waters, between different layers studied in the water column (euphotic and deep-water layer).

The abundance of ciliates in the Baltic Sea ranges from $1x10^3$ to $88x10^3$ ind./l and biomass varies from 0.023 to 0.3 mg C/l. The seasonal succession of ciliates showed peaks during spring in the northern Baltic Proper (Johansson et al., 2004), during spring and autumn in the northern and western Baltic Sea (Smetacek, 1981; Samuelsson et al., 2006), the Neva Bay (Khlebovich, 1987) and during spring and summer in the south-western

Gdańsk Basin (Witek, 1998). Generally, ciliates are more abundant in the coastal than in the open waters (Khlebovich, 1987; Samuelsson et al., 2006).

The highest abundances of Baltic planktonic ciliates were observed in shallow inlets of the Southern Baltic (0.17–88 x10^3 ind./l) (Garstecki et al., 2000). In the central Bornholm Basin, planktonic ciliates reached biomass of about 0.13–0.3 mg C/l (Beusekom et al., 2007). In the Neva Bay, abundance of ciliates on average was 3x10^3 ind./l, with maximum values in spring (8x10^3 ind./l); biomass ranged from 0.01 mg of wet mass per liter in autumn to 0.74 – in spring (Khlebovich, 1987). In the northern Baltic Proper, biomass of planktonic ciliates did not exceed 0.025 mg C/l (Johansson et al., 2004). In surface waters of the Gulf of Finland, abundance of ciliates constitute 7-20 x10^3 ind./l (Setala & Kivi, 2003). In the western Baltic, maximum biomass (0.056 mg C/l) was registered in spring; cell numbers ranged from 2x10^3 ind./l to 28x10^3 ind./l (Smetacek, 1981).

Vertical distribution of Baltic planktonic ciliates is also rather heterogeneous. For example, in the south-western Gdańsk Basin, ciliate abundance in the euphotic zone was less than 28x10^3 ind./l and biomass 0.023 mg C/l. At the same time, the ciliate biomass in the deep-water layer was similar to the ciliate biomass in the euphotic zone (Witek, 1998). In contrast to Gdańsk Basin, in the shallow waters of the Darss-Zingst ecosystem, less than 50% of ciliate individuals lived in the near-bottom layer, but more than 50% of the biomass (59.7 and 75.5%) was concentrated in this layer. This difference is caused by the composition of ciliates in the bottom layer consisting especially of large benthic and particle-associated forms (Klinkenberg & Shumann, 1994). Rather high numbers of ciliates were encountered even in some anoxic depths of the central Baltic Sea (Detmer et al., 1993). Abundance of ciliates below 120 m depth was found to increase from 160 to 480 ind./l; however, it amounted to only 10% of the surface values of ciliates' abundance.

It should be noted that vertical distribution of ciliates undergo changes not only because of hydrological reasons (e.g., water mixing). Many species of ciliates are capable of vertical migrations. For example, active vertical migrations of autotrophic ciliate *Myrionecta rubra* from deep layers to the euphotic zone were registered during the vernal bloom in the northern Baltic (Olli et al., 1998). It has been suggested that this ciliate can act as peculiar nutrient pump, which makes nutrients available to nonmigrating species.

Factors controlling the ciliate assemblages

Ciliates showed a significant relationship to latitude and salinity, which explains 12–24% of their abundance variation in the northern Baltic Sea (Samuelsson et al., 2006).

Besides the reasons given above, trophic factor (quantity/quality of food and grazers) also exerts much influence on the ciliate abundance.

Several investigators observed that ciliate abundance in the water column was positively correlated with the chlorophyll *a* concentration that increased with productivity and eutrophication (Arndt, 1991; Garstecki et al., 2000; Samuelsson et al., 2006).

As it was shown for different parts of the Baltic Sea, large ciliates would increased in numbers with increasing primary production, while the bacterial production would govern the dynamics of small ciliates (Witek, 1998; Johansson et al., 2004; Samuelsson et al., 2006). Seasonal succession of ciliate community from large predatory ciliates in spring to small microphagous and epibiotic ciliates in summer is typical and was revealed for the northern (Johansson et al., 2004; Samuelsson et al., 2006), southern (Witek, 1998) and western Baltic Sea (Smetacek, 1981).

Even though the growth rate of ciliates in summer seemed to be mainly limited by the amount of the resources in a coastal area, the biomass of ciliates in the northern Baltic Sea was found to be strongly affected by predation of mesozooplankton. Studies in the Baltic Proper suggest that the ciliate biomass is top-down predation-controlled, while the production may be bottom-up limited by the resources (Samuelsson et al., 2006).

Role in zooplankton communities

Analyses of the results of various recent studies in the pelagic regions of the Baltic Sea revealed that the protozoan biomass had been in the same range or even higher than mesozooplankton biomass (Arndt, 1991). For example, in the south-western Gdańsk Basin the heterotrophic ciliate community contributed 10 to 13% to the mean annual zooplankton biomass (Witek, 1998). In the Neva Bay, nearly 16% of the total destruction of organic matter accounted for planktonic ciliates (Khlebovich, 1987). Interestingly, it exceeded the joint decomposition of organic substances by rotifers, cladocerans and copepods (10%). Daily average ciliate production was 0.25 mg/l or 50 mg C/m^2 per day. It formed nearly 19% of primary production of phytoplankton and about 30% of bacterial production (Khlebovich, 1987). In the Gdańsk Basin, carbon demand of non-predatory ciliates calculated according to their potential production was estimated to be equivalent to 12-15% of the gross primary production (Witek, 1998).

Some unresolved research problems in ecology of Baltic ciliates

During the last decades many researches have been involved in the study of functions and role of ciliates in the Baltic pelagic ecosystems. Thus, major interest has been centered around the structure of microbial loop. Several studies were devoted to experimental investigation of nutrition of Baltic ciliates and mainly concerned with their role as predators (Kivi & Setala, 1995; Aberle et al., 2007; Moorthi et al., 2008). In particular, these investigations deal with food spectrum, herbivore selectivity and grazing impact of some common Baltic planktonic ciliates. In addition, there are some studies of trophic interactions between different levels of the Baltic Sea ecosystems which provide information about abundance and productivity of phytoplankton, microzooplankton including ciliates, and mesozooplankton (Uitto et al., 1997; Johansson et al., 2004). This ecosystem approach presents a picture of complex relationships within microbial community and opens wide range of unresolved research problems in ecology of ciliates. For example, our knowledge about functioning of microbial loop is rather schematic yet, and it is not clear enough how it changes spatially and temporally. Several other investigations provide information on the role of ciliates in food webs within poorly-studied anoxic enviroments of the Baltic Sea, with emphasis on microbial loop structure (Setala, 1991; Detmer et al., 1993). One more widely discussed question of ecology of Baltic ciliates is their contribution to the diet of different mesozooplankton species (Tiselius, 1989; Schmidt et al., 2002).

It is necessary to mention that our knowledge concerning the diversity, abundance and distribution of larger ciliates is more complete than that of smaller species, whereas particularly nanociliates (<20 μm) form the most abundant and productive component of the pelagic ciliate community. This situation is partly due to the fact that the recognition of taxonomically useful morphological characters in larger forms by means of light and electron microscopy is much easier than in smaller ciliates. Larger forms usually remain more intact throughout sampling, preservation and examination procedures. At present several molecular approaches are being developed for practical identification of tiny ciliate species which may impact our understanding of the biodiversity and biogeography of small protists.

The nano-component of the ciliate community is especially important in the coastal waters that are significantly stressed by industry and recreation. The indicative role of nanociliates is of great value for the ecosystem state evaluation due to their exceptional sensitivity to eutrophication and pollution.

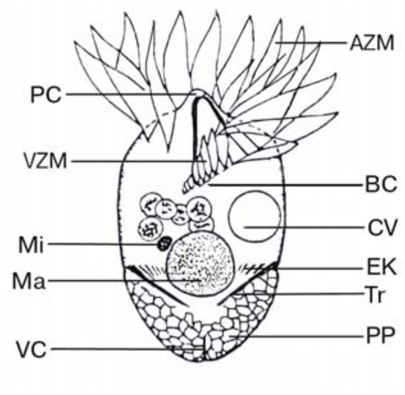


Fig. 4.1.1

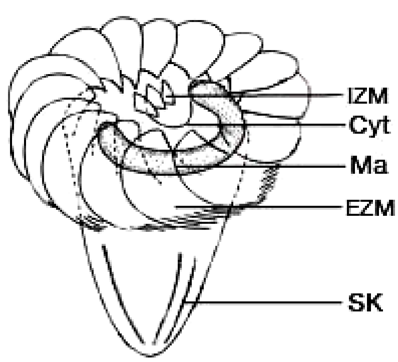


Fig. 4.1.2

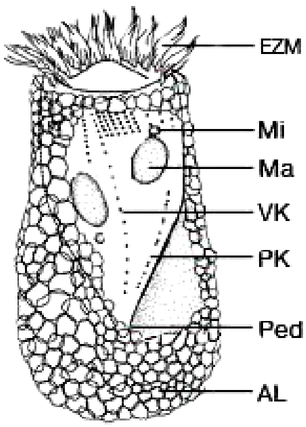


Fig. 4.1.3

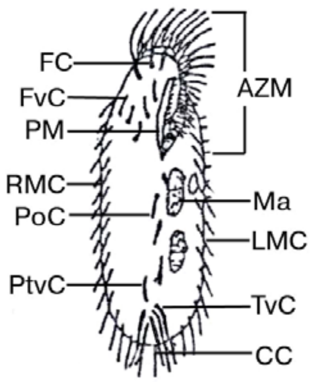


Fig. 4.1.4

Figures 4.1.1-4.1.3. Typical planktonic ciliates: **4.1.1.** *Strombidium sulcatum*, **4.1.2** - *Strobilidium* sp., **4.1.3.** *Tintinnopsis lobiancoi*. AL agglomerated lorica, AZM adoral zone of membranelles, BC buccal cavity, CV contractile vacuole, Cyt cytostome, EK equatorial kinety, EZM external zone of membranelles, IZM internal zone of membranelles, Ma macronucleus, Mi micronucleus, PC peristomial collar, Ped peduncle, PK posterior kinety, PP polygonal cortical platelet, SK somatic kinety, Tr trichites, VC ventral cleft, VK ventral kinety, VZM ventral zone of membranelles (**Fig. 4.1.1**, modified from Maeda & Carey, 1985; **Fig. 4.1.2, 4.1.3**, modified from Strüder-Kypke et al., 2003).

Figure 4.1.4. *Sterkiella histriomuscorum*, a typical benthic ciliate: CC caudal cirri, FC frontal cirri, FvC frontoventral cirri, LMC left marginal cirri, PM paroral membrane, PoC postoral cirri, PtvC pretransverse cirri, RMC right marginal cirri, TvC transverse cirri (modified from Foissner & Berger, 1996).

4.2. Checklist of ciliates of the Baltic Sea

Table 4.2.1.
Species composition of planktonic and benthic ciliates in the Baltic Sea (**BP** – Baltic Proper; **WBS** – Western Baltic Sea; **NBS** – Northern Baltic Sea, **SBS** – Southern Baltic Sea; **EBS** – Eastern Baltic Sea; "+" present; no sign = species not found; species **in bold** are illustrated by photographs).

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
1	*Acaryophrya collaris* Kahl, 1926 (Syn.*: *A. mamillata* Kahl, 1927; *Balanophrya collaris* Kahl, 1926; *Holophrya collaris* (Kahl, 1926) Dingfelder, 1962)		+		+	
2	*Acineta* sp.		+			
3	*Acineta amphiasci* Precht, 1935		+			
4	*Acineta compressa* Claparede & Lachmann, 1859 (Syn.: *A. cucullus* Claparede & Lachmann, 1860; *A. papillifera* Keppen, 1888)				+	
5	*Acineta foetida* Maupas, 1881		+		+	
6	*Acineta laomedeae* Precht, 1935		+			
7	*Acineta pyriformis* Stokes, 1891				+	
8	*Acineta schulzi* Kahl, 1934		+			
9	*Acineta sulcata* Dons, 1927 (Syn.: *A. benesaepta* Schulz, 1933)		+			
10	*Acineta tuberosa* Ehrenberg, 1834		+	+	+	
11	*Amphileptus inquieta* Biernacka, 1963				+	
12	***Amphileptus pleurosigma*** (Stokes, 1884) Foissner, 1984 (Syn.: *Hemiophrys pleurosigma* (Stokes, 1884) Kahl, 1931; *Litonotus pleurosigma* Stokes, 1884)					+
13	*Amphileptus trachelioides* Zacharias, 1893					+
14	*Amphisiella annulata* Kahl, 1932 (Syn.: *Holosticha annulata* Kahl, 1928)		+		+	
15	*Amphisiella marioni* Wicklow, 1982		+			
16	*Amphisiella milnei* Kahl, 1932		+		+	
17	***Amphisiella oblonga***** (Schewiakoff, 1893) Kahl, 1930-5 (Syn.: *Tetrastyla oblonga* Schewiakoff, 1893)					+
18	*Amphorella* sp.[P]		+			
19	*Amphorides quadrilineata*[P] Claparede & Lachmann, 1858 (Syn.: *Tintinnus quadrilineatus* Claparede & Lachmann, 1858)		+			
20	*Anigsteinia longissima* Kahl, 1928		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
21	*Anigsteinia salinaria* Kahl, 1928		+			
22	*Anophrys sarcophaga* Cohn, 1866		+			
23	*Anteholosticha arenicola* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha arenicola* Kahl, 1932; *Biholosticha arenicola* Dragesco, 1963)		+			
24	*Anteholosticha fasciola* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha fasciola* Kahl, 1932)		+			
25	*Anteholosticha grisea* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha grisea* Kahl, 1932)		+			
26	*Anteholosticha manca* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha manca* Kahl, 1932)		+			
27	***Anteholosticha monilata*** (Kahl, 1928) Berger, 2003 (Syn.: *Holosticha extensa* Kahl, 1932; *H. monilata* Kahl, 1928; *Keronopsis monilata* Kahl, 1928)		+			+
28	*Anteholosticha multistilata* (Kahl, 1932) Berger, 2003 (Syn.: *Keronopsis multistilata* Kahl, 1928; *Holosticha multistilata* Kahl, 1932)		+			
29	*Anteholosticha pulchra* (Kahl, 1932) Berger, 2003 (Syn.: *Keronopsis pulchra* Kahl, 1932)		+		+	
30	*Anteholosticha scutellum* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha scutellum* Cohn, 1866)		+			
31	*Anteholosticha violaceae* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha violacea* Kahl, 1928)		+			
32	*Apiosoma* sp.	+				
33	*Aristerostoma marinum* Kahl, 1931		+			
34	*Ascobius simplex* Dons, 1918 (Syn.: *Semifolliculina simplex* Dons, 1918)		+			
35	*Askenasia* sp.[P]		+			+
36	*Askenasia stellaris*[P] (Leegaard, 1920) Kahl, 1930	+	+			+
37	*Aspidisca* sp.		+		+	
38	*Aspidisca aculeata* Ehrenberg, 1838 (Syn.: *A. aculeata* Mansfeld, 1926)		+			
39	*Aspidisca angulata* Bock, 1952		+			
40	*Aspidisca binucleata* Kahl, 1932		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
41	*Aspidisca cicada* Muller, 1786 (Syn.: *A. sulcata* Kahl, 1932; *Coccudina costata* Dujardin, 1841; *Trichoda cicada* Muller, 1786)		+			
42	*Aspidisca dentata* Kahl, 1928		+			
43	*Aspidisca fusca* Kahl, 1928 (Syn.: *A. irinae* Burkovsky, 1970)		+			
44	*Aspidisca leptaspis* Fresenius, 1865 (Syn.: *A. baltica* sensu Borror, 1968; *A. caspica* Agamaliev, 1967; *A. crenata* Fabre-Domergue, 1885; *A. hexeris* Quennerstedt, 1869; *A. lyncaster* sensu Fleury et al., 1986; *A. orthopogon* Deroux & Tuffrau, 1965; *A. psammobiotica* Burkovsky, 1970; *A. pulcherrima* Kahl, 1932; *A. pulcherrima* f. *baltica* Kahl, 1932; *A. sedigita* Quennerstedt, 1867)		+		+	
45	*Aspidisca lyncaster* (Muller, 1773) Stein, 1859 (Syn.: *Trichoda lyncaster* Muller, 1773)		+			
46	***Aspidisca lynceus***** Muller, 1773 (Syn.: *Trichoda lynceus* Muller, 1773)					+
47	*Aspidisca major* f. *faurei* Dragesco, 1960		+			
48	*Aspidisca mutans* Kahl, 1932		+			
49	*Aspidisca polypoda* Dujardin, 1841 (Syn.: *A. quadrilineata* Kahl, 1932)		+			
50	*Aspidisca polystyla* Stein, 1859 (Syn.: *A. plana* Perejaslawzeva, 1886)		+			
51	*Aspidisca robusta* Kahl, 1932		+			
52	*Aspidisca steini* Buddenbrock, 1920 (Syn.: *A. aculeata* sensu Agamaliev, 1974; *A. aculeata* sensu Borror, 1965; *A. glabra* Kahl, 1928; *A. hyalina* Dragesco, 1960)		+		+	
53	***Aspidisca turrita*** (Ehrenberg, 1831) Claparede & Lachmann, 1858 (Syn.: *Euplotes turritus* Ehrenberg, 1831; *E. turritus* Ehrenberg, 1838)		+			+
54	*Atopochilodon arenifer* Kahl, 1933		+			
55	*Atopochilodon distichum* Deroux, 1976		+			
56	*Australothrix gibba* Claparede & Lachmann, 1858 (Syn.: *Holosticha gibba* (Muller, 1786) Stein, 1859; *Oxytricha gibba* Claparede & Lachmann, 1858)		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
57	*Australothrix zignis* Entz, 1884 (Syn.: *Uroleptus zignis* Entz, 1884)		+		+	
58	*Avelia gigas* Dragesco, 1960		+			
59	*Balanion* sp.[P]			+		
60	*Balanion comatum*[P] Wulff, 1922	+	+		+	
61	*Balladyna elongata* Roux, 1901		+			
62	*Biholosticha discocephalus* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha discocephalus* Kahl, 1932)		+			
63	*Blepharisma* sp.		+		+	
64	*Blepharisma clarissimum* Kahl, 1928 (Syn.: *Anigsteinia clarissimum* Kahl, 1928)		+		+	
65	*Blepharisma dileptus* Kahl, 1928		+			
66	*Blepharisma hyalinum* Perty, 1852 (Syn.: *B. lateritium* f. *minima* Roux, 1902)		+			
67	*Blepharisma salinarum* Florentin, 1899		+		+	
68	*Blepharisma steini* Kahl, 1932 (Syn.: *B. lateritium* Claparede & Lachmann, 1858)		+			
69	*Blepharisma tardum* Kahl, 1928		+			
70	*Blepharisma undulans* Stein, 1868					+
71	*Blepharisma vestitum* Kahl, 1928		+			
72	*Bursella spumosa*[P] Schmidt, 1921					+
73	*Caenomorpha levanderi* Kahl, 1927		+			
74	*Calyptotricha lanuginosa* (Penard, 1922) Wilbert & Foissner, 1980		+			
75	*Carchesium gammari* Precht, 1935		+			
76	*Carchesium jaerae* Precht, 1935		+			
77	*Carchesium pectinatum* (Zacharias, 1897) Kahl, 1935 (Syn.: *Zoothamnium limneticum* Svec, 1897; *Z. pectinatum* Zacharias, 1897)					+
78	***Carchesium polypinum***** (Linnaeus, 1758) Ehrenberg, 1830 (Syn.: *Carchesium corymbosum* Penard, 1922; *Sertularia polypina* Linnaeus, 1758)					+
79	*Carchesium spectabile* Claparede & Lachmann, 1858 (Syn.: *Carchesium lachmanni* Kent, 1881)		+			
80	*Carchesium steinii* Wrzesniowski, 1877 (Syn.: *Epistylis steinii* Wrzesniowski, 1877)		+			
81	*Cardiostomatella mononucleata* Dragesco, 1960		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
82	*Cardiostomatella vermiforme* (Kahl, 1928) Corliss, 1960		+		+	
83	*Caudiholosticha setifera* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha setifera* Kahl, 1932; *Holosticha obliqua* Kahl, 1928)		+			
84	*Caudiholosticha viridis* (Kahl, 1932) Berger, 2003 (Syn.: *Holosticha viridis* Kahl, 1932)		+			
85	*Certesia quadrinucleata* Fabre-Domergue, 1885 (Syn.: *C. ovata* Vacelet, 1960*)*		+			
86	*Chaenea gigas* Kahl, 1933		+			
87	*Chaenea robusta* Kahl, 1930		+			
88	*Chaenea simulans* Kahl, 1930		+			
89	*Chaenea teres* Dujardin, 1841 (Syn.: *C. elongata* Kahl, 1926; *C. limicola* Kahl, 1928; *Enchelys stricta* Dujardin, 1841)		+		+	
90	*Chaenea vorax* Quennerstedt, 1867 (Syn.: *Lagynus elongatus* Maupas, 1883)		+			
91	***Chilodonella bavariensis***** Kahl, 1931					+
92	***Chilodonella calkinsi*** Kahl, 1928 (Syn.: *C. pediculatus* Kahl, 1928; *Chlamydonellopsis calkinsi* Kahl, 1928)		+		+	+
93	*Chilodonella cyprini* (Moroff, 1902) Strand, 1928					+
94	*Chilodonella helgolandica* Kahl, 1935		+		+	
95	*Chilodonella nana* Kahl, 1928					+
96	*Chilodonella rigida* Kahl, 1933		+			
97	*Chilodonella subtilis* Kahl, 1933		+			
98	*Chilodontopsis caudata* Kahl, 1933		+			
99	***Chilodontopsis depressa***** (Perty, 1852) Blochmann, 1895 (Syn.: *Chilodon depressus* Perty, 1852)					+
100	*Chilodontopsis elongata* (Kahl, 1928) Corliss, 1960		+		+	
101	*Chilodontopsis oblonga* Maupas, 1883		+			
102	*Chilodontopsis ovalis* Biernacka, 1963				+	
103	*Chilodontopsis vorax* (Stokes, 1886) Kahl, 1931		+			
104	*Chlamydodon cyclops* Entzsen, 1884		+			
105	*Chlamydodon major* (Kahl, 1931) Carey, 1994		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
106	*Chlamydodon mnemosyne* Ehrenberg, 1838 (Syn.: *C. apsheronica* Aliev, 1987; *C. pedarius* Kaneda, 1953)		+			
107	*Chlamydodon obliquus* Kahl, 1931		+			
108	*Chlamydodon triquetrus* Muller, 1786 (Syn.: *Chilodon auricula* Gourret & Roeser, 1887; *Chlamydodon erythrorhynchus* Perejaslawzewa, 1885; *Colpoda triquetrus* Muller, 1786; *Chlamydodon kasymovi* Aliev, 1987)		+			
109	*Ciliofaurea arenicola* Dragesco, 1960		+			
110	*Ciliofaurea mirabilis* Dragesco, 1960		+			
111	***Cinetochilum margaritaceum***** Perty, 1852 (Syn.: *Cyclidium margaritaceum* Ehrenberg, 1830; *Glaucoma margaritaceum* Claparede & Lachmann, 1858)					+
112	*Climacostomum gigas* Meunier, 1907		+			
113	*Climacostomum virens* Ehrenberg, 1833 (Syn.: *Bursaria virens* Ehrenberg, 1833; *Leucophrys curvilata* Stokes, 1886; *Spirostomum virens* Ehrenberg, 1838)				+	
114	*Codonella* sp.[P]				+	
115	*Codonella cratera*[P] Leidy, 1877	+				
116	*Codonella lagenula*[P] Claparede & Lachmann, 1858			+	+	
117	*Codonella orthoceras*[P] (Haeckel, 1873) Joergensen, 1924 (Syn.: *C. orthoceras* (Haeckel, 1873) Kofoid & Campbell, 1929)		+			
118	*Codonella relicta*[P] Minkiewich, 1905			+	+	
119	*Codonellopsis* sp.[P]				+	
120	*Codonellopsis contracta*[P] Kofoid & Campbell, 1929			+	+	
121	*Codonellopsis orthoceros*[P] Haeckel, 1873	+				
122	*Cohnilembus* sp.		+			
123	*Cohnilembus stichotricha* Kahl, 1928		+			
124	*Cohnilembus vermiformis* Kahl, 1931		+			
125	*Cohnilembus verminus* (Muller, 1786) Kahl, 1933		+			
126	*Coleps* sp.	+	+			
127	*Coleps arenarius* Bock, 1952		+			
128	*Coleps bicuspis* Noland, 1925		+			
129	***Coleps elongatus***** Ehrenberg, 1830					+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
130	***Coleps hirtus*** (Muller, 1786) Nitzsch, 1827 (Syn.: *Cercaria hirta* Nitsch, 1817; *Cercaria hirta* Muller, 1786; *Coleps incurvus* Ehrenberg, 1841; *Coleps viridis* Ehrenberg, 1838; *Dictiocoleps hirtus* Diesign, 1866; *Vorticella punctata* Abildgaard, 1793)				+	+
131	*Coleps pulcher* Spiegel, 1926		+			
132	*Coleps remanei* Kahl, 1933		+		+	
133	*Coleps similis* Kahl, 1933		+		+	
134	*Coleps spiralis* Noland, 1937		+			
135	*Coleps tesselatus* Kahl, 1930 (Syn.: *Cercaria hirta* Muller, 1786)		+			
136	***Colpidium*** sp.					+
137	***Colpidium kleini**** Foissner, 1969					+
138	*Colpoda cucullus* Muller, 1786 (Syn.: *C. lucida* Greeff, 1883; *Kolpoda cucullus* Muller, 1773; *Tillina flavicans* Stokes, 1885)					+
139	*Conchostoma longissimum* Faure-Fremiet, 1963		+			
140	*Condylostoma arenarium* Spiegel, 1926		+		+	
141	*Condylostoma magnum* Spiegel, 1926				+	
142	*Condylostoma minima* Dragesco, 1960				+	
143	*Condylostoma patens* Muller, 1786 (Syn.: *Trichoda patens* Muller, 1786)		+			
144	*Condylostoma patulum* Claparede & Lachmann, 1858		+		+	
145	*Condylostoma remanei* Spiegel, 1928 (Syn.: *C. caudatum* Spiegel, 1926; *C. longissima* Kahl, 1928)		+		+	
146	*Condylostoma rugosa* Kahl, 1928		+			
147	*Condylostoma tardum* Penard, 1922				+	
148	*Condylostoma tenuis* Faure-Fremiet, 1958		+			
149	*Condylostoma vorticella* Ehrenberg, 1833 (Syn.: *C. stagnale* Wrzesniowski, 1870; *Linostomella vorticella* Ehrenberg, 1833)					+
150	*Copemetopus subsalsus* Villeneuve-Brachon, 1940	+				
151	*Corynophrya campanula* Kahl, 1934		+		+	
152	*Corynophrya marina* Kahl, 1934		+		+	
153	*Cothurnia arcuata* Mereschkowsky, 1879	+		+		

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
154	*Cothurnia borealis* (Hensen, 1890) Ostenfeld, 1916 (Syn.: *Amphorides borealis* Hensen,1890; *Tintinnus borealis* Hensen, 1890)			+		+
155	*Cothurnia ceramicola* Kahl, 1933		+			
156	*Cothurnia cordylophorea* Kahl, 1933		+			
157	*Cothurnia cypridicola* Kahl, 1933		+		+	
158	*Cothurnia gammari* Precht, 1935		+			
159	*Cothurnia harpactici* Kahl, 1933		+			
160	*Cothurnia maritima* Ehrenberg, 1838		+		+	
161	*Cothurnia ovalis* Kahl, 1933		+		+	
162	*Cothurnia pedunculata* Dons, 1918 (Syn.: *C. nodosa* Mereschkowsky, 1879)		+			
163	*Cothurnia recurva* Claparede & Lachmann, 1858		+			
164	*Cothurnia simplex* Kahl, 1933		+			
165	*Coxliella helix*[P] Claparede & Lachmann, 1858		+		+	+
166	*Coxliella helix* f. *cochleata*[P] Brandt, 1907		+	+	+	
167	*Craspedomyoschiston sphaeromae* Precht, 1935		+			
168	*Cristigera cirrifera*[P] Kahl, 1928 (Syn.: *Cristigera vestita* Kahl, 1928)		+			
169	*Cristigera media*[P] Kahl, 1928		+			
170	*Cristigera minuta*[P] Kahl, 1928		+			
171	*Cristigera penardi*[P] Kahl, 1935 (Syn.: *C. pleuronemoides* Penard, 1922)		+			
172	*Cristigera phoenix*[P] Penard, 1922		+			
173	***Cristigera setosa***[P] Kahl, 1928		+		+	+
174	*Cristigera sulcata*[P] Kahl, 1928		+			
175	*Cryptopharynx* sp.			+		
176	*Cryptopharynx setigerus* Kahl, 1928		+			
177	***Ctedoctema acanthocryptum***[P] Stokes, 1884 (Syn; *Ctedoctema acanthocrypta* Stokes, 1884)		+			+
178	*Cyclidium* sp.[P]		+			
179	***Cyclidium candens***[P] Kahl, 1928		+		+	+
180	***Cyclidium citrullus***[P] Cohn, 1865		+			+
181	*Cyclidium elongatum*[P] Schewiakoff, 1896 (Syn.: *C. glaucoma* f. *elongatum* Schewiakoff, 1896)				+	
182	*Cyclidium flagellatum*[P] Kahl, 1926		+			
183	*Cyclidium fuscum*[P] Kahl, 1935		+			
184	***Cyclidium glaucoma***[P] Muller, 1773		+			+
185	*Cyclidium plouneouri*[P] Dragesco, 1963		+			
186	*Cyclidium simulans*[P] Kahl, 1928		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
187	*Cyclidium veliferum*[P] Kahl, 1933		+			
188	*Cyclidium xenium*[P] Fenchel et.al, 1995		+			
189	*Cyclotrichium cyclokaryon*[P] Meunier, 1907		+			
190	*Cyclotrichium ovatum*[P] Faure-Fremiet, 1924		+			
191	***Cyrtolophosis mucicola***** Stokes, 1885 (Syn.: *Balantiophorus minutus* Schewiakoff, 1889)					+
192	***Dexiostoma campylum***** Ganner & Foissner, 1989 (Syn.: *Colpidium campylum* (Stokes, 1886-Bresslau, 1922) Kahl, 1931; *Dexiostoma campyla* (Stokes, 1886) Jankowski, 1967; *Cryptochilum griseolum* f. *marium* Gourret & Roeser, 1866; *Glaucoma colpidium* Schewiakoff, 1896; *Tillina campyla* Stokes, 1886)					+
193	*Dictyocysta elegans*[P] Ehrenberg, 1854	+				
194	*Didinium* sp.[P]				+	
195	*Didinium balbiani* f. *rostratum*[P] Kahl, 1926 (Syn.: *D. nasutum* f. *rostratum* Kahl, 1926)					+
196	*Didinium gargantua*[P] Meunier, 1907	+	+		+	
197	*Didinium nasutum*[P] (Muller, 1773) Stein, 1859 (Syn.: *Chytridium steini* Eberhard, 1862; *Vorticella nasuta* Muller, 1773)	+	+	+	+	+
198	*Dileptus* sp.				+	
199	*Dileptus anser* (Muller, 1786) Dujardin, 1841 (Syn.: *Amphileptus anser* Ehrenberg, 1838; *Amphileptus cygnus* Claparede & Lachmann, 1859; *Amphileptus margaritifer* Ehrenberg, 1838; *Dileptus gigas* f. *grojecensis* Wrzesniowsky, 1870; *Dileptus gigas* f. *varsaviensis* Wrzes., 1870; *Dileptus irregularis* Maskell, 1888; *Vibrio anser* Muller, 1786)					+
200	*Dileptus cygnus* Claparede & Lachmann, 1859					+
201	*Dileptus estuarinus* Dragesco, 1960		+			
202	*Dileptus marinus* Kahl, 1933		+		+	
203	*Dileptus massutii* Kahl, 1933		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
204	*Diophryopsis hystrix* (Buddenbrock, 1920) Hill & Borror, 1992 (Syn.: *Diophrys hysrix* Buddenbrock, 1920)		+			
205	*Diophrys* sp.		+			
206	*Diophrys appendiculata* (Ehrenberg, 1838) Kahl, 1932 (Syn.: *D. hystrix* Buddenbrock, 1920; *D. multicirratus* Alekperov, 1984; *D. pentacirratus* Alekperov, 1984; *Stylonychia appendiculata* Ehrenberg, 1838)	+	+		+	
207	*Diophrys scutum* Dujardin, 1841 (Syn.: *D. peloetes* Borror, 1963; *D. quadricaudatus* Agamaliev, 1967; *D. scutoides* Agamaliev, 1967)		+		+	
208	*Discocephalus ehrenbergi* Dragesco, 1960		+			
209	*Discocephalus rotatorius* Ehrenberg, 1828		+		+	
210	*Discotricha papillifera* Tuffrau, 1954		+			
211	*Disematostoma butschlii* Lauterborn, 1894 (Syn.: *Leucophrys ovum* Faure-Fremiet, 1924)					+
212	*Dysteria calkinsi* Kahl, 1931 (Syn.: *D. lanceolata* Calkins, 1902)		+			
213	*Dysteria marioni* Gourret & Roeser, 1887		+			
214	*Dysteria monostyla* (Ehrenberg, 1838) Kahl, 1931		+		+	
215	*Dysteria navicula* Kahl, 1928		+			
216	*Dysteria ovalis* Gourret & Roeser, 1886 (Syn.: *Aegyria angustata* f. *ovalis* Gourret & Roeser, 1886)		+			
217	*Dysteria procera* Kahl, 1931		+			
218	*Dysteria pusilla* Claparede & Lachmann, 1859		+			
219	*Dysteria sulcata* Claparede & Lachmann, 1858 (Syn.: *Trochilia sulcata* Claparede & Lachmann, 1858)		+			
220	*Enchelyodon elegans* Kahl, 1926 (Syn.: *Spathidium elegans* Kahl, 1926)		+		+	
221	*Enchelyodon elongatus* Claparede & Lachmann, 1859		+			
222	*Enchelyodon fascinucleatus* Kahl, 1933		+			
223	*Enchelyodon laevis* Quennerstedt, 1869		+			
224	*Enchelyodon sulcatus* Kahl, 1930		+		+	

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
225	*Enchelyodon trepida* (Kahl, 1928) Borror, 1965 (Syn.: *Trachelocerca trepida* Kahl, 1928; *Pseudotrachelo-cerca trepida* (Kahl, 1928) Song, 1990)		+			
226	*Enchelys marina* Meunier, 1907				+	
227	*Enchelys pectinata* Kahl, 1930		+			
228	*Enchelys tarda* Quennerstedt, 1869		+			
229	*Epaxiella* sp.				+	
230	*Ephelota gemmipara* Hertw., 1876		+			
231	*Epiclintes ambiguus* Muller, 1786 (Syn.: *E. auricularis* (Claparede & Lachmann, 1858) Stein, 1864; *E. felis* (Muller, 1786) Carey & Tatchell, 1983)		+		+	+
232	*Epimecophrya ambiguus* Kahl, 1933		+			
233	*Epimecophrya cylindrica* Kahl, 1933		+			
234	*Epistylis* sp.		+			+
235	*Epistylis arenicolae* Fabre-Domergue, 1888 (Syn.: *Rhabdostyla arenicolae* Fabre-Domergue, 1888)		+			
236	*Epistylis caliciformis* Kahl, 1933		+			
237	*Epistylis carcini* Precht, 1935		+			
238	*Epistylis gammari* Precht, 1935		+			
239	*Epistylis harpacticola* Kahl, 1933		+			
240	***Epistylis hentscheli*** ** Kahl, 1935					+
241	*Epistylis nitocrae* Precht, 1935		+			
242	***Epistylis plicatilis*** Ehrenberg, 1838					+
243	*Epistylis rotans* Svec, 1897			+		+
244	*Eucamptocerca longa* Cunha, 1907		+			
245	*Euplotes* sp.		+	+	+	
246	***Euplotes affinis*** Dujardin, 1842 (Syn.: *E. affinis* f. *tricirrata* Kahl, 1931; *Ploesconia affinis* Dujardin, 1841)	+	+			+
247	*Euplotes balteatus* Kahl, 1932 (Syn.: *E. quinquecarinatus* Gelei, 1950; *E. alatus* Kahl, 1932)		+			
248	*Euplotes balticus* (Kahl, 1932) Dragesco, 1966		+		+	
249	*Euplotes crassus* Dujardin, 1841 (Syn. : *Euplotes crassus* f. *minor* Kahl, 1932; *Euplotes vannus* f. *balticus* Kahl, 1932; *E. taylori* Garnjobst, 1928)				+	
250	*Euplotes cristatus* Kahl, 1932		+		+	
251	*Euplotes elegans* Kahl, 1932 (Syn.: *Euplotes bisulcatus* Kahl, 1932)		+			
252	*Euplotes gracilis* Kahl, 1932		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
253	*Euplotes harpa* Stein, 1859 (Syn.: *Ploesconia cithara* Dujardin, 1842)		+		+	
254	*Euplotes moebiusi* Kahl, 1932		+		+	
255	*Euplotes patella* Ehrenberg, 1838 (Syn.: *Coccudina keromina* Bory, 1824; *Euplotes carinatus* Stokes, 1885; *E. leticiensis* Bovee, 1957; *E. paradoxa* Kent, 1880; *E. patella* f. *lemani* Dragesco, 1960; *E. viridis* Ehrenberg, 1838; *Trichoda patella* Muller, 1773)		+			+
256	*Euplotes trisulcatus* Kahl, 1932		+		+	
257	*Euplotes vannus* (Muller, 1786) Minkjewicz, 1901 (Syn.: *E. caudatus* Meunier, 1907; *E. crassus* sensu Tuffrau, 1960; *E. longipes* Claparede & Lachmann, 1859; *E. marioni* Gourret & Roeser, 1886; *E. minuta* sensu Agamaliev, 1971; *E. mutabilis* Tuffrau, 1960; *E. roscoffensis* Dragesco, 1966; *E. sharuri* Aliev, 1986; *E. worcesteri* Griffin, 1910)		+		+	
258	*Fabrea salina*[P] Henneguy, 1890		+			
259	*Favella ehrenbergi*[P] Claparede & Lachmann, 1858			+	+	
260	*Favella serrata*[P] Moebius, 1887		+			
261	*Folliculina ampula* Muller, 1773 (Syn.: *Vorticella ampulla* Muller, 1786; *Ascobius lentus* Henneguy, 1884; *Folliculina moebiusi* Hadzi, 1951)	+	+		+	
262	*Folliculina gigantea* Dons, 1917		+			
263	*Frontonia algivora* Kahl, 1931		+			
264	*Frontonia arenaria* Kahl, 1933		+			
265	*Frontonia atra* Ehrenberg, 1833 (Syn.: *Ophryoglena atra* Ehrenberg, 1833)		+		+	
266	***Frontonia elliptica*** Beardsley, 1902 (Syn.: *F. fusca* Quennerstedt, 1869)		+			+
267	*Frontonia leucas* (Ehrenberg, 1833) Ehrenberg, 1838 (Syn.: *Bursaria leucas* Ehrenberg, 1833; *Frontonia vermalis* Ehrenberg, 1883; *Ophryoglena magna* Maupas, 1883; *O. vorax* Smith, 1897; *Plagiopyla hatchi* Stokes, 1891)		+			
268	*Frontonia macrostoma* Dragesco, 1960		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
269	*Frontonia marina* Fabre-Domergue, 1891 (Syn.: *F. leucas* f. *marina* Florentin, 1899)		+		+	
270	*Frontonia microstoma* Kahl, 1935		+			
271	*Frontonia nigricans* Penard, 1922		+			
272	*Frontonia pallida* Czapik, 1979				+	
273	*Frontonia vacuolata* Dragesco, 1960		+			
274	*Gastrostyla pulchra* (Perejaslawzewa, 1885) Kahl, 1932 (Syn.: *Holosticha coronata* Gourret & Roeser, 1887; *Keronopsis coronata* Gourret & Roeser, 1887)		+			
275	*Geleia decolor* Kahl, 1933		+		+	
276	*Geleia fossata* Kahl, 1933		+		+	
277	*Geleia nigriceps* Kahl, 1933		+			
278	*Geleia orbis* Faure-Fremiet, 1951 (Syn.: *Parduczia orbis* (Faure-Fremiet 1950) Dragesco, 1999)		+			
279	***Glaucoma scintillans*** Ehrenberg, 1830					+
280	*Gruberia* sp.		+			
281	*Gruberia lanceolata* Gruber, 1884		+			
282	*Gruberia uninucleata* Kahl, 1932		+			
283	*Gymnozoon viviparum* Meunier, 1907		+			
284	***Halteria grandinella***[P] (Muller) Dujardin, 1841 (Syn.: *H. chlorelligera* Kahl, 1935 f. *grandinelloides* Margalef-Lopez, 1945; *Trichoda grandinella* Muller, 1773; *T. grandinella* (Muller, 1773) Ehrenberg, 1830)	+	+		+	+
285	*Haplocaulus furcellariae* Precht, 1935		+			
286	*Haplocaulus nicoleae* Precht, 1935		+			
287	*Hartmannula acrobates* (Entz, 1884) Poche, 1913		+			
288	*Hartmannula entzi* Kahl, 1931		+			
289	*Helicoprorodon gigas* (Kahl, 1933) Faure-Fremiet, 1950		+			
290	*Helicoprorodon minutus* Bock, 1952		+			
291	*Helicostoma buddenbrocki*[P] Kahl, 1931		+		+	
292	*Helicostoma notatum*[P] Kahl, 1931		+			
293	*Helicostoma oblongum*[P] Cohn, 1866	+				
294	*Helicostomella edentata*[P] Ehrenberg, 1833	+				
295	*Helicostomella kiliensis*[P] Laackmann, 1906	+				

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
296	*Helicostomella subulata*^{P} Ehrenberg, 1833 (Syn.: *Amphorella subulata* Daday, 1887; *Tintinnus subulatus* Ehrenberg, 1833; *T. ussowi* Mereschkowsky, 1879)	+	+	+	+	+
297	*Helicostomella subulata* f. *kiliensis*^{P} Laackmann, 1906		+			
298	*Heliochona scheuteni* Stein, 1854		+			
299	*Heliochona sessilis* Plate, 1888		+			
300	*Heminotus caudatus* Kahl, 1933		+		+	
301	*Hemiophrys* sp.				+	
302	*Hemiophrys agilis* Penard, 1922 (Syn.: *Amphileptus agilis* Penard, 1922)		+		+	
303	*Hemiophrys filum* Gruber, 1884 (Syn.: *Amphileptus filum* Gruber, 1884)		+		+	
304	*Hemiophrys fusidens* Kahl, 1926		+			
305	*Hemiophrys marina* Kahl, 1930 (Syn.: *Amphileptus marinus* (Kahl, 1931) Song, Wilbert & Hu, 2004)		+		+	
306	*Hemiophrys rotunda* Kahl, 1930 (Syn.: *Lionotus lamella* f. *rotundus* Kahl, 1926)		+			
307	*Hippocomos loricatus* Czapik & Jordan, 1977				+	
308	*Histiobalantium majus*^{P} Kahl, 1931		+			
309	*Histiobalantium marinum*^{P} Kahl, 1933		+			
310	*Histiobalantium natans*^{P} Claparede & Lachmann, 1858 (Syn.: *Pleuronema inflatum* Lauterborn, 1915)				+	
311	*Histriculus similis* Quennerstedt, 1867 (Syn.: *Stylonychia similis* Quennerstedt, 1867)				+	
312	***Histriculus vorax***** (Stokes, 1891) Corliss, 1960 (Syn.: *Histrio vorax* Stokes, 1891)					+
313	*Holophrya* sp.	+				
314	*Holophrya biconica* Sauerbrey, 1928		+			
315	*Holophrya coronata* Morgan, 1925 (Syn.: *Trachelocerca coronata* De Morgan, 1925)		+			
316	*Holophrya lemani* Dragesco, 1960 (Syn.: *Prorodon teres* f. *lemani* Dragesco, 1960)		+			
317	*Holophrya nigricans* Lauterborn, 1894				+	+
318	*Holophrya simplex* Schewiakoff, 1893					+
319	*Holophrya sulcata* Penard, 1922				+	

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
320	*Holophrya tarda* Quennerstedt,1869		+			
321	***Holosticha brevis***** Kahl, 1932 (Syn.: *Keronopsis longicirrata* Gelei & Szabados, 1950; *Holosticha rostrata* Vuxanovici, 1963)					+
322	*Holosticha diademata* (Rees, 1884) Kahl, 1932 (Syn.: *Amphisia diademata* Rees, 1883; *Amphisiella thiophaga* Kahl, 1928; *Holosticha teredorum* Tucolesco, 1962; *H. thiophaga* Kahl, 1928; *H. milnei* Kahl, 1932; *H. teredorum* Tucolesco, 1962; *H. coronata* Vuxanovici, 1963)		+			
323	*Holosticha kessleri* Wrzesniowski, 1877 (Syn.: *Oxytricha kessleri* Wrzesniowski, 1877)		+		+	
324	***Holosticha pullaster***** (Muller, 1773) Foissner et al., 1991 (Syn.: *H. danubialis* Kaltenbach, 1960; *H. kessleri* f. *aquae-dulcis* Buchar, 1957; *H. retrovacuolata* Tucolesco, 1962; *H. rhomboedrica* Vuxanovici, 1963; *H. simplicis* Wang & Nie, 1932; *Oxytricha alba* Fromental, 1876; *Trichoda pullaster* Muller, 1773)					+
325	*Homalozoon caudatum* Kahl, 1935		+			
326	***Homalozoon vermiculare*** Stokes, 1887 (Syn.: *Craspedonotus vermicularis* (Stokes, 1887) Kahl, 1926; *Leptodesmus tenellus* Zacharias, 1888; *Litonotus vermicularis* Stokes, 1887)				+	+
327	*Intranstylum brachymyon* Precht, 1935		+			
328	*Intranstylum coniferum* Precht, 1935		+			
329	*Kentrophorus* sp.			+		
330	*Kentrophorus fasciolatum* Sauerbrey, 1928		+			
331	*Kentrophorus fistulosus* Faure-Fremient, 1950 (Syn.: *K. longissimus* Dragesco, 1954; *K. tubiformis* Raikov & Kovaleva, 1966)		+			
332	*Kentrophorus lanceolatum* Faure-Fremiet, 1951 (Syn.: *Centrophorella lanceolata* Faure-Fremiet, 1951)				+	
333	*Kentrophorus latum* Raikov, 1962					+
334	*Keronopsis arenivorus* Dragesco, 1954		+			
335	*Keronopsis gracilis* Dragesco, 1965		+			
336	*Keronopsis pernix* Wrzesniowski, 1877		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
337	*Laboea strobila*[P] Lohmann, 1908	+	+			
338	*Lacrymaria* sp.		+		+	+
339	*Lacrymaria acuta* Kahl, 1933		+			
340	*Lacrymaria affinis* Bock, 1952		+		+	
341	*Lacrymaria binucleata* Song & Wilbert, 1989		+			
342	*Lacrymaria caudata* Kahl, 1932		+		+	
343	*Lacrymaria cohni* Kent, 1881		+		+	
344	***Lacrymaria coronata*** Claparede & Lachmann, 1858 (Syn.: *L. caspia* Grimm, 1876; *Phialina coronata* Claparede & Lachmann, 1858)		+		+	+
345	*Lacrymaria cucumis* Penard, 1922 (Syn.: *L. putrina* Kahl, 1926)		+			
346	*Lacrymaria delamarei* Dragesco, 1954		+			
347	*Lacrymaria lagenula* Claparede & Lachmann, 1858		+			
348	*Lacrymaria marina* Kahl, 1933		+		+	
349	***Lacrymaria olor*** Muller, 1776 (Syn.: *L. proteus*; *Trachelocerca filiformis* Maskell, 1886; *Vibrio olor* Muller, 1786)		+			+
350	*Lacrymaria olor* f. *marina* Kahl, 1933		+			
351	*Lacrymaria pupula* Muller, 1786 (Syn.: *L. aquae dulcis* (Roux, 1901) Lauterborn, 1915; *L. coronata* f. *aquae dulcis* Roux, 1901; *L. elliptica* Burger, 1908; *L. phialina* Svec, 1907; *L. phyalina* Penard, 1922; *L. striata* Gulati, 1926)		+			
352	*Lacrymaria salinarum* Kahl, 1928 (Syn.: *Phialina salinarum* Kahl, 1928)		+		+	
353	*Lacrymaria saprorelica* Kahl, 1927		+			
354	*Lacrymaria vermicularis* Muller, 1786 (Syn.: *L. metabolica* Burger, 1908; *L. phialina* Svec, 1897; *L. spiralis* Kahl, 1926; *Phialina viridis* Ehrenberg-Claparede, 1858)		+			
355	*Lagynophrya contractilis* Kahl, 1928		+		+	
356	*Lagynophrya costata* Kahl, 1933		+			
357	*Lagynophrya halophila* Kahl, 1928		+		+	
358	***Lembadion lucens***** (Maskell, 1887) Kahl, 1931 (Syn.: *Thurophora lucens* Maskell, 1887)					+
359	*Leprotintinnus* sp.[P]					+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
360	*Leprotintinnus bottnicus*[P] (Nordqvist, 1890) Jorgensen, 1912 (Syn.: *Tintinnus bottnicus* Nordqvist, 1890; *Codonella bottnica* Levander, 1895)	+		+	+	+
361	*Leprotintinnus pellucidus*[P] Joergensen, 1924			+	+	
362	*Litonotus* sp.		+		+	+
363	***Litonotus alpestris***** Foissner, 1978 (Syn.: *Litonotus mononucleatus* Song Weibo & Wilbert, 1989; Litonotus uninucleatus (Kahl, 1931) Song Weibo & Wilbert, 1989.)					+
364	*Litonotus anguilla* Kahl, 1931		+		+	
365	*Litonotus binucleatus* Kahl, 1933 (Syn.: *L. pictus* f. *binucleatus* Kahl, 1933)		+			
366	***Litonotus cygnus*** (Muller, 1776) Wrzesniowski, 1870 (Syn.: *Gastrotricha folium* Wrzesniowski, 1866; *Lionotus anas* Levander, 1894; *L. anser* Butschli, 1889; *Litonotus wrzesniowskii* Kent, 1882; *Vibrio cygnus* Muller, 1773)		+		+	+
367	*Litonotus duplostriatus* Maupas, 1883				+	
368	*Litonotus fasciola* (Ehrenberg) Wrzesniowski, 1870 (Syn.: *Amphileptus fasciola* Ehrenberg, 1838; *Dileptus fasciola* Fromentel, 1874; *Loxophyllum fasciola* Claparede & Lachmann, 1981; *Vibrio fasciola* Muller 1786)				+	+
369	***Litonotus lamella*** (Ehrenberg, 1829) Schewiakoff, 1896 (Syn.: *Loxophyllum lamella* Claparede & Lachmann, 1861; *Trachelius lamella* Ehrenberg, 1829; *Acineria incurvata* Dujardin, 1841)		+		+	+
370	*Litonotus loxophylliforme* Dragesco, 1960		+			
371	*Litonotus pictus* Gruber, 1884		+			
372	***Litonotus varsaviensis***** Wrzesniowski, 1870 (Syn.: *Gastrotricha varsaviensis* Wrzesniowski, 1866)				+	+
373	*Lohmaniella* sp.[P]		+		+	
374	***Lohmaniella elegans***[P] (Wulff, 1919) Kahl, 1932 (Syn.: *Strobilidium elegans* Wulff, 1919)	+	+		+	+
375	*Lohmaniella oviformis*[P] Leegard, 1915	+	+			+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
376	*Lopezoterenia torpens* (Kahl, 1931) Foissner, 1997 (Syn.: *Trichopelma torpens* Kahl, 1931)		+			
377	*Loxodes* sp.				+	
378	***Loxodes rostrum***** (Muller, 1773) Ehrenberg, 1830 (Syn.: *Kolpoda rostrum* Muller, 1773)					+
379	*Loxophyllum* sp.				+	
380	*Loxophyllum fasciolatum* Kahl, 1933		+		+	
381	*Loxophyllum helus* (Stokes, 1884) Kahl, 1931 (Syn.: *Litonotus helus* Stokes, 1884; *L. verrucosum* Florentin, 1889; *Loxophyllum verrucosum* Florentin, 1889)		+		+	
382	*Loxophyllum kahli* Dragesco, 1960		+			
383	*Loxophyllum levigatum* Sauerbrey, 1928		+			
384	***Loxophyllum meleagris*** (Muller, 1773) Dujardin, 1841 (Syn.: *Kolpoda meleagris* Muller, 1773)		+			+
385	*Loxophyllum multinucleatum* Kahl, 1928		+		+	
386	*Loxophyllum multiplicatum* Kahl, 1928		+			
387	*Loxophyllum multiverrucosum* Kahl, 1933) Carey, 1991 (Syn.: *L. helus* f. *rotundatum* Kahl, 1933)		+			
388	*Loxophyllum niemeccense* Stein, 1859		+			
389	*Loxophyllum pyriforme* Gourret & Roeser, 1886		+			
390	*Loxophyllum serratum* Kahl, 1933		+		+	
391	*Loxophyllum setigerum* Quennerstedt, 1867 (Syn.: *Litosolenus armatus* Stokes, 1893)		+		+	
392	*Loxophyllum trinucleatum* Mansfeld, 1923				+	
393	*Loxophyllum undulatum* Sauerbrey, 1928				+	
394	*Loxophyllum uninucleatum* Kahl, 1928		+			
395	*Loxophyllum variabilis* Dragesco, 1960		+			
396	*Loxophyllum vermiforme* Sauerbrey, 1928		+			
397	*Lynchella aspidisciformis* Kahl, 1933		+			
398	*Lynchella gradata* Kahl, 1933		+			
399	*Magnifolliculina binalata*[P] Uhlig, 1964		+			
400	*Mesodinium* sp.	+				
401	*Mesodinium cinctum*[P] Calkins, 1902		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
402	***Mesodinium pulex***[P] (Claparede & Lachmann, 1859) Stein, 1867 (Syn.: *Halteria pulex* Claparede & Lachmann, 1858; *H. rubra* Lachmann, 1908; *Mesodinium pulex* f. *striata* Gourret & Roeser, 1886)	+	+	+	+	+
403	*Mesodinium pupula*[P] Kahl, 1933		+			
404	***Metacineta mystacina***** (Ehrenberg, 1831) Butschli, 1889 (Syn.: *Acineta mystacina* Ehrenberg, 1831; *Cothurnia mystacina* (Ehrenberg, 1831)					+
405	*Metacystis striata*[P] Stokes, 1893		+			
406	*Metacystis tesselata*[P] Kahl, 1926		+			
407	*Metanophrys durchoni* Puytorac et al., 1974		+			
408	*Metaurostyla marina* Kahl, 1932 (Syn.: *Urostyla marina* Kahl, 1932)		+		+	
409	*Metopus* sp.	+				
410	*Metopus contortus* Quennerstedt, 1867 (Syn. : *Metopides contorta* Quennerstedt, 1867 ; *Metopus bivillus* Tucolesco, 1962 ; *M. sapropelicus* Tucolesco, 1962)		+		+	
411	*Metopus es* (O.F.Muller, 1786) Kahl, 1932 (Syn.: *M. sigmoides* Claparede & Lachmann, 1858)				+	+
412	*Metopus halophilus* Kahl, 1925		+			
413	*Metopus hyalinus* (Kahl, 1927) Kahl, 1935 (Syn.: *M. laminarius* f. *hyalinus* Kahl, 1927)		+			
414	*Metopus major* Kahl, 1932		+			
415	*Metopus nivaaensis* Esteban, Fenchel & Finlay, 1995		+			
416	*Metopus pellitus* (Kahl, 1932) Carey, 1994		+			
417	*Metopus setosus* Kahl, 1927 (Syn.: *M. setifer* Kahl, 1935)		+			
418	*Metopus verrucosus* Cunha, 1915		+			
419	*Metopus vestitus* Kahl, 1932 (Syn.: *M. caudatus* Cunha, 1915)		+			
420	*Microdysteria aplanata* Kahl, 1933		+			
421	*Micromitra brevicaudata* Kahl, 1933		+			
422	*Microregma ponticum* Lepsi, 1926		+			
423	***Microthorax*** sp.		+			+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
424	***Monodinium balbiani*** [P] Fabre-Domergue, 1888 (Syn.: *Didinium balbianii* Fabre-Domergue, 1888)		+			+
425	*Myelostoma bipartitum* Kahl, 1933		+			
426	*Myoschiston balanorum* Mereschkowsky, 1877 (Syn.: *Epistylis balanorum* Mereschkowsky, 1877)		+			
427	*Myoschiston carcini* Precht, 1935		+			
428	*Myoschiston centropagidarum* Precht, 1935		+			
429	*Myoschiston duplicatum* Precht, 1935		+			
430	*Myriokaryon lieberkuhnii* Jankowski, 1973 (Syn.: *Pseudoprorodon lieberkuhni* Butschli, 1889; *Cranotheridium elongatus* Penard, 1922)					+
431	*Myrionecta rubra* [P] (Lohmann, 1908) Jancowski, 1976 (Syn.: *Halteria rubra,* Lohmann, 1908; *Mesodinium rubrum* Lohmann, 1908)	+	+	+	+	+
432	*Nassula argentula* Kahl, 1930		+			
433	*Nassula aurea* Ehrenberg, 1833		+			+
434	*Nassula citrea* Kahl, 1930		+			
435	*Nassula labiata* Kahl, 1933		+			
436	*Nassula notata* Muller, 1786		+			
437	*Nassula ornata* Ehrenberg, 1833		+			
438	*Nassula tumida* Maskell, 1887 (Syn.: *N. ambigua* f. *tumida* Maskell, 1887)				+	
439	*Omegastrombidium elegans* [P] Florentin, 1901) Agatha, 2004 (Syn.: *Strombidium elegans* Florentin, 1899)	+	+		+	
440	*Opercularia nutans* (Ehrenberg, 1831) Stein, 1854 (Syn.: *Epistylis nutans* Ehrenberg, 1831; *Opercularia allensi* Stokes, 1887)				+	
441	*Ophryoglena* sp.		+			
442	*Opistotricha* sp.		+			
443	*Opisthostyla sertularium* Kent, 1881		+			
444	*Orthodon gutta* Cohn, 1866		+			
445	*Oxytricha* sp.		+		+	+
446	*Oxytricha chlorelligera* Kahl, 1932		+			
447	*Oxytricha discifera* Kahl, 1932		+			
448	*Oxytricha halophila* Kahl, 1932		+		+	
449	*Oxytricha marina* Kahl, 1932		+		+	
450	*Oxytricha ovalis* Fromentel, 1876				+	
451	*Oxytricha oxymarina* Berger, 1999 (Syn.: *Steinia marina* Kahl, 1932)		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
452	***Oxytricha setigera***** Stokes, 1891 (Syn.: *Steinia balladynula* Kahl, 1932)					+
453	*Oxytricha tricornis* Milne, 1886		+			
454	*Parablepharisma bacteriophora* Kahl, 1935		+			
455	*Parablepharisma chlamydophorum* Kahl, 1935		+			
456	*Parablepharisma collare* Kahl, 1935		+			
457	*Parablepharisma pellitum* Kahl, 1935		+			
458	*Paracineta divisa* Fraipont, 1878		+			
459	*Paradileptus conicus* Wenrich, 1929					+
460	*Paradiophrys irmgard* (Mansfeld, 1923) Jankowski, 1978 (Syn.: *Diophrys irmgard* Mansfeld, 1923)		+			
461	*Paradiophrys kahli* (Dragesco, 1963) Foissner, 1996 (Syn.: *Diophrys kahli* Dragesco, 1963)		+			
462	*Parafavella* sp.[P]			+	+	
463	*Parafavella cylindrica*[P] (Joergensen, 1899) Kofoid & Campbell, 1929 (Syn.: *Cyttarocylis denticulata* f. *cylindrica* Joergensen, 1899)	+				
464	*Parafavella lachmanni*[P] Daday, 1887	+				
465	*Parafavella media*[P] Brandt, 1896	+				
466	*Paramecium* sp.		+		+	
467	***Paramecium aurelia*** Ehrenberg, 1838 (Syn.: *P. aurelia* Dujardin, 1841)					+
468	*Paramecium bursaria* (Ehrenberg, 1831) Focker, 1836 (Syn.: *Loxodes bursaria* Ehrenberg, 1831)					+
469	*Paramecium calkinsi* Woodruff, 1921		+		+	
470	*Paramecium caudatum* Ehrenberg, 1833 (Syn.: *P. aurelia* Muller, 1786)		+		+	+
471	*Paramecium duboscqui* Chatton & Brachon, 1933					+
472	***Paramecium putrinum*** Claparede & Lachmann, 1858				+	+
473	*Paramecium woodruffi* Wenrich, 1928		+		+	
474	*Paranassula brunnea* Fabre-Domerge, 1885 (Syn.: *Nassula brunnea* Fabre-Domergue, 1885)		+			
475	*Paranassula microstoma* (Claparede & Lachmann, 1859) Kahl, 1931		+			
476	*Paranophrys marina* Thompson & Berger, 1965		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
477	*Paraspathidium longinucleatum* Czapik & Jordan, 1976				+	
478	*Paraspathidium obliquum* Dragesco, 1963				+	
479	*Pelagostrobilidium spirale*[P] Petz et al, 1995 (Syn.: *Lohmanniella spiralis* Leegaard, 1915; *Strobilidium spiralis* (Leegaard, 1915) Petz et al., 1995)	+	+	+		+
480	*Peritromus faurei* Kahl, 1932		+			
481	*Peritromus montanus* Kahl, 1932		+			
482	*Placus buddenbrocki* Sauerbrey, 1928 (Syn.: *Spathidiopsis buddenbrocki* Sauerbrey, 1928)		+			
483	***Placus luciae***** Kahl, 1926 (Syn.: *Thoracophrya luciae* Kahl, 1926)					+
484	*Placus socialis* Fabre-Domergue, 1889 (Syn.: *Spathidiopsis socialis* Fabre-Domergue, 1889)	+				
485	*Placus striatus* Cohn, 1866 (Syn.: *Spathidiopsis striatus* (Cohn, 1866) Corliss, 1979)		+		+	
486	*Plagiocampa* sp.		+			
487	*Plagiocampa acuminata* Kahl, 1933		+			
488	*Plagiocampa incisa* Kahl, 1933		+			
489	*Plagiocampa margaritata* Kahl, 1930		+			
490	*Plagiocampa multiseta* Kahl, 1930		+		+	
491	*Plagiocampa posticeconica* Kahl, 1932		+			
492	*Plagiocampa rouxi* Kahl, 1932		+			
493	*Plagiopogon loricatus* Kahl, 1931		+		+	
494	*Plagiopyla frontata* Kahl, 1935		+			
495	*Plagiopyla marina* Kahl, 1933 (Syn.: *Plagiopyla nasuta* Lynch, 1930)		+			
496	*Plagiopyla nasuta* Stein, 1860 (Syn.: *Parameicum cucullio* Quenn, 1867)		+			+
497	*Plagiopyla ovata* Kahl, 1931		+		+	
498	*Plagiopyla vestita* Kahl, 1935		+			
499	*Platyfolliculina sahrhageana* Hadzi, 1938		+			
500	*Platynema denticulatum* Kahl, 1933		+			
501	*Platynematum hyalinum* Kahl, 1933		+			
502	*Platynematum sociale* Penard, 1922		+			
503	*Pleuronema coronatum*[P] Kent, 1881		+	+	+	
504	*Pleuronema crassa*[P] Dujardin, 1841		+			
505	*Pleuronema marinum*[P] Dujardin, 1841		+		+	
506	*Pleuronema setigerum*[P] Calkins, 1903		+			
507	*Pleuronema smalli*[P] Dragesco, 1968		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
508	*Podophrya halophila* Kahl, 1934		+		+	
509	*Porpostoma notatum* Mobius, 1888				+	
510	*Proboscidium armatum* Meunier, 1907				+	
511	*Prorodon* sp.		+		+	
512	*Prorodon binucleatus* Buddenbrock, 1920		+			
513	*Prorodon brachyodon* Kahl, 1927		+		+	
514	*Prorodon elegans* Kahl, 1928		+			
515	*Prorodon luteus* Kahl, 1930		+			
516	*Prorodon marinus* Claparede & Lachmann, 1858				+	
517	*Prorodon mimeticus* Kahl, 1930		+			
518	*Prorodon moebiusi* Kahl, 1930		+			
519	*Prorodon morgani* Kahl, 1930		+			
520	*Prorodon opalescens* Kahl, 1928		+			
521	*Prorodon ovum* (Ehrenberg, 1833) Kahl, 1930 (Syn.: *Encheyls ovum* Dies, 1866; *Holophrya atra* Svec, 1897; *H. discolor* Ehrenberg, 1833; *H. ovum* Ehrenberg, 1831; *Prorodon nucleatus* Svec, 1897; *P. rigidus* Burger, 1908)		+		+	
522	*Prorodon platyodon* Blochmann, 1895				+	
523	*Prorodon raabei* Capki, 1965				+	
524	*Prorodon teres* Ehrenberg, 1833 (Syn.: *P. griseus* Claparede & Lachmann, 1858; *P. limnetis* Stokes, 1886)				+	
525	*Protocruzia contrax* Mansfeld, 1923 (Syn.: *Blepharisma minima* Lepsi, 1926; *Protocruzia adhaerens* Mansfeld, 1923; *P. depressa* Ammermann, 1968)		+			
526	*Protocruzia granulosa* (Kahl, 1932) Faria, Cunha & Pinto, 1922		+			
527	*Protocruzia labiata* Kahl, 1932		+			
528	*Protocruzia pigerrima* (Cohn, 1866) Cunha, 1914		+			
529	*Protrachelocerca fasciolata* Sauerbrey, 1928 (Syn.: *Tracheloraphis fasciolatus* Sauerbrey, 1928; *Trachelocerca fasciolata* Sauerbrey, 1928; *Tracheloraphis flexuosus* Raikov & Kovaleva, 1968)				+	
530	*Psammomitra brevicauda* (Kahl, 1932) Borror, 1972				+	
531	*Psammomitra retractilis* Borror, 1972		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
532	*Pseudoamphisiella alveolata* (Kahl, 1932) Song & Warren, 2000 (Syn.: *Holosticha alveolata* Kahl, 1932)		+		+	
533	*Pseudoamphisiella lacazei* (Kahl, 1932) Song & Warren, 2000 (Syn.: *Holosticha lacazei* Kahl, 1932)		+			
534	*Pseudoblepharisma tenue* Kahl, 1926		+			
535	*Pseudocohnilembus pussilus* (Quennerstedt, 1869) Foissner & Wilbert, 1981 (Syn.: *Cohnilembus pussillus* (Quennerstedt, 1869) Kahl, 1931)		+			
536	*Pseudodileptus* sp.					+
537	*Pseudokeronopsis carnea* Cohn, 1866		+			
538	*Pseudokeronopsis decolor* Wallengren, 1890 (Syn.: *Keronopsis decolor* Wallengren, 1900; *Holosticha wrzesniowskii* f. *punctata* Rees, 1884)		+			
539	*Pseudokeronopsis flava* (Cohn, 1866) Wirnsberger et al., 1987		+			
540	*Pseudokeronopsis flavicans* (Kahl, 1932) Borror & Wicklow, 1983 (Syn.: *Keronopsis flavicans* Kahl, 1932)				+	
541	*Pseudokeronopsis ovalis* (Wulff, 1919) Johnson, Hargraves & Sieburth, 1988 (Syn.: *Keronopsis ovalis* Kahl, 1932)		+		+	
542	*Pseudokeronopsis rubra* (Ehrenberg, 1838) Borror & Wicklow, 1983 (Syn.: *Holosticha flavorubra* Entz, 1884; *H. rubra* Ehrenberg, 1838; *Keronopsis rubra* Ehrenberg, 1838)		+			
543	*Pseudoplatynematum loricatum* Bock, 1952				+	
544	*Pseudoplatynematum parvum* Bock, 1952				+	
545	*Pseudoprorodon arenicola* Kahl, 1930		+			
546	*Pseudoprorodon halophilus* Kahl, 1930		+			
547	*Pseudoprorodon incisus* Bock, 1952				+	
548	*Pseudoprorodon mononucleatus* Bock, 1952				+	
549	*Pseudovorticella difficilis* Kahl, 1933 (Syn.: *Vorticella difficilis* Kahl, 1933)		+			
550	*Pseudovorticella punctata* (Dons, 1918) Warren, 1986 (Syn.: *Vorticella punctata* Dons, 1918; *V. perlata* Kahl, 1933)		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
551	*Ptychocylis urnula*[P] Claparede & Lachmann, 1858		+			
552	*Ptychocylis minor*[P] Gruber, 1879		+		+	
553	*Quasillagilis constanciensis* Busch, 1920		+			
554	*Remanella* sp.				+	
555	*Remanella brunnea* Kahl, 1933	+	+		+	
556	*Remanella caudata* Dragesco, 1953		+			
557	*Remanella gigas* Dragesco, 1954		+			
558	*Remanella granulosa* Kahl, 1933		+		+	
559	*Remanella margaritifera* Kahl, 1933		+		+	
560	*Remanella minuta* Dragesco, 1954		+			
561	*Remanella multinucleata* Kahl, 1933		+			
562	*Remanella rugosa* Kahl, 1933		+			
563	*Remanella rugosa* f. *unicorpusculata* Kahl, 1933				+	
564	*Remanella swedmarki* Dragesco, 1953		+			
565	*Remanella trichocysta* Dragesco, 1953		+			
566	*Rhabdostyla arenaria* Cuenot, 1891		+		+	
567	*Rhabdostyla commensalis* Moebius, 1888		+			
568	*Rhabdostyla inclinans* (Muller, 1786) D'Udekem, 1864 (Syn.: *R. chaeticola* Stokes, 1887; *R. lumbriculi* Penard, 1922; *Vorticella inclinans* Muller, 1773)				+	
569	*Rhabdostyla libera* Kahl, 1933		+			
570	*Rhabdostyla nereicola* Precht, 1935		+			
571	*Rhabdostyla putrina* (Muller, 1776) Warren, 1986 (Syn.: *Vorticella putrina* Muller, 1776)		+			
572	*Rhabdostyla variabilis* Dons, 1918 (Syn.: *Scyphidia variabilis* Dons, 1918)		+			
573	*Salpingella acuminata*[P] Claparede & Lachmann, 1858		+			
574	*Saprodinium halophila* Kahl, 1935		+			
575	*Scaphidiodon navicula* (Muller, 1786) Stein, 1859		+			
576	*Schistophrya aplanata* Kahl, 1933				+	
577	*Scyphidia gasterostei* Faure-Fremiet, 1905 (Syn.: *Epistylis gasterostei* Faure-Fremiet, 1905)		+			
578	*Scyphidia hydrobiae* Kahl, 1933		+			
579	*Scyphidia physarum* Lohmann, 1856		+			
580	*Sonderia cyclostoma* Kahl, 1930		+		+	
581	*Sonderia macrochilus* Kahl, 1930		+			
582	*Sonderia mira* Kahl, 1930		+			
583	*Sonderia pharyngea* Kirby, 1934		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
584	*Sonderia schizostoma* Kahl, 1930		+			
585	*Sonderia sinuata* Kahl, 1930		+		+	
586	*Sonderia tubigula* Kahl, 1930		+			
587	*Sonderia vestita* Kahl, 1930		+			
588	*Sonderia vorax* Kahl, 1928		+			
589	*Spathidium chlorelligerum* Kahl, 1930		+			
590	*Spathidium curvatum* Kahl, 1928		+			
591	*Spathidium deforme* Kahl, 1928		+			
592	*Spathidium extensum* Kahl, 1933		+			
593	*Spathidium fossicola* Kahl, 1933		+			
594	***Sphaerophrya stentori***** Maupas, 1881					+
595	*Spirostomum ambiguum* (Muller, 1786) Ehrenberg, 1835 (Syn.: *Trichoda ambigua* Muller, 1786)		+			+
596	*Spirostomum loxodes* Stokes, 1885		+			
597	*Spirostomum minus* Roux, 1901 (Syn.: *S. ambiguum* f. *minor* Roux, 1901; *S. intermedium* Kahl, 1932)		+		+	
598	*Spirostomum teres* Claparede & Lachmann, 1859		+		+	+
599	*Spirostrombidium cinctum*[P] (Kahl, 1932) Petz et al., 1995 (Syn.: *Strombidium cinctum* Kahl, 1932)		+			
600	*Spirostrombidium sauerbreyae*[P] Kahl, 1932) Petz et al., 1995 (Syn.: *Strombidium sauerbreyae* (Sauerbrey, 1928) Kahl, 1932)		+		+	
601	*Stenosemella nucula*[P] Joergensen, 1927 (Syn.: *Codonella ventricosa* Entz, 1884; *Tintinnopsis nivalis* Meunier, 1910; *T. nucula* Laackmann, 1906; *T. ventricosa* Daday, 1887)		+			
602	*Stenosemella steinii*[P] Joergensen, 1912		+			+
603	*Stenosemella ventricosa*[P] (Claparede & Lachmann, 1858) Joergensen, 1924 (Syn.: *Codonella ventricosa* Joergensen, 1899; *Tintinnopsis ventricosa* Cleve, 1900; *T. ventricosoides* Meunier, 1910; *Tintinnus ventricosus* Claparede & Lachmann, 1858)		+	+	+	
604	*Stentor auricula* Kent, 1881		+		+	
605	***Stentor coeruleus*** Ehrenberg, 1830 (Syn.: *Brachionus stentoreus* f. *coerulei* Pallas, 1766; *Stentor attenuatus* Maskell, 1888; *S. striatus* Barraud-Maskell, 1886)				+	+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
606	***Stentor muelleri*** (Vincent, 1824) Ehrenberg, 1838				+	+
607	*Stentor multiformis* (Muller, 1786) (Syn.: *Vorticella multiformis* Muller, 1786)		+		+	
608	***Stentor niger*** (Muller, 17 73) Ehrenberg, 1831 (Syn.: *S. pediculatus* Fromentel; *Vorticella nigra* Muller, 1773)		+			+
609	*Stentor polymorphus* (Muller, 1773) Ehrenberg, 1830 (Syn.: *Vorticella polymorpha* Muller, 1773)					+
610	***Stentor roeselii*** Ehrenberg, 1835 (Syn.: *S. gracilis* Maskell, 1886; *S. viridis* Ghosh, 1921)		+		+	+
611	***Sterkiella histriomuscorum***** Foissner, Blatterer, Berger & Kohmann, 1991 (Syn.: *Histriculus muscorum* (Kahl, 1932) Corliss, 1960; *Histrio muscorum* Kahl, 1932; *Opistotricha terrestris* Horvath, 1956; *Oxytricha histrioides* Gellert, 1957; *Stylonychia curvata* Giese & Alden, 1938)		+			+
612	*Stichotricha aculeata* Wrzesniowski, 1866 (Syn.: *S. acuminata* Wang, 1930)		+			
613	*Stichotricha gracilis* Moebius, 1888		+			
614	*Stichotricha marina* Stein, 1867 (Syn.: *S. horrida* Moebius, 1888; *S. inquilinus* Entz, 1884)		+			
615	*Stichotricha mereschkowski* Kahl, 1932		+			
616	*Stichotricha simplex* Kahl, 1932		+			
617	***Stichotricha secunda***** Perty-Stein, 1859					+
618	***Stokesia vernalis*** Wenrich, 1929					+
619	*Stomatophrya aplanata* Kahl, 1933		+			
620	*Stomatophrya singularis* Kahl, 1933		+			
621	*Strobilidium* sp.[P]	+	+	+		+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
622	***Strobilidium caudatum***[P] (Fromental, 1874) Foissner, 1987 (Syn.: *S. adhaerens* Schewiakoff, 1892; *S. caudatum* (Fromental, 1874) Foissner, 1987; *S. caudatum* Kahl, 1932; *S. cometa* (Muller, 1786) Dingfelder, 1962; *S. gyrans* Schewiakoff, 1893 – Deroux, 1974; *Strombidion caudatum* Fromentel, 1876; *Strombidium claparedi* Kent, 1881; *S. gyrans* Stokes f. *transsylvanicum* Lepsi, 1926; *S. intermedium* Maskell, 1887; *S. velox* Beardsley, 1902; *Strombilidium gyrans* Schewiakoff, 1893 – Fernandez-Leborans, 1983; *Turbilina instabilis* Enriques, 1908)		+		+	+
623	*Strobilidium conicum*[P] Kahl, 1932		+			
624	***Strobilidium humile*****[P] (Penard, 1922) Petz & Foissner, 1992					+
625	*Strobilidium minimum*[P] (Gruber, 1884) Kahl, 1932 (Syn.: *Arachnidium becheri* Buddenbrock, 1920; *Strombidium minimum* Gruber, 1884)		+		+	
626	*Strobilidium velox*[P] Faure-Fremiet, 1924 (Syn.: *Rimostrombidium velox* (Faure-Fremiet, 1924) Jankowski, 1978)					+
627	*Strombidinopsis acuminatum*[P] Faure-Fremiet, 1924 (Syn.: *Strombidium typicum* (Lankester, 1874) Butschli, 1889; *S. tintinnodes* Entz, 1884; *S. acuminatum* (Leegaard, 1915) Kahl, 1932; *Laboea acuminata* Leegaard, 1915)	+	+			
628	*Strombidium* sp.[P]	+	+	+	+	+
629	*Strombidium calkinsi*[P] Faure-Fremiet, 1932 (Syn.: *S. caudatum* Fromentel-Calkins, 1902)		+		+	
630	*Strombidium conicum*[P] (Lohmann, 1908) Wulff, 1919 (Syn.: *Laboea acuminata* Leegaard, 1915; *L. conica* Lohmann, 1908; *Strombidium acuminatum* (Leegaard, 1915) Kahl, 1932)	+	+		+	+
631	*Strombidium crassulum*[P] (Leegaard, 1915) Kahl, 1932 (Syn.: *Laboea crassula* Leegaard, 1915)	+	+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
632	*Strombidium delicatissimum*[P] (Leegaard, 1915) Busch, 1921 (Syn.: *Laboea delicatissima* Leegaard, 1915)	+				
633	*Strombidium kahli*[P] Bock, 1952		+		+	
634	*Strombidium latum*[P] Kahl, 1932		+		+	
635	*Strombidium longiceps* Meunier, 1910		+			
636	*Strombidium mirabile*[P] Penard, 1916 (Syn.: *Psilotricha fallax* Zacharias, 1895; *Strombidium fallax* (Zacharias, 1895) Kahl, 1932)					+
637	*Strombidium oblongum*[P] (Entz, 1884) Kahl, 1932 (Syn.: *Clypeolum corsicum* Gourret & Roeser, 1888; *Strombidium corsicum* Gourret & Roeser, 1888; *S. sulcatum* Entz, 1884; *S. sulcatum* f. *oblongum* Entz, 1884)		+		+	
638	*Strombidium oculatum*[P] Faure-Fremiet, 1948		+			
639	*Strombidium purpureum*[P] Kahl, 1932		+			
640	*Strombidium strobilus*[P] (Lochmann, 1908)		+			
641	*Strombidium styliferum*[P] Levander, 1894		+		+	
642	*Strombidium sulcatum*[P] Claparede & Lachmann, 1858 (Syn.: *S. minutum* Wulff, 1919)	+	+		+	
643	*Strombidium vestitum*[P] (Leegaard, 1915) Kahl, 1932 (Syn.: *Laboea delicatissima* Leegaard, 1915; *Laboea vestita* Leegaard, 1915; *Strombidium delicatissimum* (Leegaard, 1915) Kahl, 1932)	+				
644	*Strombidium viride*[P] Stein, 1859 (Syn.: *S. nasutum* Smith, 1897; *Limnostrombidium viride* (Stein, 1867) Krainer, 1995)				+	+
645	*Strombidium viride* f. *pelagica*[P] Kahl, 1932 (Syn.: *S. pelagoviride* (Krainer, 1991) Krainer, 1993; *Limnostrombidium pelagicum* (Kahl, 1932) Krainer, 1995)		+			+
646	*Strongylidium labiatum* Kahl, 1932		+			
647	*Strongylidium muscorum* Kahl, 1932		+			
648	***Stylonychia*** sp.		+			+
649	***Stylonychia mytilus*** Ehrenberg, 1838				+	+
650	*Swedmarkia arenicola* Dragesco, 1954		+			
651	*Tachysoma parvistyla* Stokes, 1887		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
652	***Tachysoma pelionellum*** [P] (Muller, 1773) Kahl, 1932 (Syn.: *Oxytricha pellionella* Muller, 1786)		+			+
653	*Tachysoma rigescens* (Kahl, 1932) Borror, 1972		+			
654	*Tachysoma saltans* (Cohn, 1866) Borror, 1972 (Syn.: *Oxytricha saltans* (Cohn, 1866) Kahl, 1932; *Actinotricha saltans* Cohn, 1866)		+			
655	*Thecacineta* sp.				+	
656	*Thecacineta halacari* Shulz, 1933		+			
657	*Thigmokeronopsis crassa* (Claparede & Lachmann, 1858) Berger, 2006 (Syn.: *Trichotaxis crassa* Claparede & Lachmann, 1858; *Oxytricha crassa* Claparede & Lachmann, 1858)		+			
658	*Thuricola* sp.				+	
659	*Thuricola elegans* Biernacka, 1963				+	
660	*Thuricola obconica* Kahl, 1933				+	
661	*Thuricola valvata* Wright, 1858 (Syn.: *Cothurnia operculata* Gruber, 1879)		+			
662	*Tiarina* sp. [P]		+			
663	*Tiarina borealis* [P] (Dogiel, 1940) Shulman &. Shulman-Albova, 1953	+				
664	*Tiarina fusus* [P] (Claparede & Lachmann, 1858) Bergh, 1881		+		+	
665	*Tintinnidium fluviatile* [P] Stein, 1863 (Syn.: *Tintinnus fluviatile* Stein, 1863)		+			+
666	*Tintinnidium mucicola* [P] Claparede & Lachmann, 1858 (Syn.: *Tintinnus mucicola* Claparede & Lachmann, 1858)	+	+			
667	***Tintinnidium semiciliatum*** ** [P] Sterki, 1879 (Syn.: *Strombidinopsis gyrans* Kent, 1881; *Tintinnidium fluviatile* f. *emarginatum* Maskell, 1887; *Tintinnidium ranunculi* Penard, 1922; *Tintinnus semiciliatus* Sterki, 1879)					+
668	*Tintinnopsis* sp. [P]	+			+	
669	*Tintinnopsis acuminata* [P] Daday, 1887	+				
670	*Tintinnopsis baltica* [P] Brandt, 1896 (Syn.: *T. vasculum* Meunier, 1919; *T. strigosa* Meunier, 1919)	+	+	+	+	+
671	*Tintinnopsis baltica* f. *rotundata* [P] Laackmann		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
672	*Tintinnopsis beroidea*^{P} Stein, 1867 (Syn.: *Codonella beroidea* Entz, 1884; *Tintinnopsis beroidea* f. *acuminata*, Daday, 1887)		+	+	+	+
673	*Tintinnopsis brandti*^{P} Nordqvist, 1890 (Syn.: *Codonella brandti* Nordqvist, 1890)	+		+		+
674	*Tintinnopsis campanula*^{P} Ehrenberg, 1840	+	+	+	+	+
675	*Tintinnopsis cochleata*^{P} Brandt, 1906	+				
676	*Tintinnopsis compressa*^{P} Daday, 1887			+	+	
677	*Tintinnopsis cratera*^{P} Hada, 1936 (Syn.: *Codonella cratera* Kofoid & Campbell, 1929; *C. lacustris* Entz, 1885; *Difflugia cratera* Leidy, 1879; *Tintinnopsis lacustris* Brandt, 1906)					+
678	*Tintinnopsis cylindrata*^{P} Kofoid & Campbell, 1892 (Syn.: *T. cylindrica* Daday, 1892; *T. fusiformis* (Daday, 1892) Entz, 1909)			+	+	
679	*Tintinnopsis fennica*^{P} Kofoid & Campbell, 1929	+				
680	*Tintinnopsis fimbriata*^{P} Meunier, 1919 (Syn.: *T. ventricosa* Levander, 1900)		+	+	+	+
681	*Tintinnopsis karajacensis*^{P} Brandt, 1896		+	+	+	
682	*Tintinnopsis lobiancoi*^{P} Daday, 1887 (Syn.: *T. brasiliensis,* Kofoid & Campbell, 1929)	+		+	+	+
683	*Tintinnopsis lohmanni*^{P} Laackmann, 1906 (Syn.: *T. tubulosa* f. *lohmanni* Joergensen, 1927)		+			
684	*Tintinnopsis major*^{P} Meunier, 1910	+				
685	*Tintinnopsis meunieri*^{P} Kofoid & Campbell, 1929			+	+	
686	*Tintinnopsis minuta*^{P} Wailes, 1925		+	+	+	+
687	*Tintinnopsis nana*^{P} Lohmann, 1908		+			
688	*Tintinnopsis nitida*^{P} Brandt, 1986	+				
689	*Tintinnopsis parvula*^{P} Joergensen, 1912	+				
690	*Tintinnopsis pistillum*^{P} Kofoid & Campbell, 1929	+			+	
691	*Tintinnopsis rapa*^{P} Meunier, 1910	+				
692	*Tintinnopsis rotundata*^{P} Joergensen, 1912	+				
693	*Tintinnopsis sacculus*^{P} Brandt, 1896	+				
694	*Tintinnopsis subacuta*^{P} Joergensen, 1899		+			
695	*Tintinnopsis tubulosa*^{P} Levander, 1900	+	+	+	+	+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
696	*Tintinnopsis turbo*[P] Meunier 1919				+	
697	*Tintinnopsis urnula*[P] Meunier, 1910			+	+	
698	*Tintinnus inquillinum*[P] Muller, 1776	+				
699	*Tokophrya* sp.		+			
700	*Tontonia appendiculariformis* [P] Fauré-Fremiet, 1914		+			
701	*Trachelius gutta* Sahrhage, 1915		+		+	
702	*Trachelius ovum* Ehrenberg, 1831 (Syn.: *Amphileptus ovum* Dujardin, 1841; *A. rotundus* Maskell, 1887; *Harmodirus ovum* Perty, 1852; *Ophryocerca ovum* Ehrenberg, 1831; *Trachelius leidyi* Foulke, 1884)		+			+
703	*Trachelocerca* sp.				+	
704	*Trachelocerca coluber* Kahl, 1933		+		+	
705	*Trachelocerca entzi* Kahl, 1927		+		+	
706	*Trachelocerca fusca* Kahl, 1928 (Syn.: *Paraspathidium fuscum* (Kahl, 1928) Fjeld, 1955)		+		+	
707	*Trachelocerca laevis* Quennerstedt, 1867 (Syn.: *Enchelyodon striatus* Gourret & Roeser, 1886; *Lagynus crassicollis* Maupas, 1883; *L. ornatus* Stokes, 1893; *L. sulcatus* Gruber, 1884; *Trachelocerca sulcata* Kahl, 1927)		+			
708	*Trachelocerca longissima* Kahl, 1928 (Syn.: *Gruvelina longissima* Delphy, 1939)		+			
709	*Trachelocerca phoenicopterus* f. *margaritata* Kahl, 1930		+		+	
710	*Trachelocerca subviridis* Sauerbrey, 1928				+	
711	*Trachelocerca tenuicolis* Quennerstedt, 1867		+			
712	***Trachelophyllum apiculatum*** (Perty, 1852) Claparede & Lachmann, 1859 (Syn.: *Trachelius apiculatus* Perty, 1852; *Trachelophyllum tachyblastum* Stokes, 1884)				+	+
713	*Trachelophyllum brachypharynx* Levander, 1894		+			
714	*Tracheloraphis arenicola* (Sauerbrey, 1928) Dragesco, 1960		+			
715	*Tracheloraphis bimicronucleata* Raikov, 1962					+
716	*Tracheloraphis drachi* Dragesco, 1960				+	

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
717	*Tracheloraphis grassei* Kahl, 1933 (Syn.: *Trachelonema grassei* Dragesco, 1966)		+			
718	*Tracheloraphis griseus* Kahl, 1933 (Syn.: *Trachelocerca grisea* Kahl, 1933)		+			
719	*Tracheloraphis incaudatus* Kahl, 1930 (Syn.: *Trachelocerca incaudata* Kahl, 1933)		+		+	
720	*Tracheloraphis indistincta* Kahl, 1930		+			
721	*Tracheloraphis kahli* Raikov, 1962				+	
722	*Tracheloraphis margaritatus* Kahl, 1930		+		+	
723	*Tracheloraphis oligostriata* Raikov, 1962 (Syn.: *Trachelonema oligostriata* Raikov, 1962)				+	
724	*Tracheloraphis phenicopterus* (Cohn, 1866) Dragesco, 1960 (Syn.: *Trachelocerca phoenicopterus* Cohn, 1866)				+	
725	*Trachelostyla caudata* Kahl, 1932		+			
726	*Trachelostyla pediculiformis* (Cohn, 1866) Kahl, 1932 (Syn.: *Gonostomum ped* Maupas, 1883; *G. pediculiforme* Maupas, 1883; *Stichochaeta corsica* Gourret & Roeser, 1887; *Stichochaeta pediculiformis* Cohn, 1866)		+		+	
727	*Trichodina astericola* Precht, 1935 (Syn.: *Cyclochaeta astericola* Precht, 1935)		+			
728	*Trichodina claviformis* Dobberstein & Palm, 2000			+		
729	*Trichodina domerguei* Wallengren, 1897	+	+			
730	*Trichodina jadranica* Raabe, 1958	+	+			
731	*Trichodina pediculus* Ehrenberg, 1831 (Syn.: *T. baltica* Quennerstedt, 1869)		+			
732	*Trichodina raabei* Lohmann, 1962	+	+			
733	*Trichodina scoloplontis* Precht, 1935		+			
734	*Trichodina serpularum* Fabre-Domergue, 1888 (Syn.: *Cyclochaeta serpularum* Fabre-Domergue, 1888)		+			
735	*Trichophrya piscium* Butschli, 1889					+
736	*Trithigmostoma cucullulus* (Muller, 1786) Jankowski, 1967 (Syn.: *Chilodonella cucullulus* (O. F. Muller, 1786) Kahl, 1931; *Chilodon cucullulus* Ehrenberg-Kelin, 1927; *Kolpoda cucullio* Muller, 1786; *K. cucullulus* Muller, 1786)		+		+	+

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
737	***Trithigmostoma srameki***** (Sramek-Husek, 1952) Foissner, 1987 (Syn.: *Chilodonella hyalina* Sramek-Husek, 1952; *Trithigmostoma hyalina* (Sramek-Husek, 1952) Foissner, 1987)					+
738	***Trochilia minuta***** (Roux, 1901) (Syn.: *Dysteropsis minuta* Roux, 1899)					+
739	*Trochilia sigmoides* Dujardin, 1841				+	
740	*Trochilioides oculata* Kahl, 1933		+			
741	*Trochilioides recta* Kahl, 1928 (Syn.: *Trochilia recta* Kahl, 1928)		+			
742	*Trochilioides striata* Buddenbrock, 1920		+			
743	***Urocentrum turbo***[P] (Muller, 1786) Kahl, 1931 (Syn.: *Calceolus cypripedium* Diesing, 1866; *Cercaria turbo* Muller, 1786; *Peridinopsis cyripedium* Clark, 1866; *Urocentrum trichocystus* Smith, 1897)		+			+
744	*Uroleptopsis citrina* Kahl, 1932		+			
745	*Uroleptopsis viridis* (Perejaslawzewa, 1885) Kahl, 1932		+			
746	*Uroleptus* sp.				+	
747	*Uroleptus musculus* Kahl, 1932 (Syn.: *Holosticha contractilis* Dragesco, 1970; *Paruroleptus musculus* Kahl, 1932)		+			
748	*Uroleptus piscis* (Muller, 1773) Ehrenberg, 1831 (Syn.: *Trichoda piscis* Muller, 1773; *Paruroleptus piscis* Kowalewski, 1882)		+		+	
749	*Uronema* sp.[P]		+			
750	*Uronema elegans*[P] Maupas, 1883		+			
751	***Uronema marinum***[P] Dugardin, 1841 (Syn.: *Loxocephalus putrinus* Kahl, 1926)		+		+	+
752	*Uronema nigricans*[P] (Muller, 1786) Florentin, 1901 (Syn.: *Cyclidium nigricans* Muller, 1786; *Uronema parduczi* Foissner, 1971)		+			
753	*Uronemella filificum*[P] (Kahl, 1931) Song & Wilbert, 2002		+			
754	*Uronychia* sp.		+			
755	*Uronychia heinrothi* Buddenbrock, 1920		+			

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
756	*Uronychia setigera* Calkins, 1902 (Syn.: *U. transfuga* Curds & Wu, 1983; *U. transfuga* Petz, Song & Wilbert, 1995; *U. uncinata* Kahl, 1932; *U. uncinata* Taylor, 1928)		+			
757	*Uronychia transfuga* Muller, 1786		+		+	
758	*Uropedalium pyriforme* Kahl, 1928		+			
759	*Urosoma cienkowskii* Kowalewski, 1882		+			
760	*Urostrongylum* sp.				+	
761	*Urostrongylum caudatum* Kahl, 1932		+		+	
762	*Urostrongylum contortum* Kahl, 1928 (Syn.: *Stichotricha contorta* Kahl, 1928)		+			
763	*Urostrongylum lentum* Kahl, 1932		+			
764	*Urostyla dispar* Kahl, 1932 (Syn.: *Paraurostyla dispar* Kahl, 1932)		+			
765	*Urostyla gracilis* Entz, 1884		+			
766	***Urostyla grandis*** Ehrenberg, 1830 (Syn.: *U. trichogaster* Stokes, 1885)					+
767	*Urotricha* sp.	+				
768	***Urotricha armata***^{P} Kahl, 1927 (Syn.: *U. corlissiana* Song Weibo & Wilbert, 1989; *U. platystoma* Stokes, 1886)		+		+	+
769	*Urotricha baltica*^{P} Czapik & Jordan, 1977				+	
770	*Urotricha globosa*^{P} Schewiakoff, 1892		+			
771	*Urotricha pelagica*^{P} Kahl, 1932		+			+
772	*Vaginicola amphora* Kahl, 1928		+			
773	*Vaginicola crystallina* Ehrenberg, 1830		+			
774	*Vaginicola sulcata* Kahl, 1928		+			
775	*Vaginicola wangi* Kahl, 1935 (Syn.: *Cothurnia acuta* Levander, 1915)	+	+			
776	*Vasicola parvula*^{P} Kahl, 1926		+			
777	***Vorticella*** sp.	+	+	+	+	+
778	*Vorticella anabaena* Stiller, 1940 (Syn.: *V. chlorellata* Stiller, 1940)					+
779	*Vorticella annulata* Gourret & Roeser, 1888		+		+	
780	*Vorticella calisiformis* Kahl, 1933		+			
781	*Vorticella campanula* Ehrenberg, 1831 (Syn.: *V. aperta* Fromentel, 1874)		+			+
782	***Vorticella convallaria*** (Linnaeus, 1758) Linnaeus, 1767 (Syn.: *Hydra convallaria* Linnaeus, 1758)					+
783	*Vorticella dudekemi* Kahl, 1933 (Syn.: *V. patellina* D'Udekem, 1862)		+			
784	*Vorticella fromenteli* Kahl, 1935		+		+	

No	Taxa	BP [1]	WBS [2]	NBS [3]	SBS [4]	EBS [5]
785	*Vorticella fusca* Precht, 1935		+		+	
786	*Vorticella jaerae* Precht, 1935		+			
787	*Vorticella lima* Kahl, 1933		+			
788	*Vorticella longifilum* Kent, 1881				+	
789	*Vorticella marina* Greeff, 1870 (Syn.: *V. constricta* Kahl, 1933)		+		+	
790	*Vorticella mayeri* Faure-Fremiet, 1920 (Syn.: *Pelagovorticella mayeri,* (Faure-Fremiet, 1920) Jankowski, 1980)					+
791	*Vorticella microstoma* Ehrenberg, 1830 (Syn.: *V. infusionum* Dujardin, 1841)		+			
792	*Vorticella nebulifera* Muller, 1786		+		+	
793	*Vorticella octava* Stokes, 1885		+			
794	*Vorticella ovum* Dons, 1917				+	
795	*Vorticella patellina* D'Udekem, 1862 (Syn.: *V. d'udekemi* Kahl, 1933)		+		+	
796	***Vorticella picta***** (Ehrenberg, 1831) Ehrenberg, 1838 (Syn.: *Carchesium pictum* Ehrenberg, 1831)					+
797	*Vorticella striata* Dujardin, 1841		+		+	
798	*Vorticella striatula* Dons, 1915				+	
799	*Vorticella urceolaris* Linnaeus, 1767				+	
800	*Vorticella verrucosa* Dons, 1915				+	
801	*Woodruffia rostrata* Kahl, 1931		+			
802	*Zoothamnium* sp.		+	+	+	+
803	*Zoothamnium alternans* Precht, 1935		+			
804	*Zoothamnium arbuscula* Ehrenberg, 1839 (Syn.: *Z. geniculatum* Ayrton, 1902)				+	
805	*Zoothamnium commune* Kahl, 1933		+		+	
806	*Zoothamnium duplicatum* Kahl, 1933 (Syn.: *Z. kahli* Caspers, 1949)		+		+	
807	*Zoothamnium hentscheli* Kahl, 1935 (Syn.: *Z. kentii* Grenfell, 1884)				+	
808	*Zoothamnium hiketes* Precht, 1935		+			
809	*Zoothamnium hydrobiae* Hofker, 1930		+			
810	*Zoothamnium intermedium* Precht, 1935		+			
811	*Zoothamnium nanum* Kahl, 1933		+			
812	*Zoothamnium nutans* Claparede & Lachmann, 1858				+	
813	*Zoothamnium rigidum* Precht, 1935		+			
814	*Zoothamnium vermicola* Precht, 1935		+			

[1] **BP, Baltic Proper:** after Gaevskaya (1948), Mamaeva (1987), Axelsson & Norrgren (1991), Arndt (1991), Detmer et al. (1993), Wasik et al. (1998),

Setala & Kivi (2003), Johansson et al. (2004), Vannini et al. (2005), Granskog et al. (2006), van Beusekom et al. (2007);

[2] **WBS, Western Baltic Sea** (Kieler Bight): after Sauerbrey (1928), Kahl (1930-1935, 1933), Bock (1960), Fenchel (1967, 1968, 1969), Hirche (1974), Smetacek (1981), Klinkeberg & Shuman (1994), Palm & Dobberstein (2000), Gerlach (2000), Aberle et al. (2007), Moorthi et al. (2008);

[3] **NBS, Northern Baltic Sea** (Archipelago Sea, Bothnian Sea): after Lindquist (1959), Hedin (1974, 1975), Kivi & Setala (1995), Uitto et al. (1997), Olli et al. (1998), Garstecki et al. (2000), Schmidt et al. (2002), Setala (2004), Samuelsson et al. (2006);

[4] **SBS, Southern Baltic Sea** (Gdansk Basin and North-Rugian Bodden): after Biernacka (1948, 1952, 1962, 1963), Czapik & Jordan (1976, 1977), Boikova (1984, 1989), Wiktor & Krajewska-Sołtys (1994), Witek (1998), Jakobsen & Montagnes (1999), Dietrich & Arndt (2000), Rychert (2008);

[5] **EBS, Eastern Baltic Sea** (Gulf of Finland**,** including the freshwater Neva Bay): after Vuorinen et al. (1945), Khlebovich (1987), Kivi & Setala (1995), Smurov & Fokin (1999), Setala (2004), Visse (2007), this study.

(*) – Synonyms

(**) – First record

(^{P}) – Typical planktonic ciliates

4.3. Photo plates: ciliates of the Baltic Sea

Plate 4.3.1

Ciliophora, Prostomatida. Apical cytopharynx is supported by well-developed nematodesmata. **1, 2,** *Placus luciae,* with pellicle furrowed spirally and distinct dorsal brush which terminates in the small depression (**2,** arrow); **3,** *Placus luciae,* with single ovoid macronucleus (arrow), body length 80 µm; **4,** *Coleps hirtus*, barrel-shaped ciliate with fenestrated armour plates, body length 45 µm; **5,** *Coleps elongatus*, cylindrical, a few spines are present in the caudal area, body length 55 µm; **6**, **7, 8,** *Coleps elongatus,* carnivorous ciliates gobbling up *Urocentrum turbo*; **9, 10,** *Urotricha armata*, small ciliate, 35 µm in size, with characteristic single caudal cilium (**9,** arrow), ovoid macronucleus and conspicuous layer of trichocysts underneath pellicle (photos E. Mironova).

Photos **1-3, 9, 10**: live, differential interference contrast (DIC); **4**: live, phase-contrast (PHC); **5-8**: live, bright-field (BF).

Plate 4.3.1

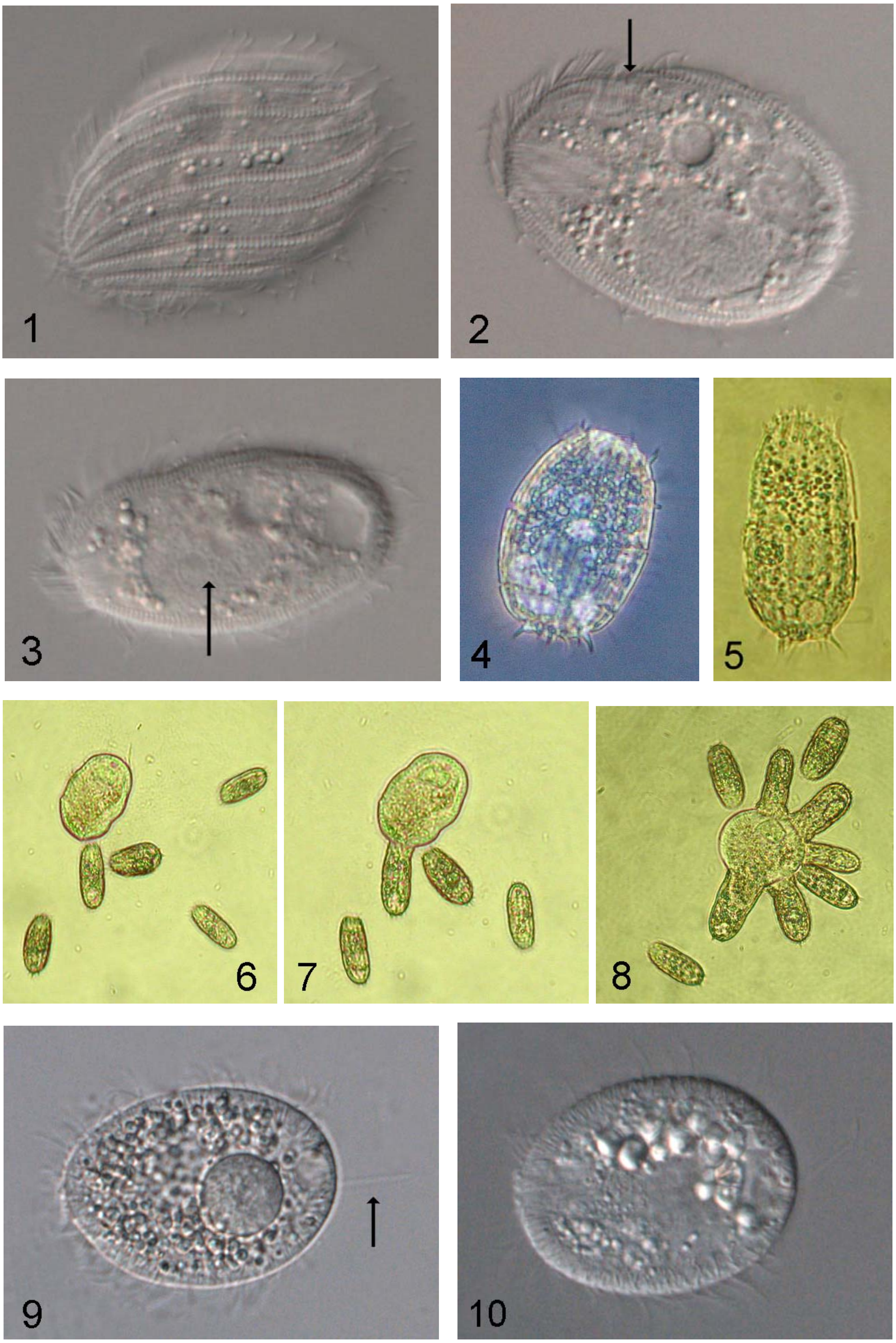


Plate 4.3.2

Ciliophora, Gymnostomatida. Anterior end of the cell with distinct oral cone, bulge or head. **1,** *Homalozoon vermiculare*, large ciliate, up to 570 µm long, with oral bulge containing toxicysts anteriorly (inset), and several contractile vacuoles along the cell; **2**, *Trachelophyllum apiculatum*, flask-shaped, with an elongated neck and cytostome on the snout-like tip (inset), contractile vacuole is terminal, body length 120 µm; **3,** *Lacrymaria olor,* with the long extensile neck, body length 380 µm; **4,** *Lacrymaria olor*, contracted, with protrusable snout which has two distinct zones (inset); **5**, *Lacrymaria coronata* group, flask-shaped ciliate, with the single terminal contractile vacuole, ovoid macronucleus and head region (inset) typical for the genus, body length 110 µm; **6**, *Mesodinium pulex,* with three pre-equatorial ciliary belts, the equatorial ciliary belt close to globular trunk, body length 18 µm; **7, 8, 9, 10, 11,** *Monodinium balbiani*, carnivorous ciliate, with one ciliary wreath encircling the oral cone, composed of nematodesmata (**8,** arrow) and sausage-shaped macronucleus, body length 60 µm (photos E. Mironova).

Photos **1-6, 9**: live, BF; **7, 8, 10, 11**: live, DIC.

Plate 4.3.2

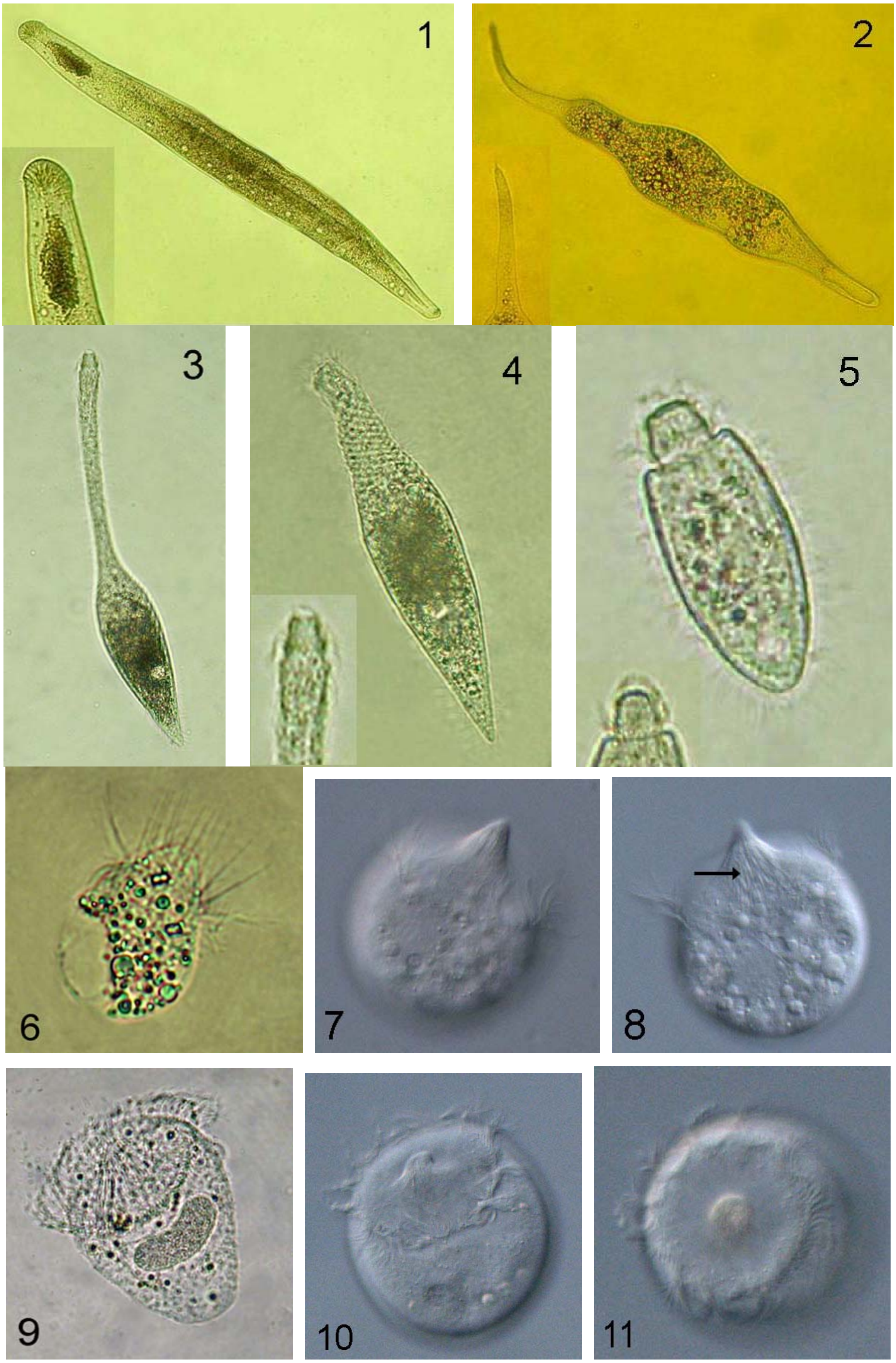


Plate 4.3.3

Ciliophora, Pleurostomatida. Cytostome slit-like along edge of laterally compressed body with circumoral ciliature showing left/right differentiation. **1,** *Litonotus cygnus,* with long and thin neck region, body length 230 µm; **2,** *Litonotus cygnus*, posterior end of the cell, two macronuclear nodules (arrows) and single contractile vacuole; **3, 4,** *Litonotus alpestris*, small ciliate (body length 25 µm) with slit-like oral aperture, equipped with distinct brush of cilia, single contractile vacuole in the posterior end; **5,** *Litonotus alpestris,* distinct single macronucleus (arrow); **6, 7,** *Litonotus varsaviensis,* with two contractile vacuoles, body length 37 µm; **8, 9,** *Litonotus lamella,* with single, thorn-shaped rib (**8,** arrow), body length 90 µm; **10,** *Litonotus lamella,* anterior end with many conspicuous extrusomes and two macronuclear nodules (arrow) (photos E. Mironova).

Photos **1, 2**: live, BF; **3-10**: live, DIC.

Plate 4.3.3

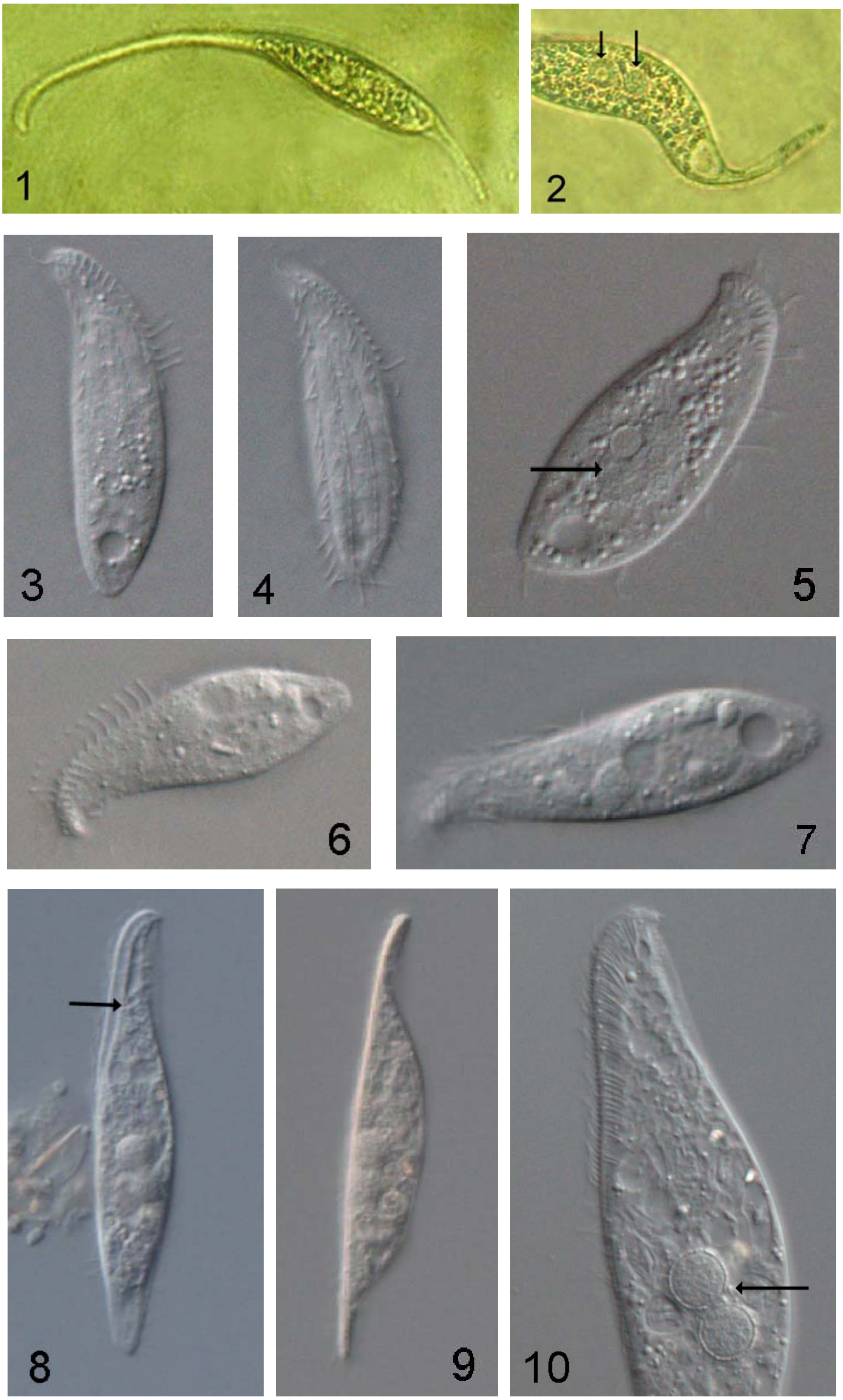


Plate 4.3.4

Ciliophora. 1, *Loxophyllum meleagris,* flat ciliate, with distinct extrusome warts at ventral side and moniliform macronucleus (arrow), body length 350 µm; **2, 3,** *Amphileptus pleurosigma,* with two macronuclear nodules (**2,** arrows) and numerous contractile vacuoles with long sigmoid canal (**3,** arrow), body length 180 µm; **4,** *Loxodes rostrum,* with rounded posterior end of the body and pronounced anterior «hood» over the buccal region (arrow), body length 190 µm; **5, 6,** *Cyrtolophosis mucicola*, small ciliate (body length 18 µm) with distinct apical vestibulum bearing long cilia (**6,** arrow); **7,** *Cyrtolophosis mucicola*, single macronucleus (arrow) and subterminal contractile vacuole are seen; **8, 9, 10,** *Microthorax* sp., small flattened ciliate, with the mouth near posterior end (**8,** arrow) and three longitudinal ciliary rows, body length 20 µm; **11, 12,** *Chilodontopsis depressa,* cyrtophorida with prominent large contractile vacuole posteriorly, ovoid macronucleus with single micronucleus and synhymenium (**12,** arrow), body length 55 µm (photos E. Mironova).

Photos **1-4**: live, BF; **5-12**: live, DIC.

Plate 4.3.4

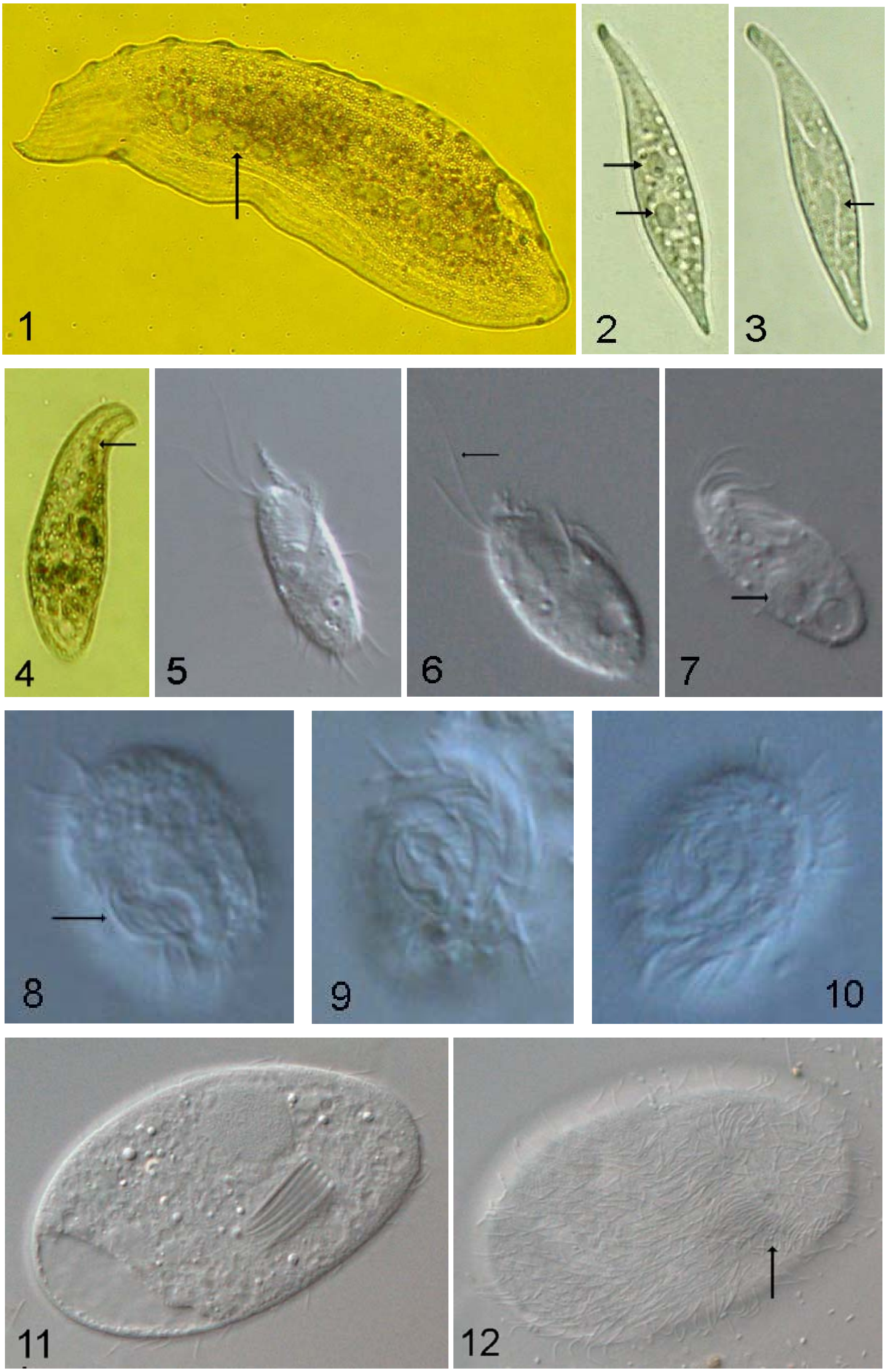


Plate 4.3.5

Ciliophora, Cyrtophorida. 1, 2, 3, *Chilodonella bavariensis*, the central zone is devoid of cilia, typically for this genus (**1,** arrow), five contractile vacuoles are present (**2,** arrows) along with macronucleus, body length 45 µm; **4, 5, 6,** *Chilodonella calkinsi,* with small rounded left anterior rostrum and four contractile vacuoles (**5,** arrows), body length 35 µm; **7, 8,** *Trochilia minuta*, small cyrtophorida (body length 23 µm), left ciliary field is characteristically reduced, large secretory podite is present in the posterior; **9,** *Trochilia minuta,* with distinct heteromeric macronucleus (arrow); **10,** *Trithigmostoma srameki,* with sixteen ciliary rows, numerous contractile vacuoles and large ovoid macronucleus, body length 60 µm (photos E. Mironova).

Photos **1-9**: live, DIC; **10**: live, BF.

Plate 4.3.5

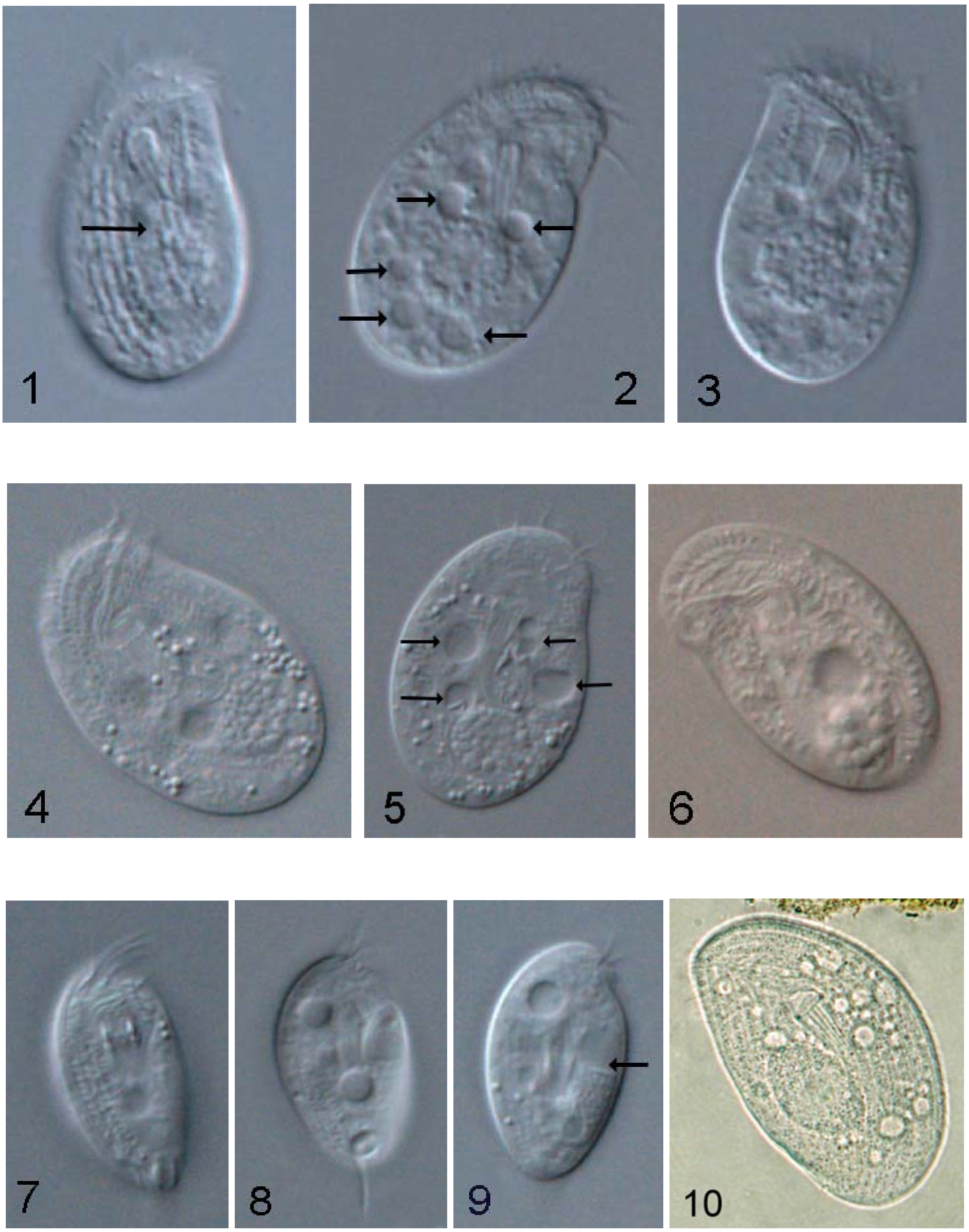


Plate 4.3.6

Ciliophora, Hymenostomata. 1, 2, 3, 4, *Colpidium kleini,* with small preoral top, large ellipsoid macronucleus (**2**, arrow) and distinct preoral seam (**3**, arrow), body length 105 µm; **5, 6,** *Colpidium* sp., with single ovoid macronucleus (**5,** arrow) and contractile vacuole near the posterior end, number of ciliary rows is fewer than in other species of this genus, body length 50 µm; **7**, **8, 9,** *Dexiostoma campylum*, preoral top is narrowly rounded, contractile vacuole in posterior end of the body (**7,** arrow), ovoid macronucleus is seen (**8,** arrow), body length 45 µm (photos E. Mironova).

Photos **1-9**: live, DIC.

Plate 4.3.6

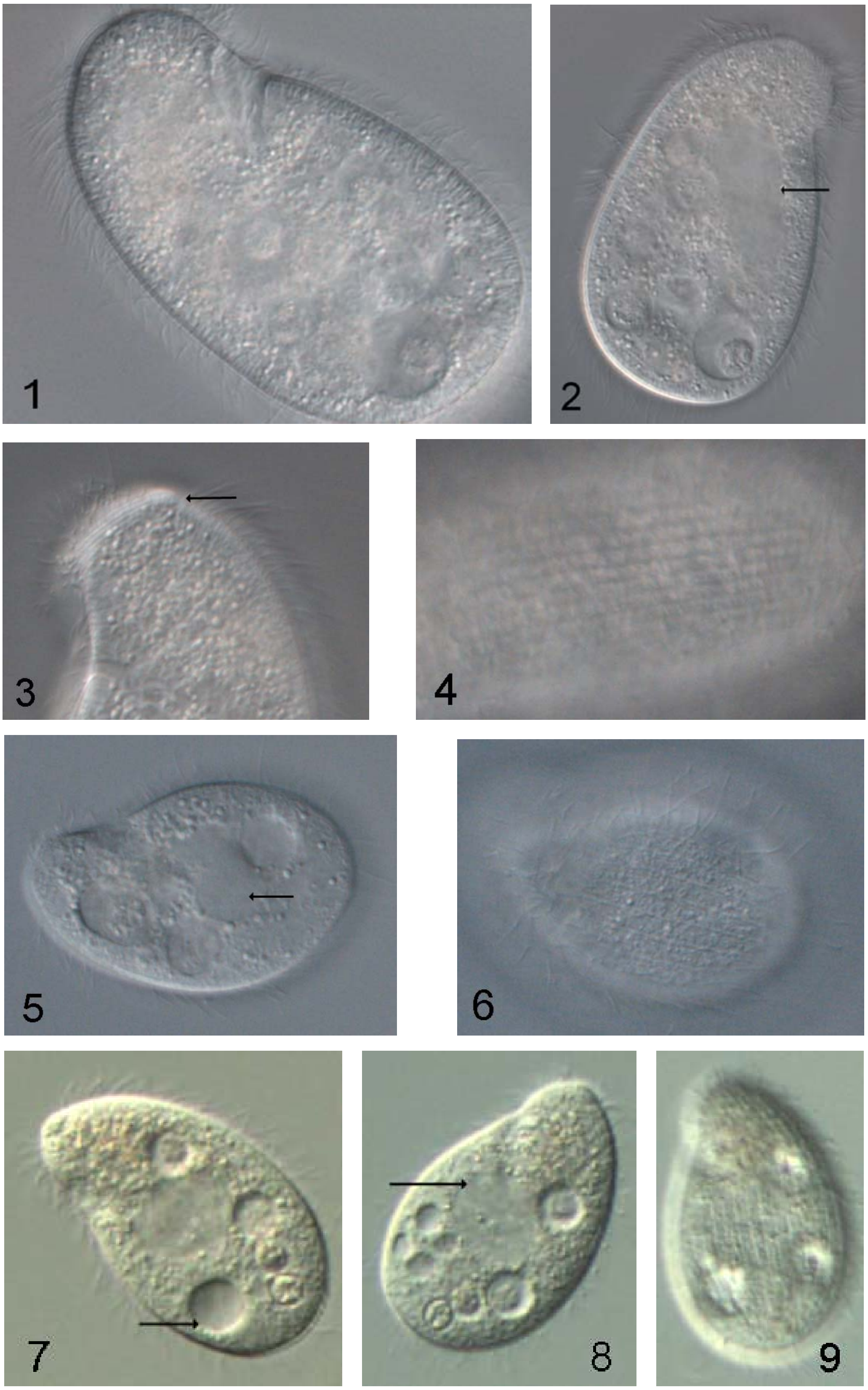


Plate 4.3.7

Ciliophora, Hymenostomata with distinct extrusome layer underneath pellicle. **1,** *Frontonia elliptica,* with triangular mouth in anterior third of the cell (arrow), body length 100 µm; **2,** *Frontonia elliptica,* two contractile vacuoles are seen (arrows); **3, 4,** *Paramecium putrinum*, oviform ciliate with conspicuous oral groove and oral aperture near mid-body, two contractile vacuoles, body length 80 µm; **5,** *Paramecium putrinum*, contractile vacuole with satellite vacuoles (arrow); **6, 7,** *Paramecium putrinum,* silver nitrate impregnation, silverline pattern and ovoid macronucleus with single micronucleus are readily observable (**7,** arrow); **8, 9, 12,** *Paramecium aurelia*-complex, fusiform ciliate with ovoid macronucleus and two micronuclei (**9,** arrow), body length 115 µm; **10, 11** *Paramecium aurelia*-complex, contractile vacuoles with radial channels, different stages of contraction (photos E. Mironova).

Photos **1, 2**: live, BF; **3-5, 9-11**: live, DIC; **8, 12**: live, PHC; **6, 7**: total preparation (wet silver nitrate impregnation).

Plate 4.3.7

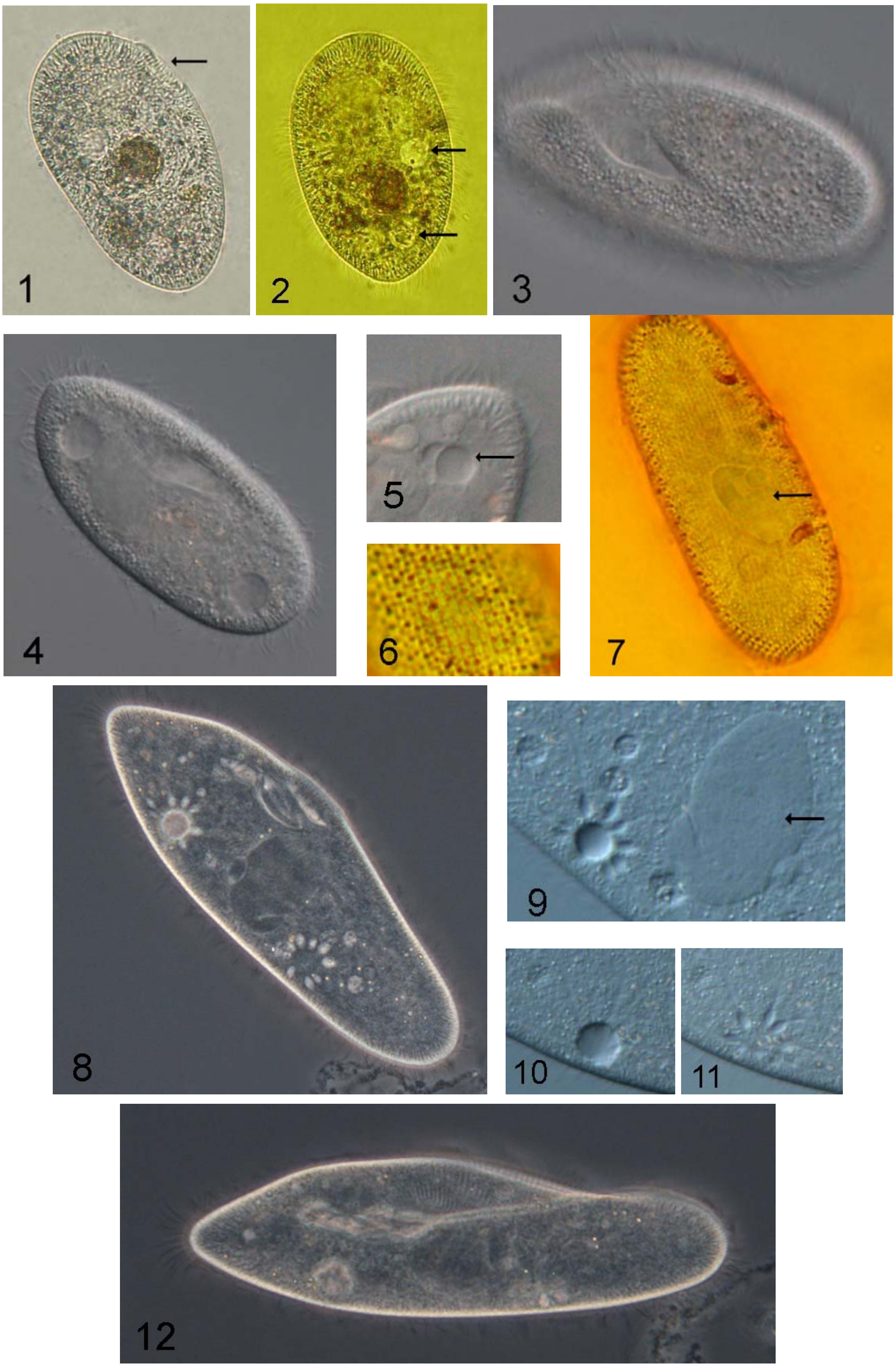


Plate 4.3.8

Ciliophora, Hymenostomata. 1, 2, *Lembadion lucens,* with broad deep furrow on ventral side, caudal cilia seen posteriorly (**2,** arrows), body length 55 µm; **3, 4, 5,** *Glaucoma scintillans*, buccal cavity with large undulating membrane, macronucleus is ovoid (**5,** arrow), contractile vacuole located posteriorly, body length 75 µm; **6,** *Glaucoma scintillans,* oral apparatus; **7, 8,** *Stokesia vernali*s, cordiform in ventral view, with chinked mouth near mid-body (**8,** arrow), body length 90 µm; **9, 10, 11,** *Urocentrum turbo*, dumbbell-shaped ciliate, with a tuft of caudal cilia and a single contractile vacuole located posteriorly (**10,** arrow), body length 55 µm (photos E. Mironova).

Photos **1, 2, 5, 7-11**: live, BF; **3, 4, 6**: live, DIC.

Plate 4.3.8

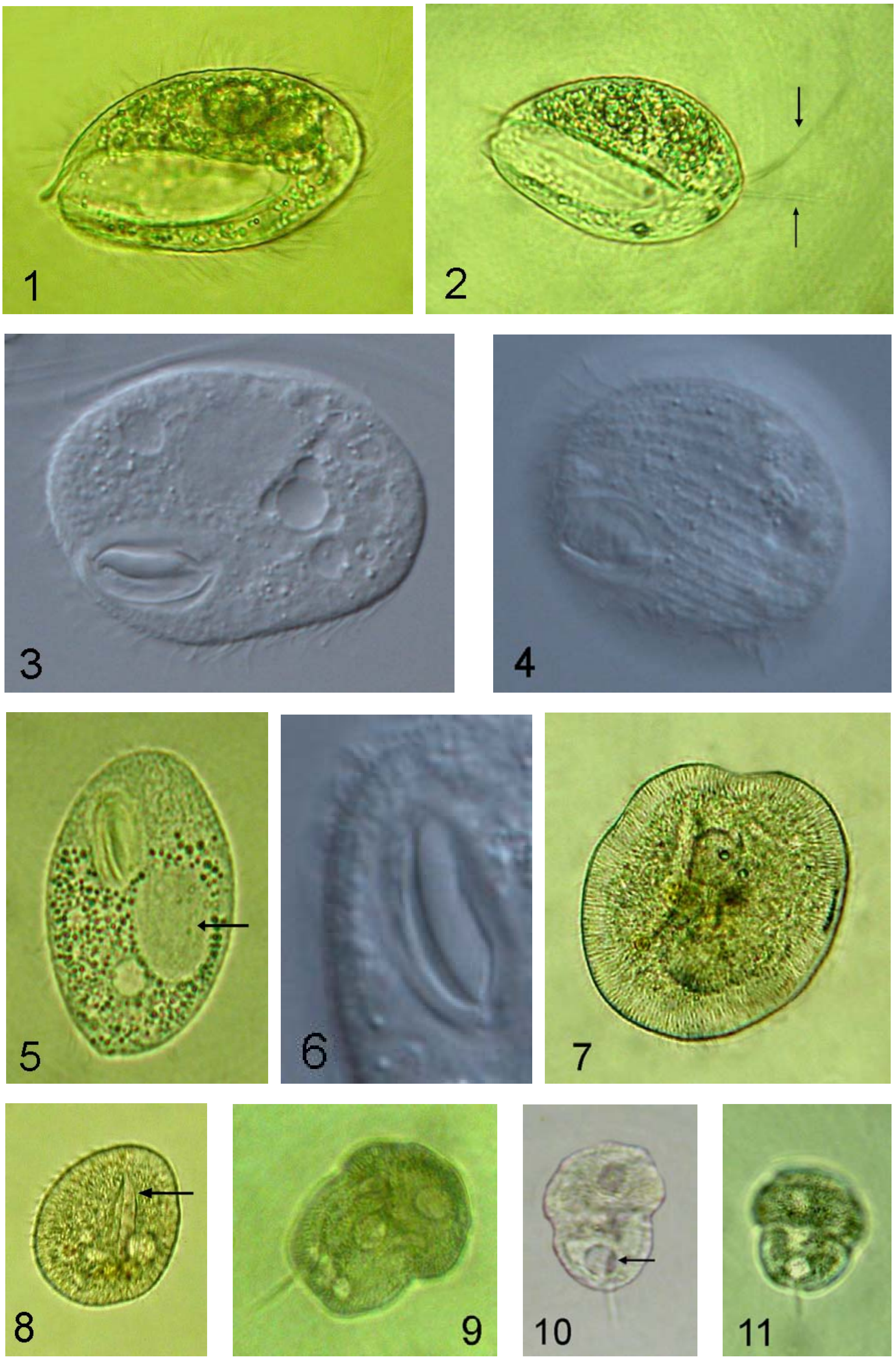


Plate 4.3.9

Ciliophora, Hymenostomata. 1, 2, 3, *Cinetochilum margaritaceum*, small scuticociliate (body length 20 μm) with few long caudal cilia at posterior end, subequatorial oral apparatus (**1,** arrow) and a contractile vacuole opposed to it (**2,** arrow); **4,** *Ctedoctema acanthocryptum,* slender-ellipsoid scuticociliate with distinct extrusomes in pellicle, contractile vacuole located posteriorly, blister in posterior end (arrow) is usual for dying ciliates; **5, 6,** *Ctedoctema acanthocryptum,* large undulating membrane is conspicuous (**5,** arrow), body length 30 μm; **7, 8,** *Ctedoctema acanthocryptum*, cell division; **9, 10,** *Uronema marinum*, scuticociliate, with small indentation in anterior third of the cell, single caudal cilium is present (**10,** arrow), contractile vacuole located posteriorly; **11,** *Uronema marinum*, undulating membrane is small, inconspicuous (arrow), body length 20 μm (photos E. Mironova).

Photos **1-4**, **6-11**: live, DIC; **5**: live, PHC.

Plate 4.3.9

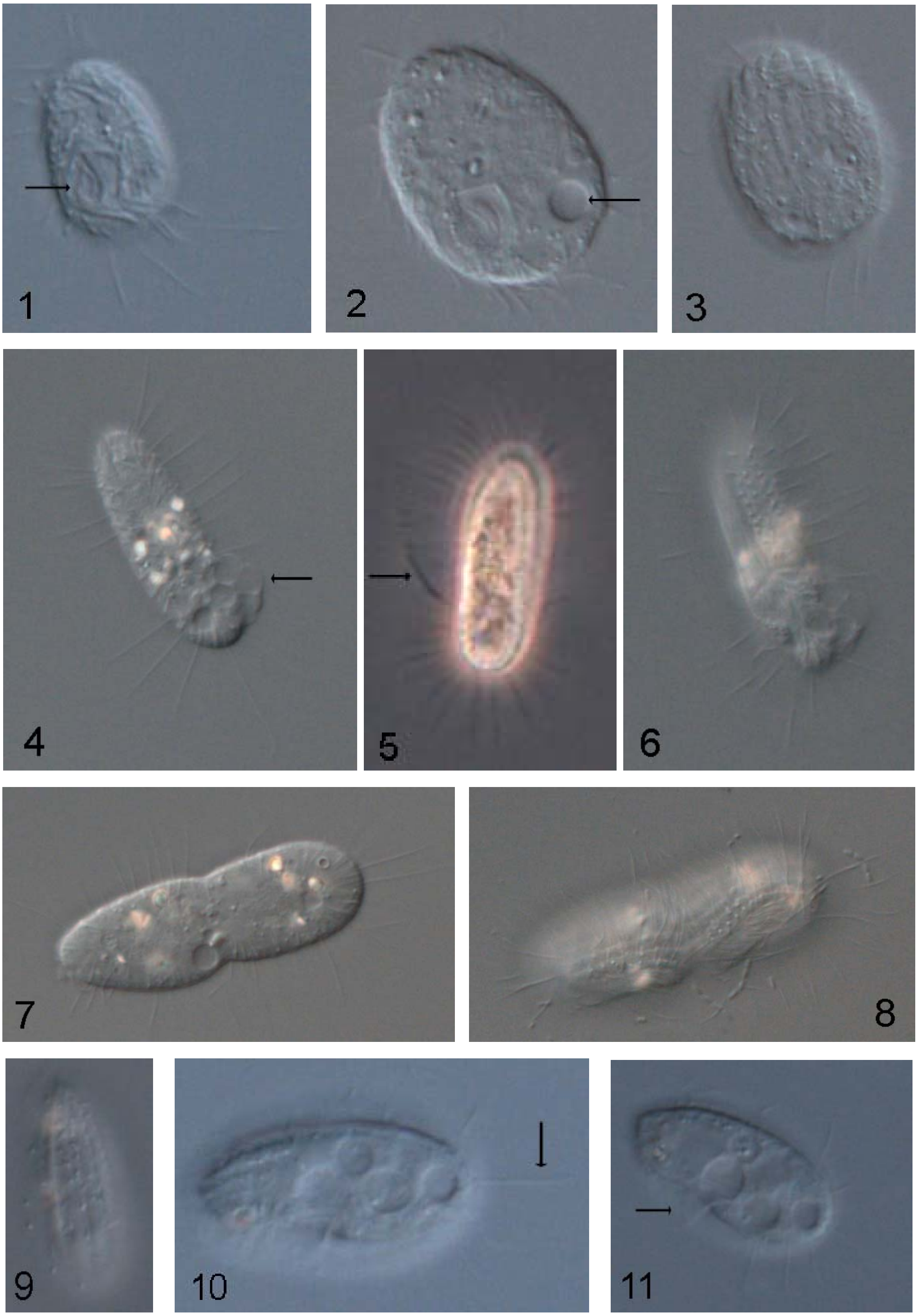


Plate 4.3.10

Ciliophora, Hymenostomata. 1, *Cristigera setosa,* scuticociliate with very large undulating membrane (arrow), ciliation is typically sparse in mid-body, posteriorly it is reduced to a few long rigid cilia, body length 25 µm; **2, 3,** *Cyclidium citrullus*, scuticociliate with small bumps at either end of the body, the posterior end is slightly notched (**3,** arrow), body length 18 µm; **4, 5,** *Cyclidium citrullus,* with single caudal cilium, contractile vacuole is located posteriorly, buccal area extends to two-thirds of the body length (**5,** arrow); **6, 7, 8, 9,** *Cyclidium glaucoma,* barrel-shaped scuticociliate with single caudal cilium, contractile vacuole in posterior end (**6,** arrow) and buccal apparatus which extends only half-way down the body (**8,** arrow), body length 15 µm; **10, 11,** *Cyclidium candens*, elongated scuticociliate with domed anterior (**10,** arrow), longitudinal ridges in the pellicle and a single long caudal cilium, body length 20 µm; **12,** *Cyclidium candens*, sail-like undulating membrane is conspicuous (arrow) (photos E. Mironova).

Photos **1-5**: live, PHC; **6-12**: live, DIC.

Plate 4.3.10

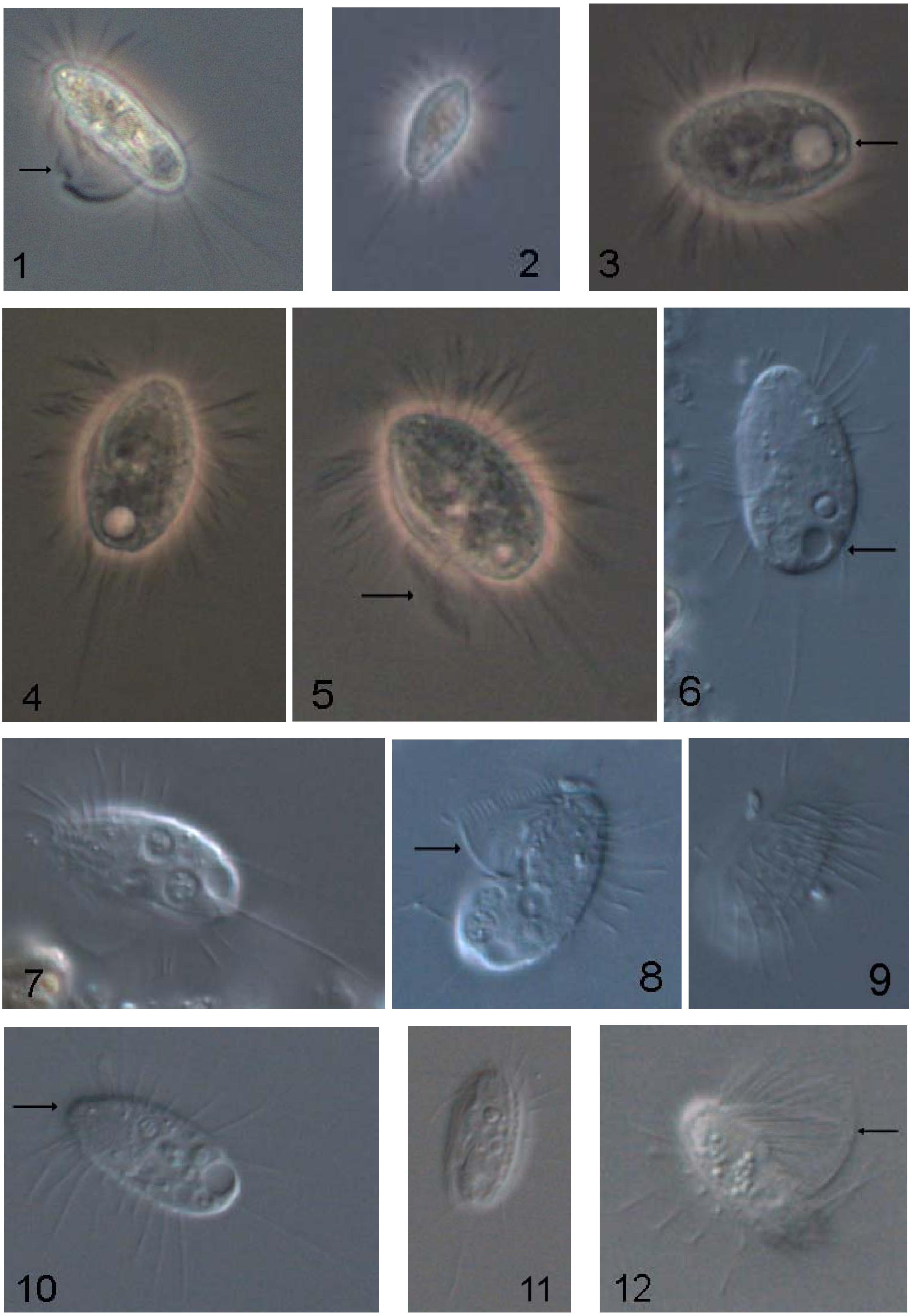


Plate 4.3.11

Ciliophora, Heterotrichida. 1, 4, *Stentor coeruleus*, blueish because of presence of numerous blue cortical granules (**4,** arrow), body length ca. 1 mm; **2**, *Stentor coeruleus*, moniliform macronucleus well seen (arrow); **3**, *Stentor coeruleus,* anterior of the body, with typical for the genus powerful zone of adoral membranelles; **5**, *Stentor coeruleus*, contracted; **6**, *Stentor coeruleus,* with captured rotifer *Keratella tecta* (arrow) (photos E. Mironova).

Photos **1-6**: live, BF.

Plate 4.3.11

Plate 4.3.12

Ciliophora, Heterotrichida. 1, 2, *Sphaerophrya stentori*, a parasitic suctorian ciliate (arrows) leaving it's host *Stentor* sp.; **3**, *Sphaerophrya stentori,* ciliated swarmer with four tentacles and conspicuous ovoid macronucleus, body length 15 µm; **4**, *Stentor roeselii*, in mucous lorica, with distinct vermiform macronucleus (arrow), body length 800 µm; **5**, *Stentor roeselii*, contracted; **6**, *Stentor roeselii*, without mucous lorica, feeding on cyanobacteria colony; **7**, **8**, *Stentor niger*, brownish, with single ellipsoid macronucleus (**8**, arrow), body length 200 µm; **9,** *Stentor muelleri*, colourless, with moniliform macronucleus (arrow), body length 600 µm (photos E. Mironova).

Photos **1, 2, 4-9**: live, BF; **3**: live, PHC.

Plate 4.3.12

Plate 4.3.13

Ciliophora, Oligotrichida. 1, 2, *Halteria grandinella*, somatic ciliature reduced to equatorial bundles of long bristles, body length 30 μm; **3,** *Halteria grandinella*, top view, split jumping bristles are conspicuous (arrow); **4,** *Strobilidium humile*, small ciliate (body length 20 μm) with horseshoe-shaped macronucleus (arrow); **5,** *Strobilidium humile*, top view, circle adoral zone of membranelles; **6, 9,** *Strobilidium caudatum,* pyriform ciliate, narrowed posteriorly, with horseshoe-shaped macronucleus (**6,** arrow) and contractile vacuole near the posterior end, body length 35 μm; **7, 8,** *Strobilidium caudatum,* with distinct anlagen of new oral apparatus (arrows); **10, 11, 12,** *Strobilidium caudatum,* top view of adoral zone, with prominent external and internal (**12,** arrow) adoral membranelles; **13,** *Strobilidium caudatum,* caudal view, spiral ciliary rows are conspicuous (arrow); **14, 15,** *Lohmaniella elegans*, globular ciliate with ovoid macronucleus (**14,** arrow), body length 15 μm; **16,** *Tintinnidium semiciliatum,* cylindrical irregular in form lorica with agglutinated particles, body length 23 μm (photos E. Mironova).

Photos **1, 2, 4, 5, 16**: live, BF; **3**: live, PHC; **6-15**: live, DIC.

Plate 4.3.13

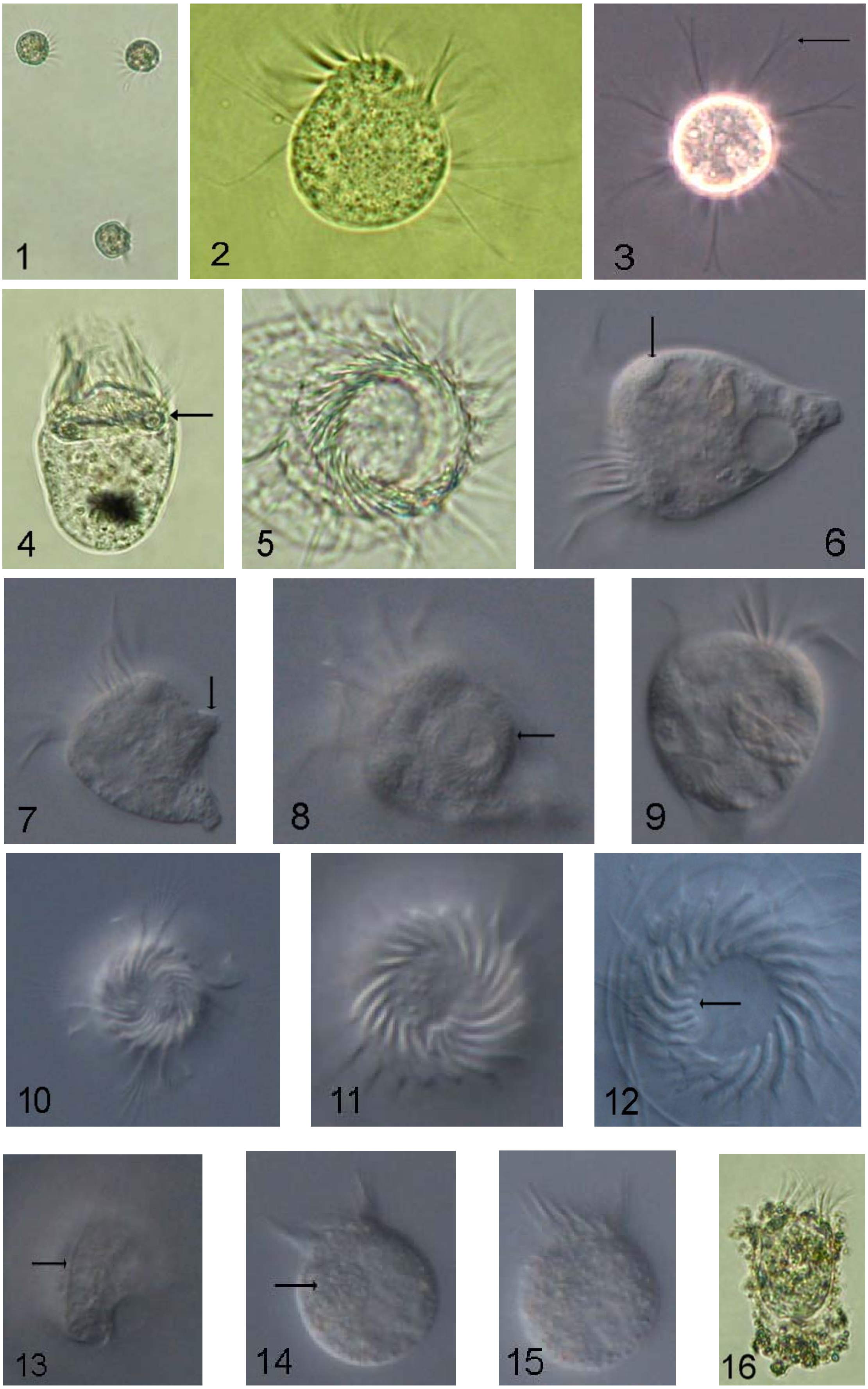


Plate 4.3.14

Ciliophora. Hypotrichia. 1, *Holosticha pullaster*, ventral side, two midventral cirral rows are conspicuous (arrow), typically for the genus, body length 78 µm; **2**, *Holosticha pullaster,* with two distinct macronuclear nodules (arrows) and subequatorial contractile vacuole; **3,** *Holosticha pullaster*, anterior part, with adoral zone of membranelles; **4, 5,** *Holosticha brevis,* ovoid hypotrichida with dumbbell-shaped macronucleus (**4,** arrow), body length 60 µm; **6,** *Anteholosticha monilata,* with moniliform macronucleus (arrow), body length 110 µm; **7,** *Anteholosticha monilata,* cyst; **8,** *Anteholosticha monilata*, anterior part of the body with small adoral zone of membranelles and contractile vacuole near it (arrow) (photos E. Mironova).

Photos **1-5**: live, DIC; **6-8**: live, BF.

Plate 4.3.14

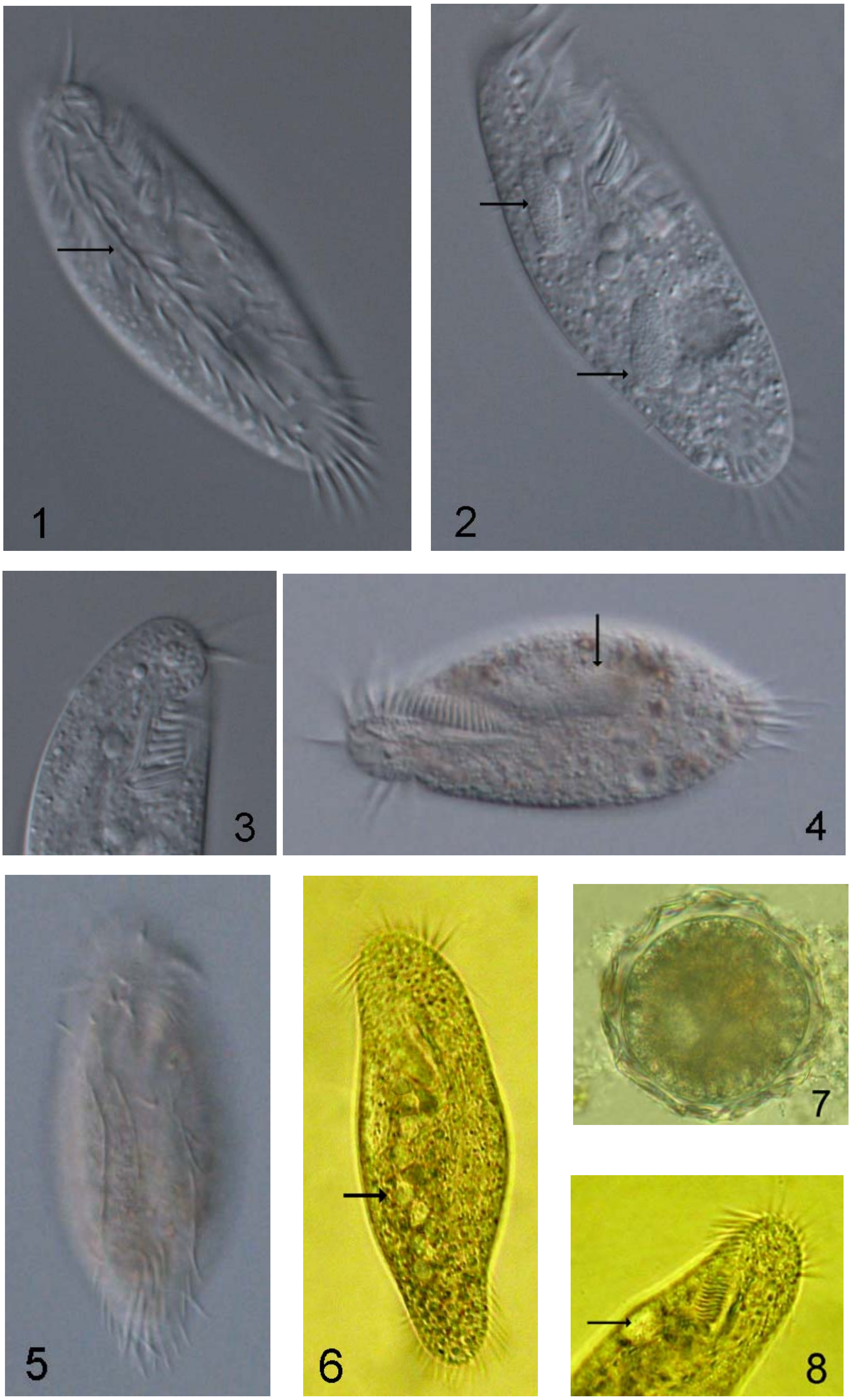


Plate 4.3.15

Ciliophora, Hypotrichia. 1, *Stichotricha secunda*, in lorica, many zoochlorellae are present in the cell, body length 110 µm; **2**, *Stichotricha secunda*, out of lorica, long straight adoral zone of membranelles is conspicuous (arrow); **3, 4,** *Amphisiella oblonga,* with spatulate posterior large adoral zone of membranelles and two ovoid macronuclei (**4,** arrows), body length 90 µm; **5,** *Urostyla grandis,* large ciliate (body length 280 µm), brownish due to presence of numerous cortical granules; **6,** *Urostyla grandis*, posterior end, with distinct cortical granules and many macronuclear nodules; **7, 8,** *Urostyla grandis,* with prominent adoral zone of membranelles and contractile vacuole (**7,** arrow) located in mid-body (photos E. Mironova).

Photos **1-4**: live, BF; **5-8**: live, DIC.

Plate 4.3.15

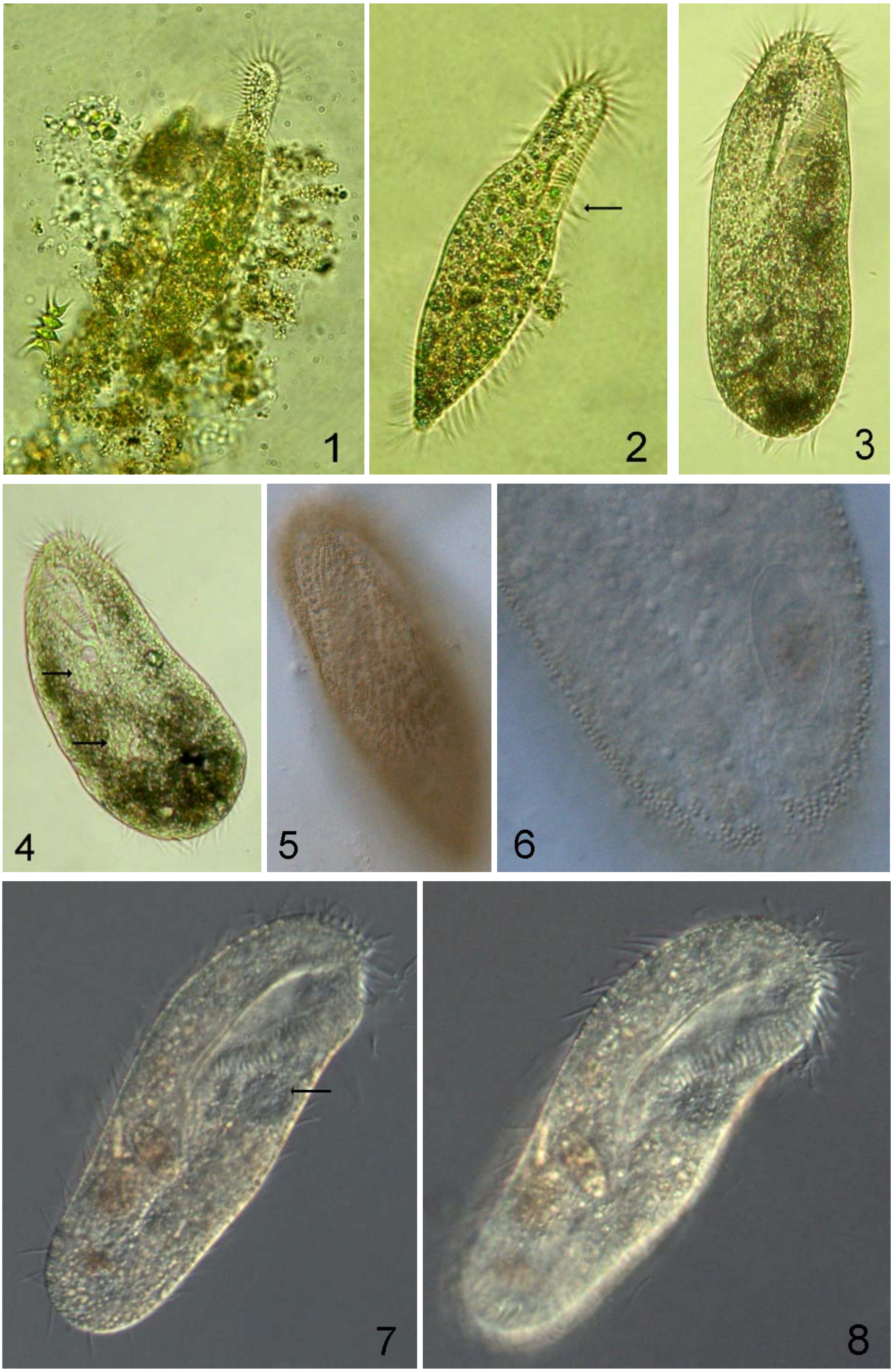


Plate 4.3.16

Ciliophora, Hypotrichia. 1, *Euplotes affinis,* with large adoral zone of membranelles, body length 45 µm; **2, 3,** *Euplotes affinis,* nine frontoventral cirri (**2,** arrows), three caudal (**3,** arrows) and five transverse cirri are present; **4,** *Euplotes affinis,* with 3-shaped macronucleus (arrow); **5,** *Euplotes affinis,* dorsal surface, with conspicuous longitudinal ribs; **6,** *Euplotes affinis,* ventral surface, with three prominent ridges; **7, 8,** *Aspidisca lynceus,* with smooth outline of body, seven frontoventral and five transverse cirri, contractile vacuole is located posteriorly (**7,** arrow), body length 35 µm; **9,** *Aspidisca lynceus,* with adoral zone of membranelles (arrow) and distinct frontal membranelles anteriorly (photos E. Mironova).

Photos **1-3, 5, 6, 9**: live, DIC; **4, 7, 8**: live, BF.

Plate 4.3.16

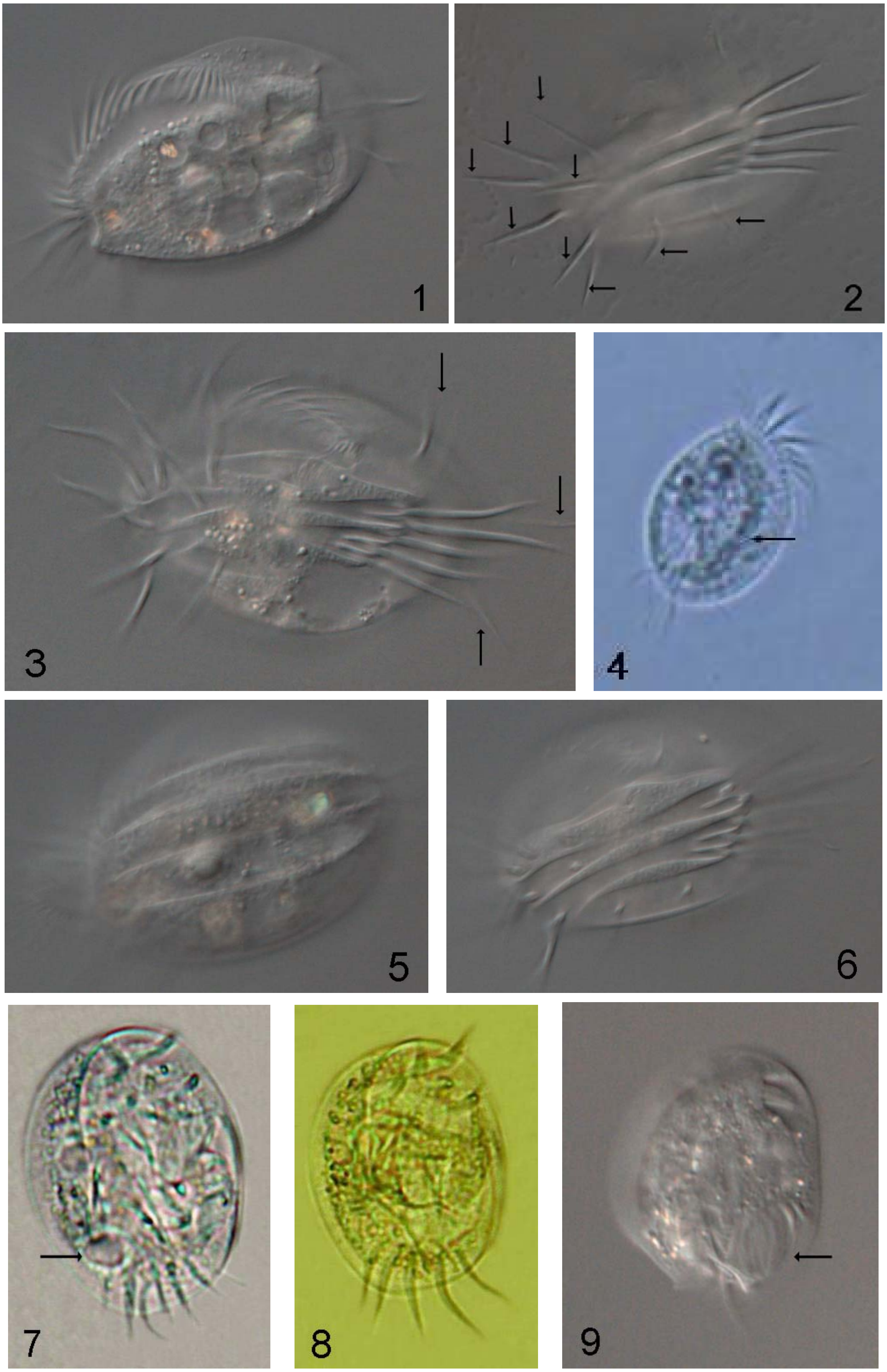


Plate 4.3.17

Ciliophora, Hypotrichia. 1, 2, *Aspidisca turrita,* with seven frontoventral and five transverse cirri, adoral zone of membranelles is located near the posterior end (**2,** arrow), body length 25 µm; **3, 4,** *Aspidisca turrita,* dorsal surface carries characteristic backwardly curving thorn; **5,** *Stylonychia* sp., body shape is atypical for the genus but three distinct caudal cirri (arrows) are seen, body length 47 µm; **6,** *Stylonychia* sp., cyst; **7,** *Stylonychia mytilus*–complex, with three long and distinctly separate caudal cirri, anterior area is notably widened, body length 120 µm; **8,** *Stylonychia* sp., posterior end is narowly rounded, with three indistinctly separate caudal cirri (arrow), body length 95 µm; **9,** *Stylonychia* sp., division, two macronuclear nodules are conspicuous (photos E. Mironova).

Photos **1-3**: live, DIC; **4**: live, PHC; **5-9**: live, BF.

Plate 4.3.17

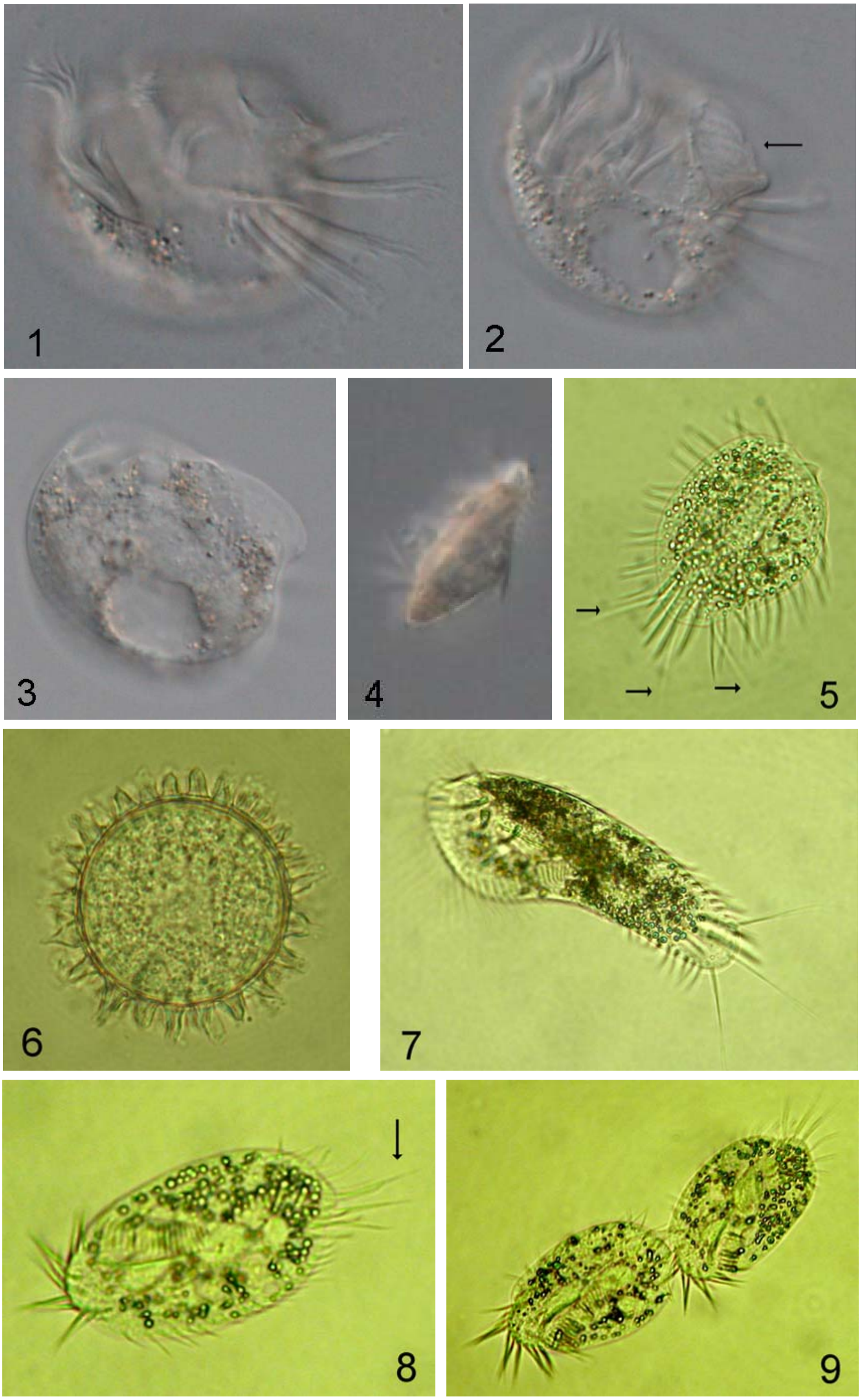


Plate 4.3.18

Ciliophora, Hypotrichia. 1, *Histriculus vorax,* with large adoral zone of membranelles and two macronuclear nodules, body length 210 µm; **2,** *Histriculus vorax*, posterior body end is broadly rounded and notched (arrow); **3,** *Sterkiella histriomuscorum,* with short inconspicuous caudal cirri (arrow) and contractile vacuole located in the mid-body, body length 105 µm; **4, 5, 6,** *Oxytricha setigera,* with inconspicuous caudal cirri (**4**, arrows), dorsal cilia (**5**, arrow) and contractile vacuole located in mid-body between two macronuclear nodules (**6**, arrows), body length 38 µm; **7, 8,** *Tachysoma pellionellum,* with contractile vacuole located in mid-body (**7**, arrow) and stiff dorsal cilia (**8**, arrow), body length 65 µm; **9, 10,** Unidentified ciliate, body length 70 µm (photos E. Mironova).

Photos **4-6**: live, DIC; **1, 3, 7-10**: live, BF; **2**: live, PHC.

Plate 4.3.18

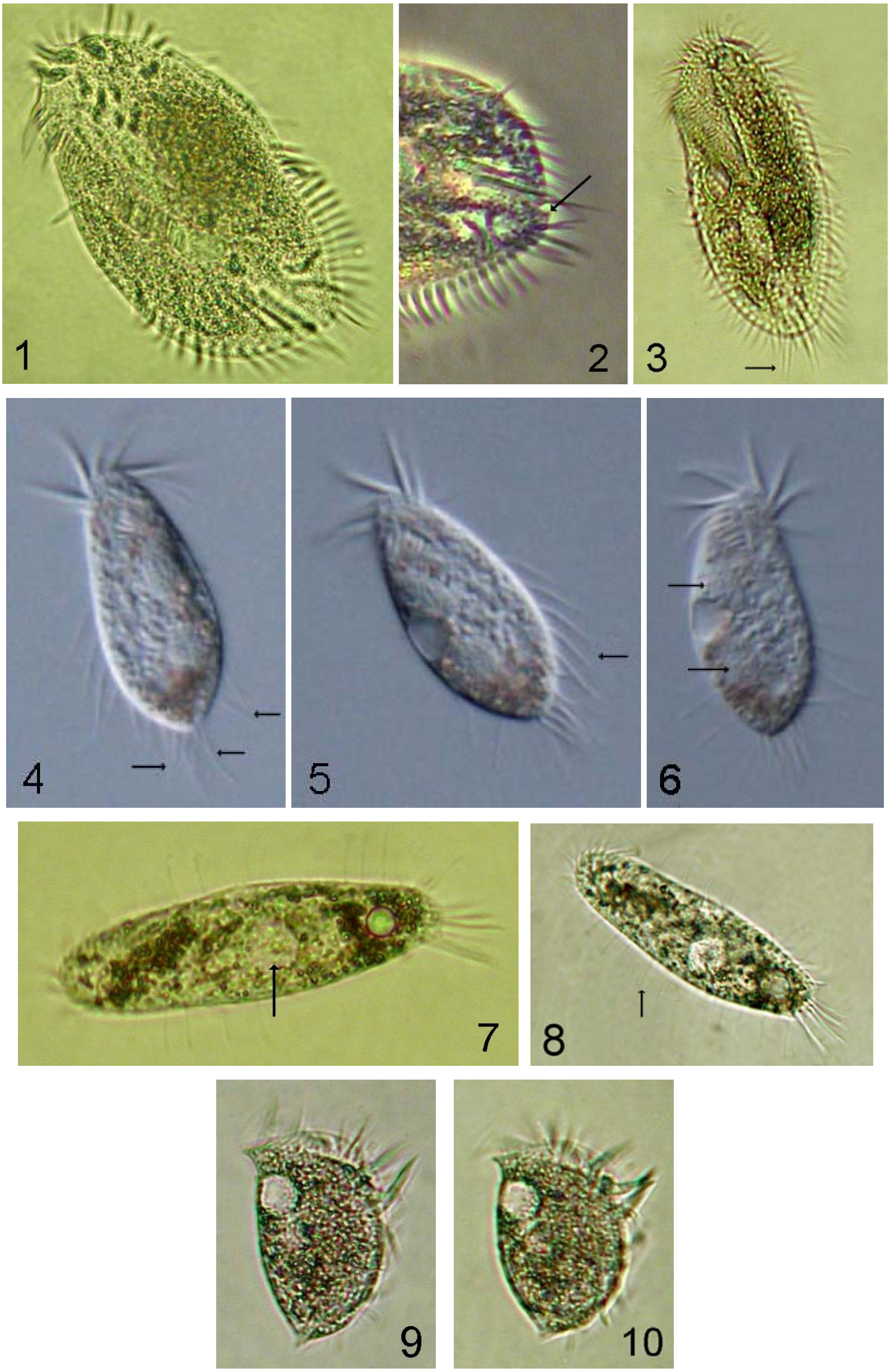


Plate 4.3.19

Ciliophora, Peritrichia. 1, *Carhesium polypinum,* part of colony, stalk muscle is distinctly interrupted (arrow); **2,** *Carhesium polypinum,* two zooids, with J-shaped macronucleus (arrow), body length 110 µm; **3,** *Carhesium polypinum,* swarmer; **4,** *Epistylis hentscheli,* part of colony, stalk is not contractile, typically for this genus; **5,** *Epistylis hentscheli,* zooid, with horseshoe-shaped macronucleus (arrow), body length 125 µm; **6,** *Epistylis plicatilis,* elongated zooid with "C-shaped" macronucleus (arrow), body length 155 µm; **7,** *Epistylis plicatilis,* colony (photos E. Mironova).

Photos **1-7**: live, BF.

Plate 4.3.19

Plate 4.3.20

Ciliophora, Peritrichia. 1, *Vorticella picta*, small sessilid ciliate with conspicuous refractive granules in the stalk, body length 55 µm; **2,** *Vorticella* sp., with large distinctive J-shaped macronucleus (arrow), body length 80 µm; **3,** *Vorticella* sp., colony, attached the filamentous cyanobacteria, body length 67 µm; **4,** *Vorticella convallaria*-complex, bell-shaped ciliate, body length 70 µm (photos E. Mironova).

Ciliophora, Suctoria. 5, 6, 7, *Metacineta mystacina*, small suctorian ciliate with stalked lorica pierced by slits (**6,** arrow), contractile vacuole located in the mid-body (**5**, arrow), and single spherical macronucleus (**7,** arrow), body length 45 µm (photos E. Mironova).

Photos **1-4**: live, BF; **5**: live, PHC; **6, 7**: live, DIC.

Plate 4.3.20

5. MESO- AND MACROZOOPLANKTON OF THE OPEN BALTIC SEA

5.1. Description of most abundant meso- and macrozooplankton groups

This chapter provides brief information on morphology, reproduction modes, development and ecology of the most common representatives from the major taxonomic groups of meso- and macrozooplankton in the Baltic Sea.

Cnidaria

(Plates 5.3.38, 5.3.39)

Cnidarians are diploblastic metazoans; i.e. they consist of two epithelial layers only – an ectodermal epidermis and an endodermal gastrodermis, separated by a primarily acellular extracellular matrix, called mesogloea. The most characteristic structures are the cnidae (nematocysts) produced by specific cells and generally used to catch prey that may be much larger than the individual itself.

Cnidarians not only use larvae as means of dispersal in the open waters; additionally they have achieved an obligate generation completely committed to propagation: a clear and concise description of this process is given by Larink and Westheide (2006). In an alternation of generations, called metagenesis, this sexually reproducing generation usually is a free-swimming **medusa**, which arises from a polypoid generation through budding. In some cases medusae may also arise asexually from other medusae, but characteristically medusae produce and broadcast either sperm or eggs. The fertilized eggs develop into ciliated free-swimming planulae, which later become attached to the bottom and metamorphose into the **polyp**. Polyp and medusa of one species may be very different in phenotype and their relationship is rarely apparent; thus in many cases they were described as different species.

The Cnidaria comprise **Anthozoa**, **Cubozoa** (exclusively tropical forms), **Scyphozoa**, and **Hydrozoa**, all different in structure, size and reproduction of their polypoid and medusoid forms. In the Hydrozoa, medusae are often secondarily suppressed, in which case the asexual buds of the polyp do not develop into free-swimming medusae but remain sessile. In the Anthozoa, no metagenesis occurs and the exclusively polypoid forms reproduce both sexually and asexually. Planktonic stages of all Anthozoa, Scyphozoa and Hydrozoa can be found in the sea (Fig. 5.1.1-5.1.3).

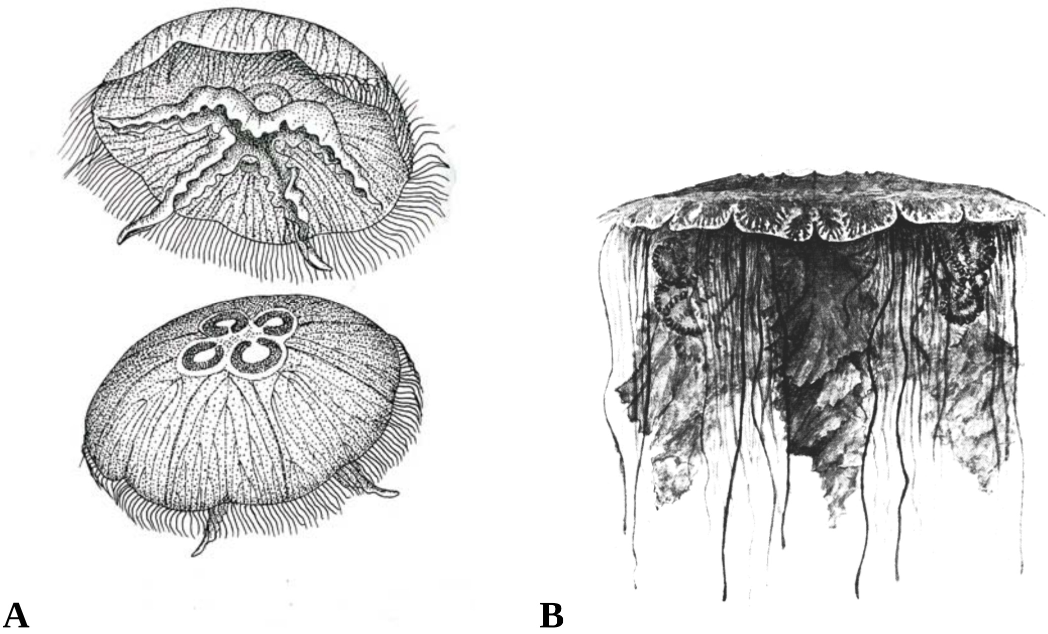


Figure 5.1.1. Cnidaria, Scyphomedusae: **A**, *Aurelia aurita* (modified from Hayward & Ryland, 2005); **B**, *Cyanea capillata* (after Russel, 1970).

The moon jelly, a scyphomedusae *Aurelia aurita* is the most dominant species of the Baltic Sea. Barz et al. (2006) characterise it as a species, which can reduce the stocks of mesozooplankton communities considerably in years of high abundance (e.g. Möller, 1980; Matsakis & Conover, 1991; Purcell, 1992; Olesen, 1995; Omori et al., 1995; Lucas et al., 1997; Schneider & Behrends, 1998). These medusae compete for zooplankton with commercially important planktivorous fish species and ctenophores, but they may also prey on fish eggs and larvae and thus directly affect their recruitment (Barz et al., 2006). The Belt Sea, the western Baltic Sea and also the Archipelago Sea are known as strobilation areas for *A. aurita*. However, ephyra are not regularly found in the Baltic Proper. Some authors concluded that *A. aurita* does not strobilate in this area (Janas & Witek, 1993; Barz & Hirche, 2005). However, the occurrence of the other larger Scyphomedusae, *Cyanea capillata* in the western Baltic Sea and in the Baltic Proper is always a sign for salt water influx from the Kattegat area. An indication for strong salt water influxes is the occurrence of the hydromedusae *Euphysa aurata* in the western Baltic Sea (Wasmund et al., 2004).

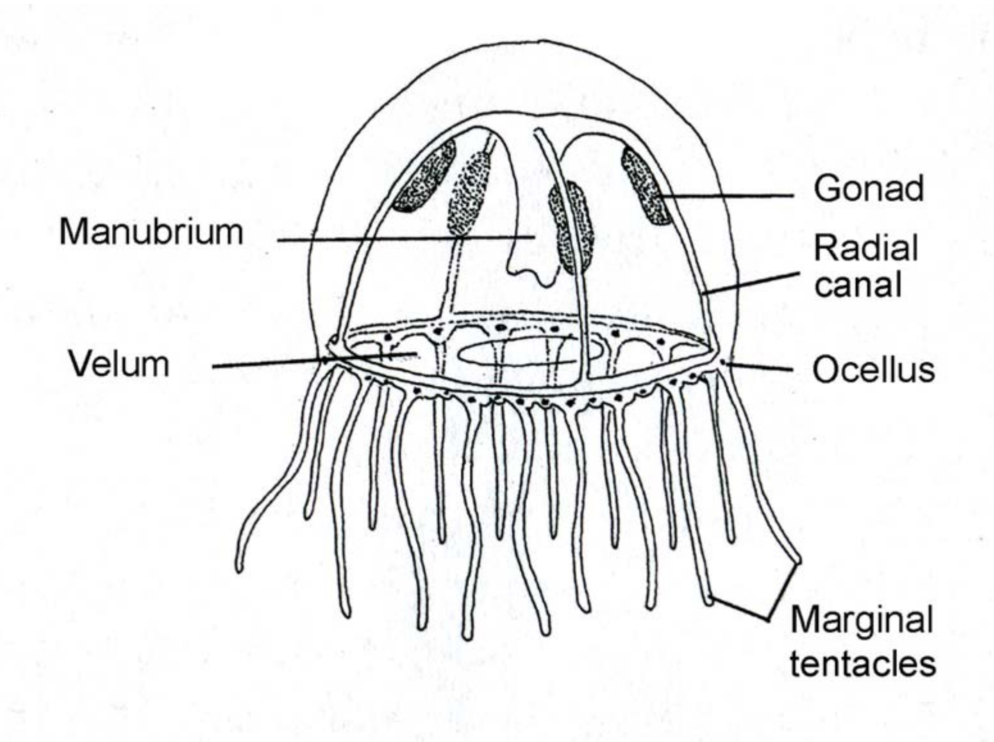


Figure 5.1.2. Typical hydroid medusae (modified from Hayward & Ryland, 2005).

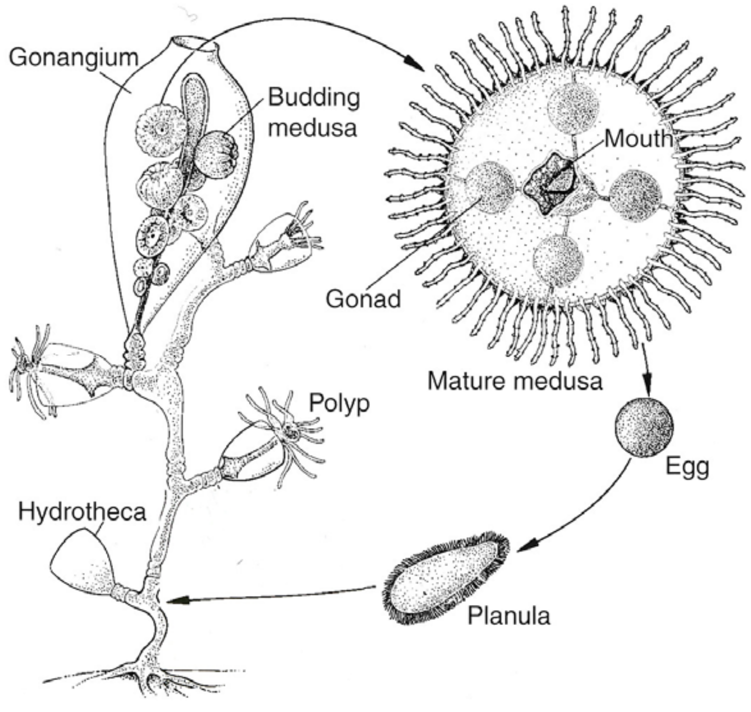


Figure 5.1.3. Life cycle of a Leptomedusae *Obelia geniculata* (modified from Larink & Westheide, 2006).

Ctenophora
(Plates 5.3.36, 5.3.37)

Ctenophora, or comb jellies, are presumably holoplanktonic organisms. Usually they are several centimetres long; they occur in all seas, and major part of species are considered to be cosmopolitan (Larink & Westheide, 2006).

The body of comb jellies has biradial symmetry: one central plane passes through both tentacle pouches and another plane is at a right angle to this, passing through the mouth slit (Fig. 5.1.4). Each plane divides the body into equal halves. Eight comb rows (ctenes) consisting of transverse plates of fused cilia are the locomotory organs by which the animals actively swim, the oral pole forward.

The **Tentaculifera** species have two long contractile tentacles, each emerging from the bottom of a deep epidermal pouch – the example is the ovoid species of the genus *Pleurobrachia*; their regular branches (tentilles) are covered with sticky colloblasts that on contact adhere to prey (e.g. planktonic copepods and other smaller organisms). **Lobata** (e.g. *Bolinopsis infundibulum*) are tentaculiferan comb jellies with two additional oral lobes flanking the mouth and the reduced tentacles. The lobes form a large cavity into which water with potential prey organisms are drawn by large cilia during the mouth-forward locomotion. The **Atentaculata** lack tentacles completely; the example is the cylindrical species of the genus *Beroe*. They feed on other ctenophores by swallowing them through their large slit-like mouth.

The ctenophore species are usually closely paired in predator-prey relationships that control their abundance. In the Northern and Baltic seas, once per year (between March and July) these comb jellies multiply massively, increasing in abundance by as much as four orders of magnitude within one to three months (Larink & Westheide, 2006). *Pleurobrachia pileus* feeds on herbivorous zooplankton, especially on the copepods that appear in spring: one individual *P. pileus* may eat as many as 300 copepods per day; then usually *Beroe gracilis* appears which feeds exclusively on *P. pileus* and practically eliminates it within three weeks. *Beroe cucumis* feeds chiefly on *B. infundibulum*.

The ctenophore species *Mnemiopsis leidyi* is one of the most recent invaders to the Baltic Sea. In summer 2006 the first observations of this West Atlantic comb jelly in Northern Europe were reported from the North Sea, the Skagerrak and the south-western Baltic Sea (Faasse & Bayha, 2006; Hansson, 2006; Javidpour et al., 2006). During autumn/winter 2006 and spring 2007 the further spread of this invasive ctenophore from the south-western towards the central Baltic Sea up to the south eastern Gotland Basin was reported (Kube et al., 2007a). The abundances were generally low (max. 4 ind. m^{-3}). While *M.*

leidyi was found in the entire water column in Kiel Bight, it occurred exceptionally below the halocline in the deep stratified central Baltic basins. Data of a weekly sampling program at a near shore location in Mecklenburg Bight between January and May 2007 showed that up to 80% of the individuals were juveniles, smaller than 1 mm total body length and that *M. leidyi* survived the winter in the Southern Baltic Sea, even when abundances dropped down to <1 ind.m^{-3} in February. During summer 2007, a regional gradient in population density of *M. leidyi* remained. The abundances west off Darss Sill exceeded those in the Baltic Proper by one to two orders of magnitude. The maximum abundances of 500 ind.m^{-3} in Kiel Bight corresponded to those in the area of origin of *M. leidyi* – off the North American coast, and to those in the Black Sea during the 1980-s. Generally, the adults were smaller in the Baltic Sea (6 cm) than in the Black Sea (18 cm) (Kube et al., 2007b). In 2007, *M. leidyi* spread up to the entrance of the Gulf of Finland and the central Bothnian Sea. It was recorded by the Finish Institute of Marine Research in August/September 2007 in quantities less than 10 ind.m^{-3}. The highest densities including juveniles were found in the water layers around halocline.

A first assessment of the physiological demands of this species versus the environmental conditions of the Baltic Sea showed that the successful establishment of this ctenophore is probable in the south-western and central Baltic Sea (Kube et al., 2007a). At present it is likely that *M. leidyi* has been successfully established in the Baltic Sea as the fifth ctenophore species.

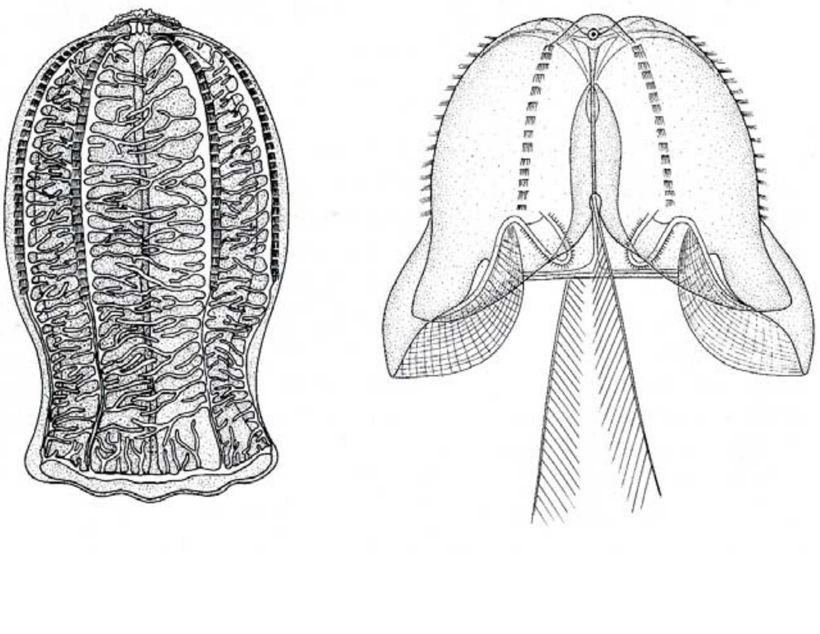


Figure 5.1.4. Ctenophores; general schematic view of *Beroe* sp. (left) and *Bolinopsis* sp. (right, modified from Westheide & Rieger, 1996).

Rotifera

(Plates 5.3.1 – 5.3.5)

The phylum Rotifera, or rotifers in English usage, is a group of microscopic aquatic or semi-aquatic invertebrates, that comprises around 2000 species of unsegmented, bilaterally symmetrical pseudocoelomates. The majority of rotifers inhabit fresh waters; however, some genera also occur in brackish and marine habitats. For example, about 20 of 32 species comprising the genus *Synchaeta* are described as marine (Nogrady, 1982). Only one order (Seisonidea, containing a single genus) and about 50 species of rotifers are exclusively marine; only two species are encountered in the plankton of the open Atlantic Ocean (Nogrady et al., 1993). Rotifers are not as diverse or abundant in marine environments as microcrustaceans but they are common in many brackish, coastal, near shore and interstitial marine communities (Egloff, 1988) where they occasionally comprise the dominant portion of the biomass (Schnese, 1973; Johansson, 1983).

In the brackish waters of the open Baltic Sea rotifers form a highly diverse and widely distributed group due to the significant influence of the waters from the extended coastal areas with the rich fauna of freshwater and euryhaline rotifers (Telesh & Heerkloss, 2002; Telesh, 2004).

Morphologically, rotifers possess two main distinctive features: corona and mastax. The ciliated region at the apical end (head) of a rotifer is called the **corona** ("wheel organ"); it is used for locomotion and food gathering (Fig. 5.1.5). In adults of some rotifer families, ciliation is reduced and the corona is replaced by a funnel or bowl-shaped structure (the infundibulum) at the bottom of which the mouth is located. Along the edge of the infundibulum of most species there is a series of long setae (bristles).

The other universal characteristic of rotifers is a muscular pharynx, the **mastax**, possessing a complex set of hard jaws called **trophi** (Fig. 5.1.6).

Most rotifers are free living, they swim in the pelagial or crawl on substrata (bottom sediments, stems of macrophytes); however, many species live permanently attached to plants (the latter are called sessile rotifers). Very few rotifers are parasitic; the vast majority of rotifers are solitary but some (ca. 25 species) form colonies of various sizes (Wallace, 1987).

Nearly all free-living rotifers are suspension-feeders that utilise microalgae smaller than 12 µm in diameter (sometimes as large as 18 µm), bacteria and detritus (Pourriot, 1977); some are obligate or occasional predators.

Most rotifers are either obligatory parthenogenetic (the whole class of bdelloids) or produce males for a brief period, sometimes only a few days, each year or season (Nogrady et al., 1993). Male rotifers are usually strongly reduced in size and sometimes only slightly resembling the females of the same species (Fig. 5.1.5).

Polymorphism is a common phenomenon to many rotifer species. Individuals of the same species collected from one locality over a period of time often show changes in one or more characteristics from one generation to another (e.g. length of spines, or proportions of the body). In some localities these variations are season-specific: a phenomenon known as cyclomorphosis which is most common in some loricate rotifers (e.g. *Keratella* and *Brachionus*), but also can be observed in the illoricates (e.g. *Asplanchna*).

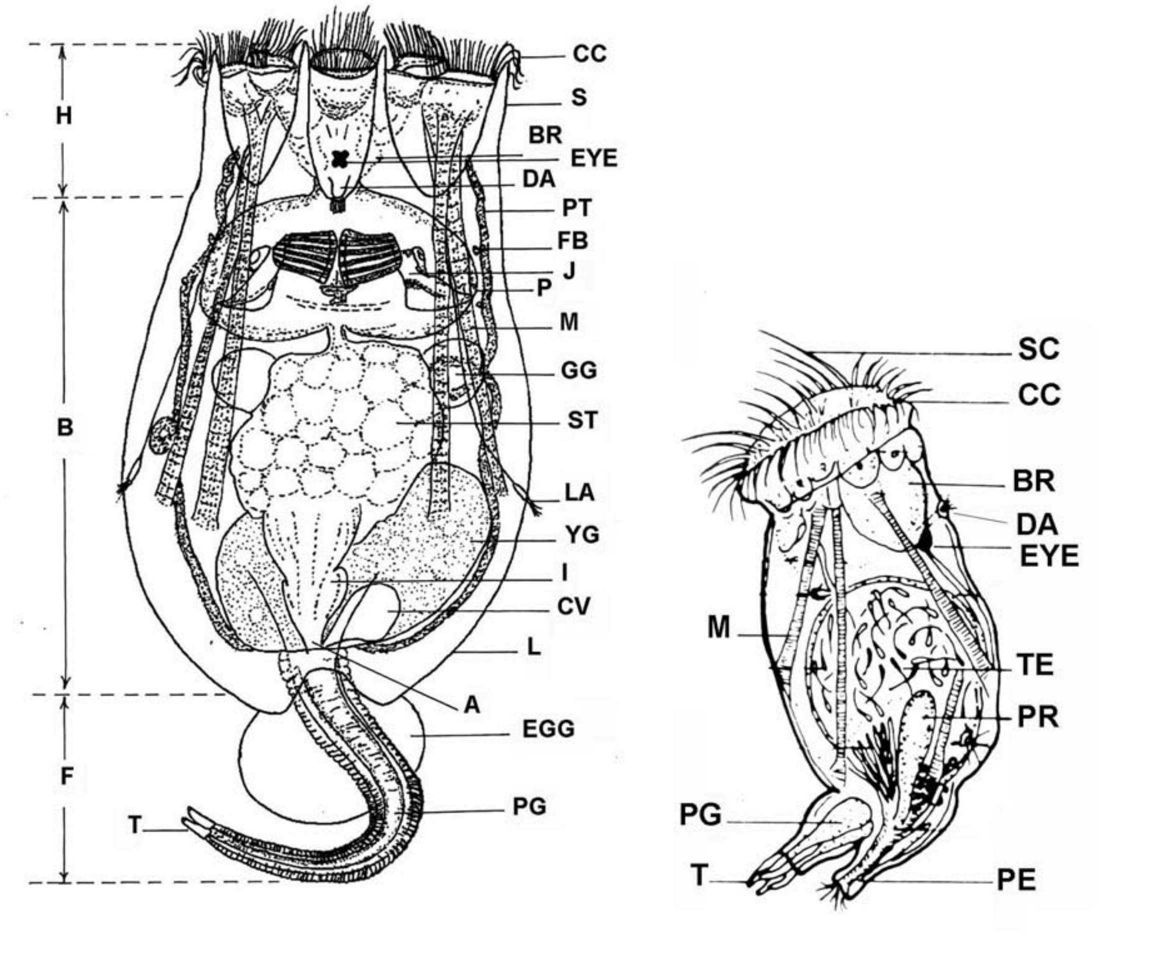


Figure 5.1.5. Morphology of a rotifer *Brachionus calyciflorus*: female on the left (dorsal view, modified from Pontin, 1978), male on the right (lateral view, modified from Koste, 1978). H – head, B – body, F – extended foot, A – anus position, BR – brain, CC – coronal cilia, CV – contractile vesicle, DA – dorsal antenna, EYE – eye, EGG – egg, FB – flame bulb, GG – gastric gland, I – intestine, J – jaws, L – lorica, LA – lateral antenna, M – muscle, P – pharynx, PE – penis, PG – pedal gland, PR – prostate, PT – protonephridium, S – spine, SC – sensory cirry, ST – stomach, T – toes, TE – testis, YG – yolk gland (after Telesh & Heerkloss, 2002).

Although rotifers can be considered as a relatively small phylum, they are extremely important in the environments that they inhabit because their reproductive rates are fastest for any metazoan (Nogrady et al., 1993). They can populate vacant niches with exceptional rapidity, convert primary (algal) and bacterial production into a form usable for secondary consumers (e.g. insect larvae and fish fry), and perform this transformation with remarkable efficiency producing up to 95% of total zooplankton biomass (e.g. in rivers and estuaries) (Telesh, 1995; Telesh & Heerkloss, 2002).

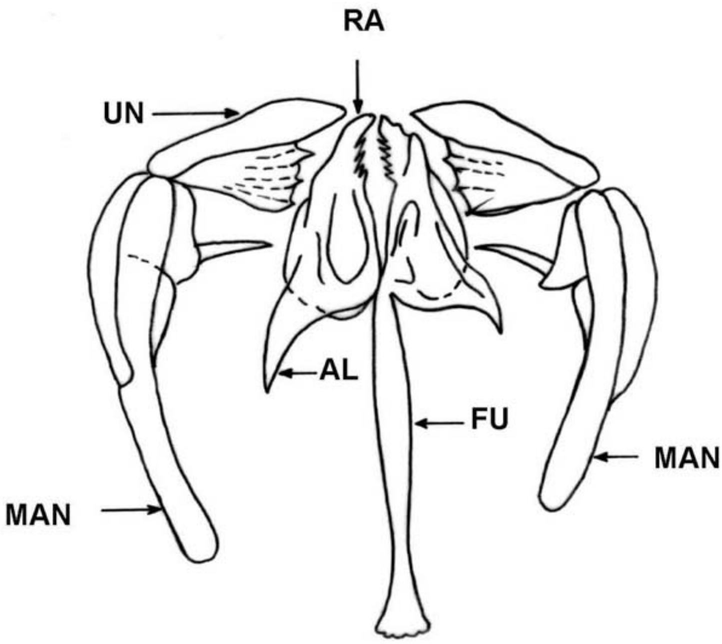


Figure 5.1.6. General structure of trophi, dorsal view: RA – ramus, UN – uncus, MAN – manubrium, FU – fulcrum, AL – alula (after Telesh & Heerkloss, 2002).

Cladocera
(Plates 5.3.6 – 5.3.10)

The commonly accepted today name of the order Cladocera according to Fryer (1987) belongs to a group of crustaceans of polyphyletic origin (see Telesh & Heerkloss, 2004, and references therein). The order Cladocera includes crustaceans which nearly all, with exception of several species, range in size from 0.2 to 3.0 mm. Cladocera are primarily freshwater organisms, and aside from rapid streams and strongly polluted waters, they can be abundant in every water body. In the estuaries, the greatest abundance of species may be collected in the vegetation, and at margins of the macrophytes stands and open water. Many species inhabit weedy littoral areas, some live on/near bottom. Limnetic forms (*Daphnia*, *Diaphanosoma*, *Holopedium*, *Leptodora* and others) are usually light-coloured and translucent; littoral and bottom species are ranging in colour of carapace and body tissues from yellowish-brown to reddish-brown, greyish, or nearly black.

The general schemes of body morphology of different cladocerans are presented on Figures 5.1.7 – 5.1.9.

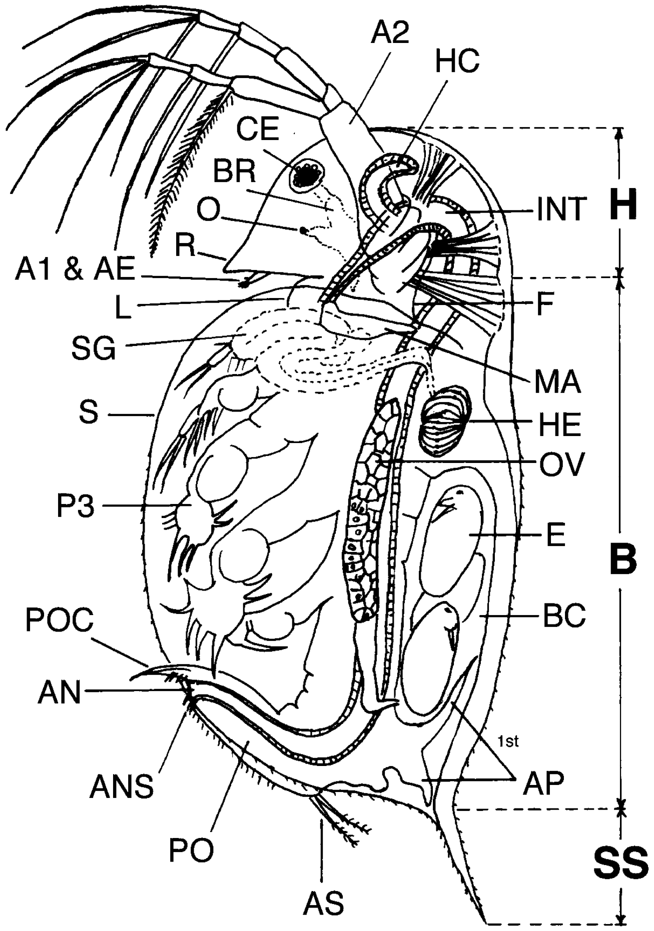
In the Baltic Sea, the Onychopod cladocerans from the genera *Podon*, *Pleopsis* and *Evadne* can be very abundant, especially in spring time (see also Chapter 2). *Evadne* individuals consume dinoflagellates and tintinnids, various other particles as well as small zooplankters. *Bosmina* spp. are among other common zooplankters in the open Baltic waters. The majority of species and almost all common ones are eurythermal. Many species can withstand oxygen concentrations of less than one part per million. **The taxonomy of the genus *Bosmina* even now remains a field in need of revision** (see review in Telesh & Heerkloss, 2004, p. 36).

Patchiness in spatial distribution and diurnal vertical migrations are common features of cladoceran crustaceans. However, characteristics of aquatic environment expose greater changes along the vertical than along the horizontal dimensions in the water body. Thus, the two contrasting needs of many zooplankters: to feed within the most illuminated zone and not to be seen by visual predators – result in regular movements of the whole populations into and out of the upper illuminated layers (Brandl, 2002).

Among cladocerans there are three non-indigenous species that have recently invaded different regions of the open Baltic Sea: *Cercopagis pengoi*, *Evadne anonyx* and *Cornigerius maeoticus* (for details see: Ojaveer & Lumberg, 1995; Rodionova et al., 2005; Rodionova & Panov, 2006, and the references in Chapter 2).

The great importance of planktonic Cladocera in the aquatic trophic webs as food for fish was emphasised first in late XIX century, and since then by innumerable investigators (see also Telesh & Heerkloss, 2004, and references therein). The dynamics of fish and zooplankton have been intimately linked ever since fish evolved from macrophagy to microphagy (Kerfoot & Lynch, 1987). Various studies of the stomach contents of young fish show from 1% to 95% Cladocera by volume, and very few studies show less than 10% (Pennak, 1978). However, some cladoceran species (e.g. a large-bodied predatory cladoceran of the Ponto-Caspian origin, *Cercopagis pengoi*, one of the recent invaders in the Baltic Sea), being a suitable food item for planktivorous fish, may also demonstrate structural and functional impact on zooplankton community thus performing competitive interactions for food (smaller crustaceans) with fish populations as shown recently for the Baltic Proper (Gorokhova, 1998), Gulf of Riga (Ojaveer & Lumberg, 1995), and Gulf of Finland (Antsulevich & Välipakka, 2000; Telesh et al., 2000; Telesh & Ojaveer, 2002).

In general, the role of zooplankton for the earlier juvenile fish is critical to high fish survival so that they can take advantage of an abundance of phytoplankton and detritus when available (Fernando, 2002).



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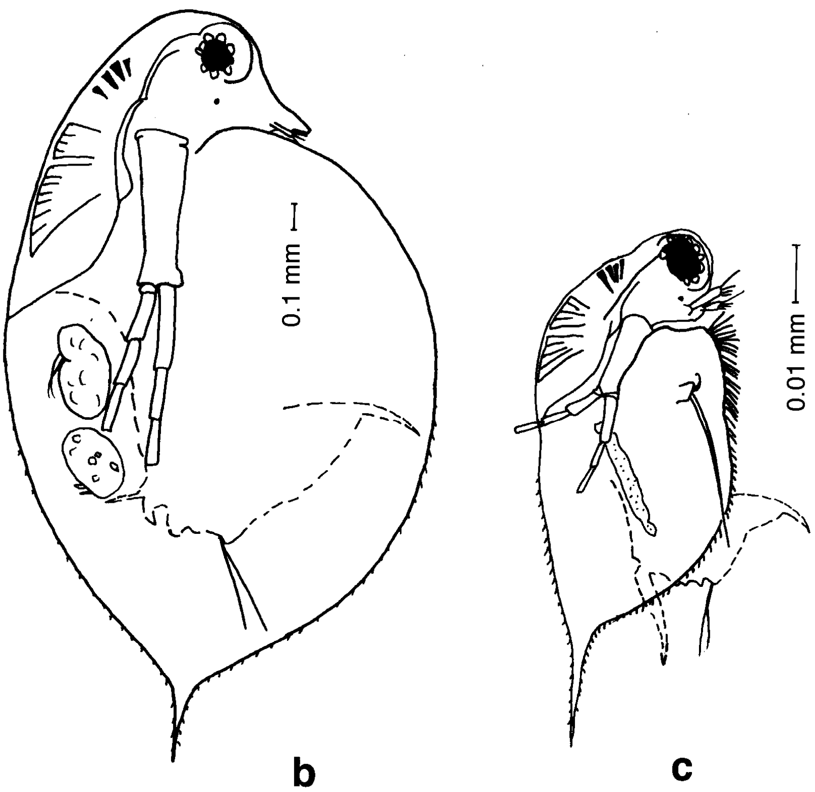

Figure 5.1.7. *Daphnia*, schematic, lateral. **a,** Female: A1 – first antenna (antennule), A2 – second antenna, AE – aesthetascs, AN – anus, ANS – anal spines, AP – abdominal processes, AS – abdominal setae, B – body, BR – brain, BC – brood chamber, CE – compound eye, E – embryo, F – fornix, H – head, HC – hepatic caecum, HE – heart, INT – intestine, L – labrum, MA – mandible, O – ocellus, OV – ovary, PO – postabdomen, POC – postabdominal claw, P1 – P5 – trunk limbs 1–5, R – rostrum, S – shell, SG – shell gland, SS – shell spine; **b,** *D. pulex*, female; **c,** *D. pulex*, male (after Telesh & Heerkloss, 2004).

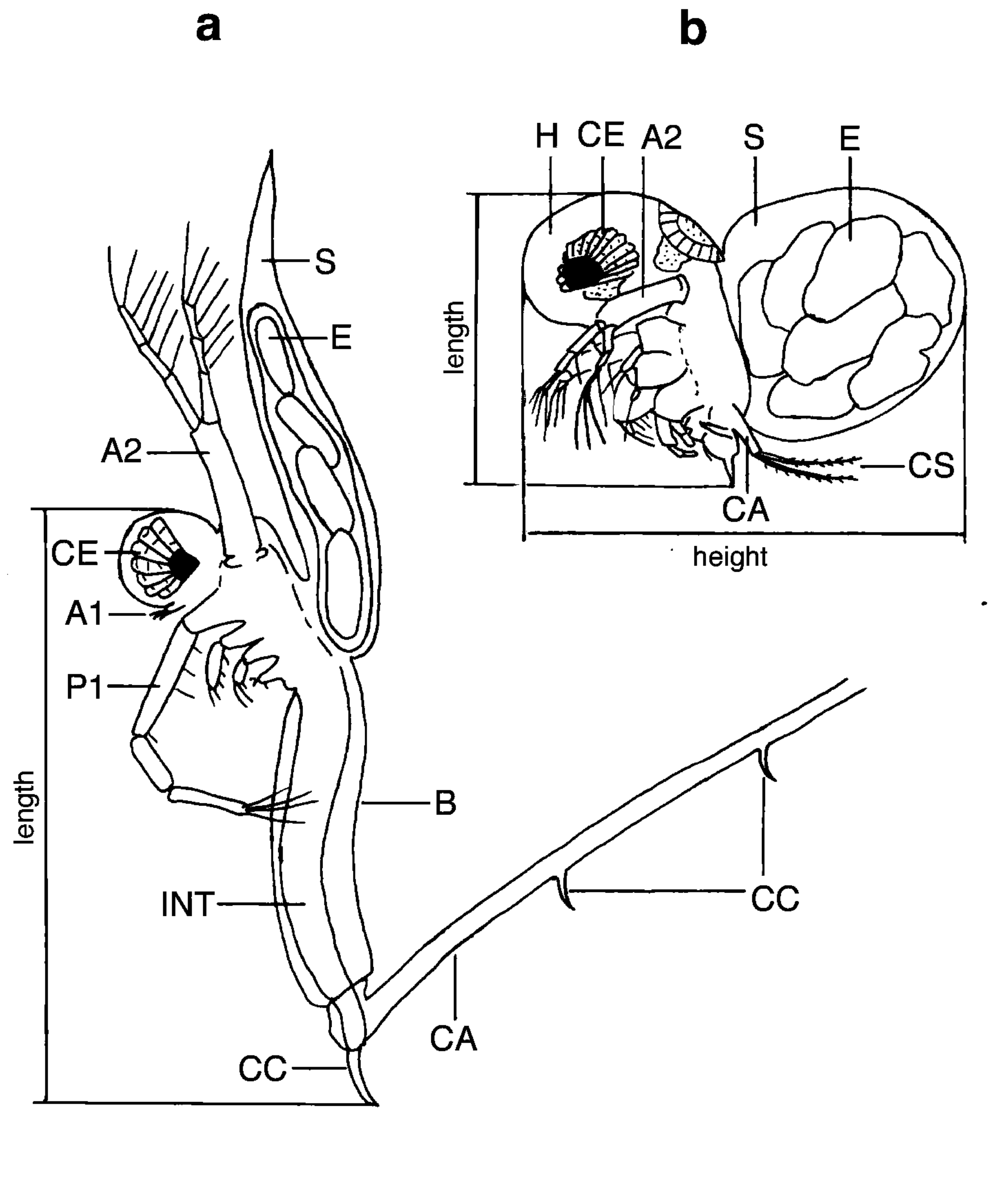

Figure 5.1.8. Morphology of Onychopoda: **a** – *Cercopagis*, **b** – *Polyphemus*: A1 – first antenna (antennule), A2 – second antenna, B – body, CA – caudal appendage, CC – caudal claw, CE – compound eye, E – parthenogenetic embryos, CS – caudal setae (setae notatoria), H – head, INT – intestine, P1 – trunk limb (thoracic leg) 1, S – shell (brood chamber) (after Telesh & Heerkloss, 2004).

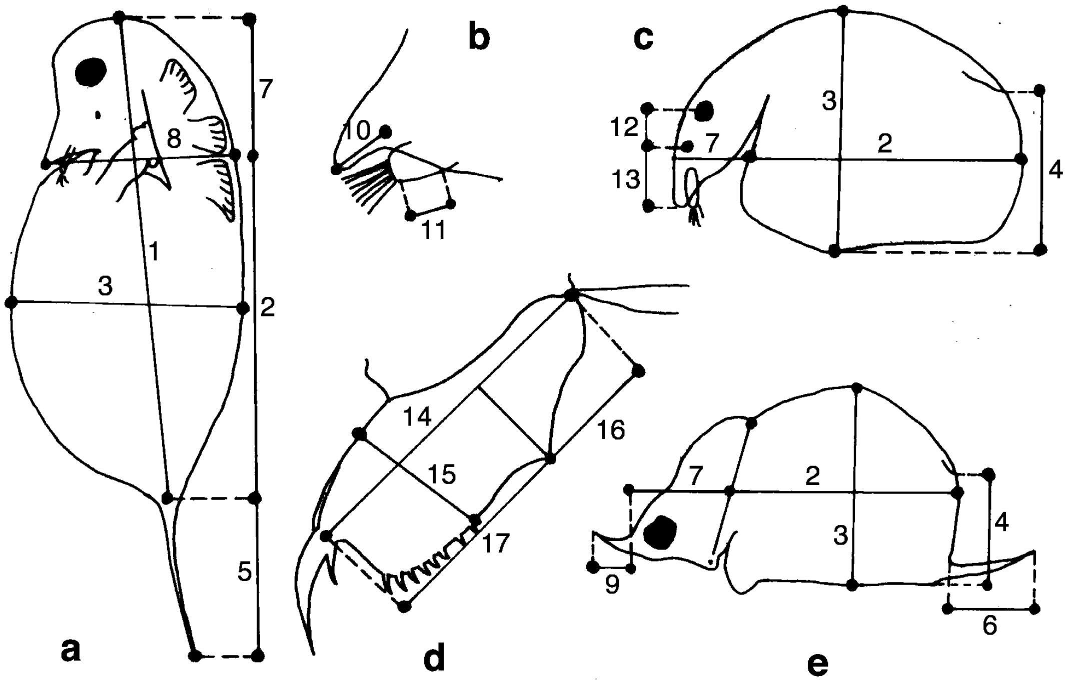

Figure 5.1.9. How to measure Cladocera: **a, b** – *Daphnia*; **c, d** – *Alona*; **e** – *Scapholeberis*. **1** body length, **2** length of carapace, **3** maximum height of valve, **4** height of posterior margin of valves, **5** length of shell spine, **6** length of mucro, **7** length of head, **8** height of the posterior margin of head shield, **9** length of the horn on the proximal edge (vertex) of head shield, **10** length of rostrum, **11** length of antenna 1, **12** distance between eye and ocellus, **13** distance between ocellus and the end of rostrum, **14** length of postabdomen, **15** maximum width of postabdomen, **16** length of proximal part of postabdomen, **17** length of distal part of postabdomen (from Telesh & Heerkloss, 2004, adapted from Flössner, 2000).

Copepoda
(Plates 5.3.11 – 5.3.28)

Copepoda is a very diverse and the most abundant group of metazoans in the pelagial of the world's oceans (Larink & Westheide, 2006). Free-living planktonic copepods range in length from 0.5 to 5 mm. Copepod crustaceans from three suborders inhabit the open waters of the Baltic Sea: Calanoida, Cyclopoida and Harpacticoida (Fig. 5.1.10). These crustaceans form a ubiquitous component of the zooplankton community.

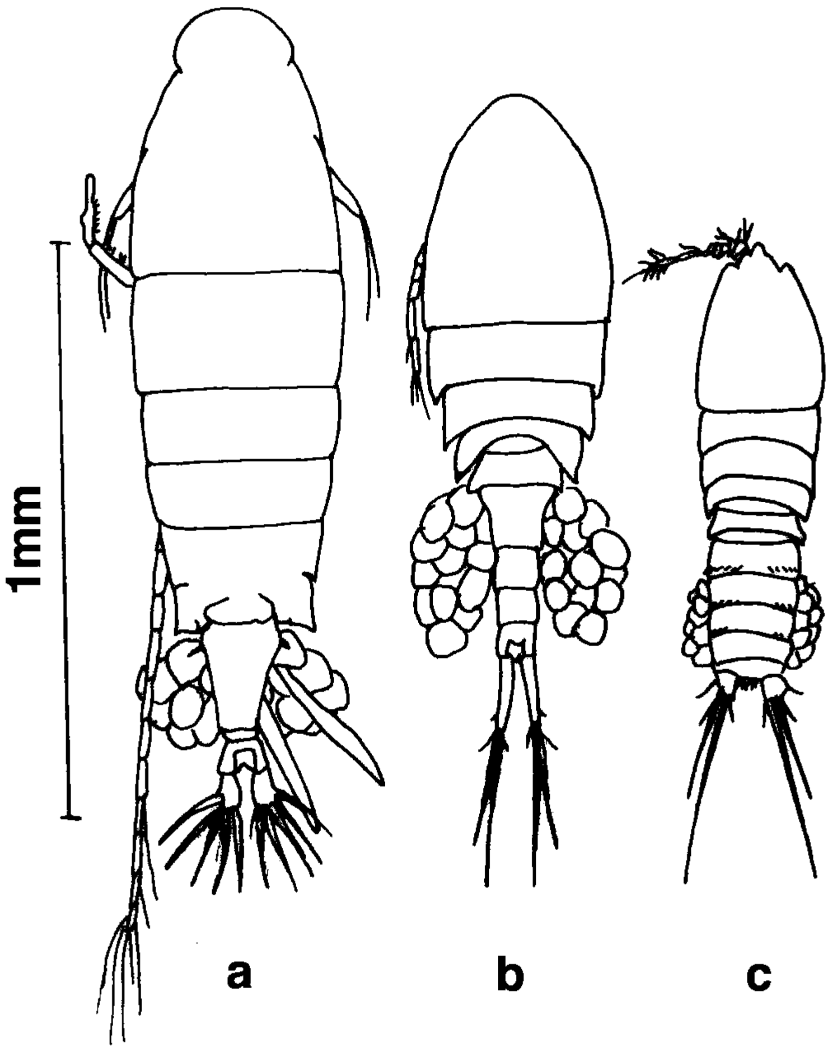

Figure 5.1.10. Scheme of calanoid (**a**), cyclopoid (**b**) and harpacticoid (**c**) copepods (after Telesh & Heerkloss, 2004).

Copepods differ in size, external morphology, ecology and feeding habits. Most Calanoida are free-living, planktonic, herbivorous, fine particles filter feeders. Cyclopoida are also planktonic crustaceans but very often they inhabit near-bottom biotopes; they are generally micro-predators that feed on small invertebrates and even fish larvae but also consume algae. Harpacticoida are mainly meiobenthic or epibenthic grazers, they occur in plankton only sporadically, being washed out from their bottom habitats by strong water movements. In general Harpacticoida are only temporarily in plankton, although these crustaceans are often found in zooplankton samples collected in the shallow estuarine waters.

Copepods are food to many predators, mainly planktivorous fish. The choice of a copepod as a prey is a function of its size, morphology, motion (angle, speed, escape ability) and pigmentation. The coloured species are more vulnerable to predation than pale or transparent ones. Presence of fish can influence physiological parameters and population dynamics of copepods. To limit predation, some copepods can retreat to habitats devoid of the predator, perform vertical migrations, form swarms, or enter into dormancy (Dussart & Defaye, 2001).

Copepods have different tolerance to salinity; the presence or absence of some species allows deductions on the physical-chemical characteristics or the degree of pollution of the environment (Dussart & Defaye, 2001).

However, the important role of copepods as biological indicators cannot be assessed unless the copepod species identification is properly fulfilled. Taxonomic differentiation of copepods is based mainly on external morphology of mature females and males. Species identification of copepods is an important though tedious procedure. Shape, colour and size of the body, relative size of the appendages (particularly the length of antennules relative to the cephalosome or the urosome) and other measurements are noted. After general observations drawings of the whole animal should be made.

For cleaning the crustacean and making its body more transparent, the animal must be kept in a drop of concentrated lactic acid ($CH_3CHOHCOOH$) for a time from 1 h up to overnight, depending on the size of the crustacean. Sometimes it is possible to recognize the copepod species without dissection (Alekseev, 2002; Telesh & Heerkloss, 2004). However, in most cases species identification of copepods requires not only examination of the whole crustacean under the microscope but also a dissection and mounting of relevant structures. For more details of this procedure see Downing and Rigler (1984), Huys and Baxshall (1991), ICES (2000), Dussart and Defaye (2001), Alekseev (2002).

Copepods can be of different shape: elongated, fusiform, or cylindrical. General schemes of body morphology of cyclopoid and calanoid copepods are presented in Figures 5.1.11 and 5.1.12; schematic drawings of their nauplia and copepodites are given in Figures 5.1.13 – 5.1.15.

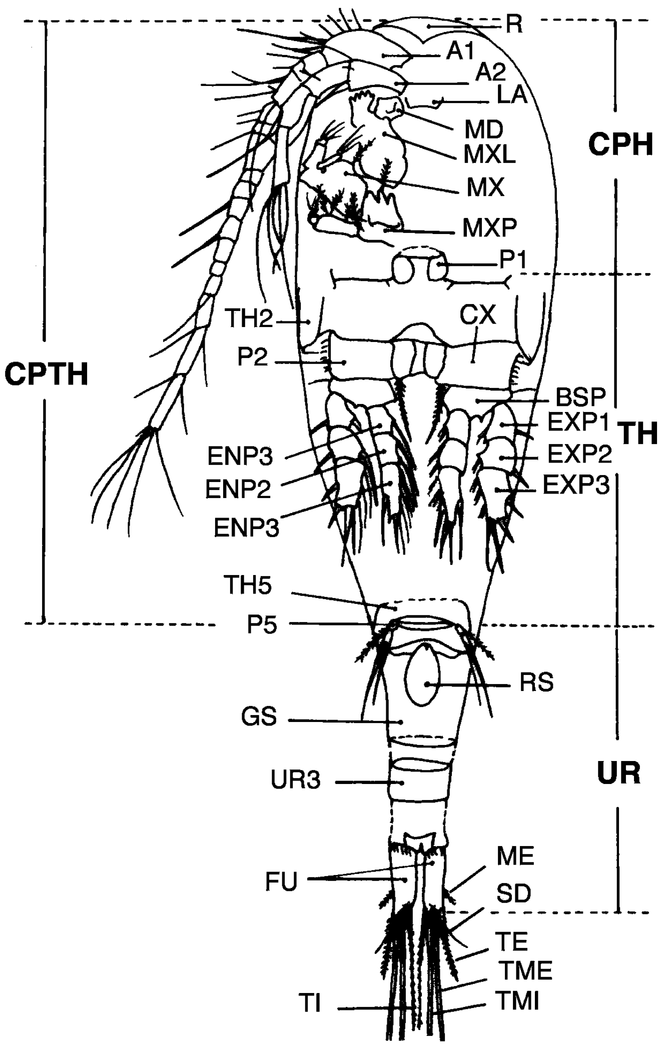

Figure 5.1.11. Morphology of a female cyclopoid (ventrally): CPH – cephalosome, TH – thoracosome, CPTH – cephalothorax, UR – urosome, R – rostrum, A1 – antennule, A2 – antenna, LA – labrum, MD – mandible, MXL – maxillule, MX – maxilla, MXP – maxilliped, P1, P2, P5 – swimming legs 1, 2, 5, TH2, TH5 – thoracic somites 2 and 5, CX – coxa, BSP – basipodite, EXP1, EXP2, EXP3 – exopodites 1-3, ENP1, ENP2, ENP3 – endopodites 1-3, GS – genital double somite, RS – seminal receptacle (= *receptaculum seminis*), UR3 – urosomite 3, FU – furca, ME – marginal (external) furcal seta, SD – dorsal furcal seta, TE – terminal external furcal seta, TME – terminal medial external seta, TMI – terminal medial internal seta, TI – terminal internal furcal seta (from Telesh & Heerkloss, 2004, after Dussart & Defaye, 1995, with modifications).

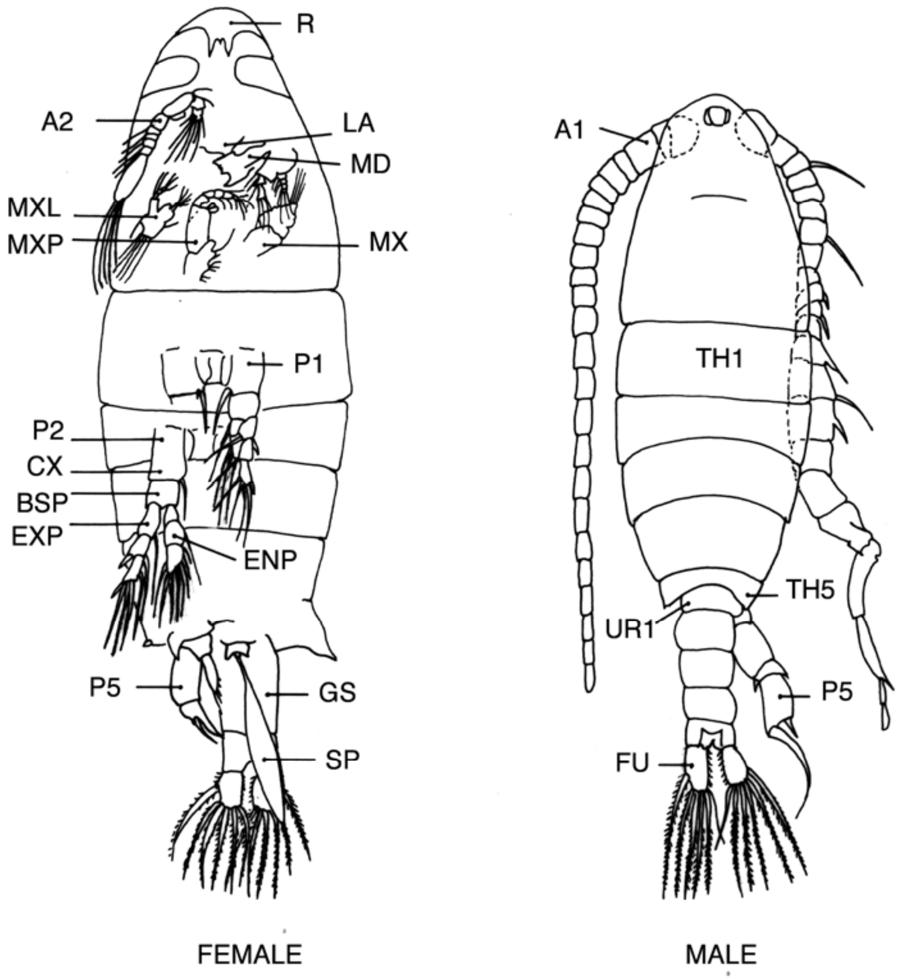

Figure 5.1.12. Morphology of a female (ventrally) and male (dorsally) calanoid: R – rostrum, A1 – antennule, A2 – antenna, LA – labrum, MD – mandible, MXL – maxillule, MX – maxilla, MXP – maxilliped, P1, P2, P5 – swimming legs 1, 2, 5, TH1, TH5 – thoracic somites 1 and 5, CX – coxa, BSP – basipodite, EXP – exopodite 1, ENP – endopodite, GS – genital double somite, RS – seminal receptacle (= *receptaculum seminis*), UR1 – urosomite 1, FU – furca, SP – spermatophore (from Telesh & Heerkloss, 2004, after Dussart & Defaye, 1995, with modifications).

Most copepods reproduce sexually; however some cases of parthenogenesis have been reported and checked experimentally (Dussart & Defaye, 2001). The sex ratio (males/females) in a copepod population is usually below 1, often due to a different behaviour of the sexes.

Sexual reproduction implies that the male deposits a spermatophore near the genital aperture of the female. Fertilized eggs develop within a single egg-sac attached to the ventral side of the genital somite centrally in Calanoida, or in two symmetrically located egg-sacs in Cyclopoida. The duration of the embryonic development depends on many factors, among which temperature is one of the most important. When embryonic development is completed, in most copepods the female looses the egg-sac(s), and the eggs hatch together.

Among Crustacea, copepods have been cited as exhibiting the most complete example of metamorphosis (Dussart & Defaye, 2001). They consequently pass through 6 nauplial (Fig. 5.1.13) and 5 copepodite stages (Figs. 5.1.14, 5.1.15) before maturation. The eggs hatch into a larva called nauplius – the typical planktivorous larva of crustacean arthropods. In calanoids, naupliar larvae are ovoid, slender and somewhat compressed laterally (Fig. 5.1.13a). In cyclopoids, nauplii are dorsoventrally compressed and have a compact, pear-shaped body (Fig. 5.1.13b).

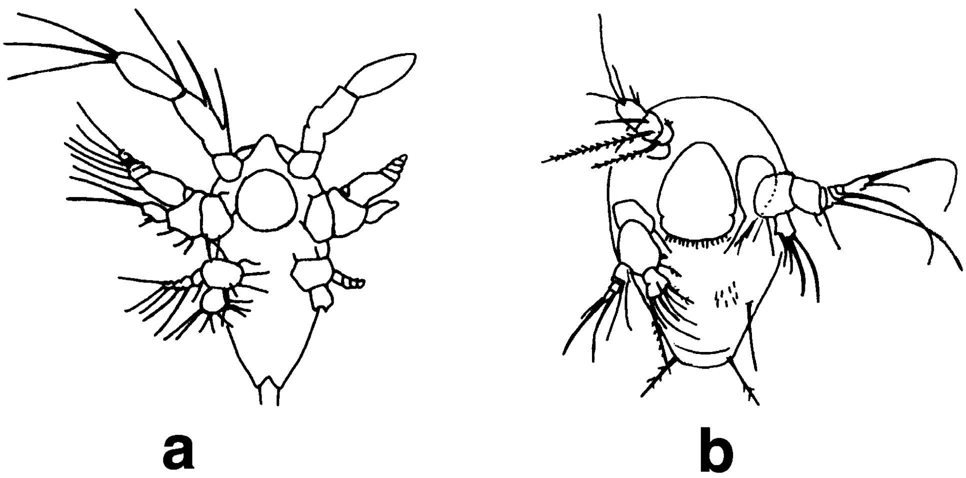

Figure 5.1.13. Nauplii N2 of calanoid (**a**) and cyclopoid (**b**) copepods, ventral view (modified from Einsle, 1993).

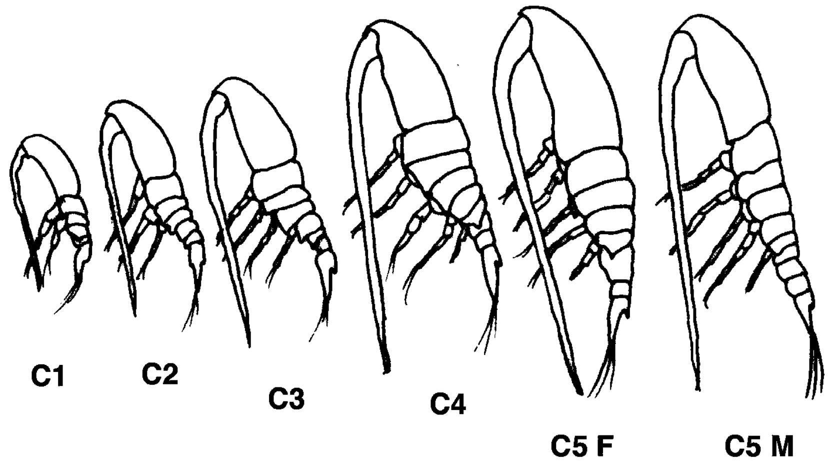

Figure 5.1.14. Development of copepodite stages (C1-C5, C5F – female, C5M – male) of a calanoid copepod, lateral view (modified from Einsle, 1993).

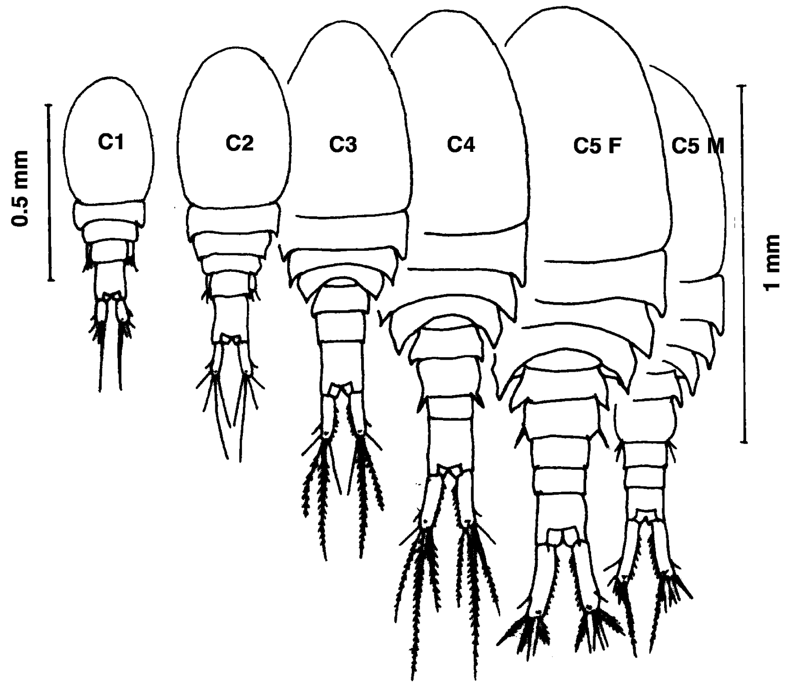

Figure 5.1.15. Development of copepodite stages of a cyclopoid copepod, dorsal view (modified from Einsle, 1993). Abbreviations as in Fig. 5.1.14.

Chaetognatha

(Plate 5.3.30)

The majority of Chaetognatha (arrow worms) are holoplanktonic marine invertebrates that can reach relatively high densities in the sea pelagial waters.

Arrow worms of the genera *Sagitta* and *Parasagitta* perfectly represent the type of optimally adapted voracious predators in the plankton community: they are relatively large (15-45 mm), fast, visual, transparent and streamlined animals that see and attack their prey by short forward darting motions when attacking various pelagic organisms, mostly copepods but also small fish, as large as themselves. A *Sagitta* may consume the equivalent of 64% of its body mass in food per day; otherwise, they are an important prey for fish (Larink & Westheide, 2006).

Chaetognaths are protandrous hermaphrodites: paired testes are located in the tail of the elongate body, paired ovaries – in the posterior part of the trunk (Fig. 5.1.16). Arrow worms have no larvae: development is direct and very rapid for the feeding juveniles.

The most common species in the Baltic Sea are *Parasagitta elegans* and *Parasagitta setosa* (Plate 5.3.30). These two species are difficult to distinguish, especially when the specimens are juveniles; but when adult, *P. elegans* becomes larger (up to 20 mm) than *P. setosa* (up to 14 mm). Besides, *P. setosa* is known to prefer more saline waters, and thus its distribution varies with the extent to which Atlantic oceanic water penetrates into the coastal water bodies (Larink & Westheide, 2006).

Chaetognaths are very mobile and are able to swim against substantial water current. They migrate horizontally some hundred meters per day and undergo daily vertical migration. Many of them escape when sampling is performed with inappropriately small plankton net.

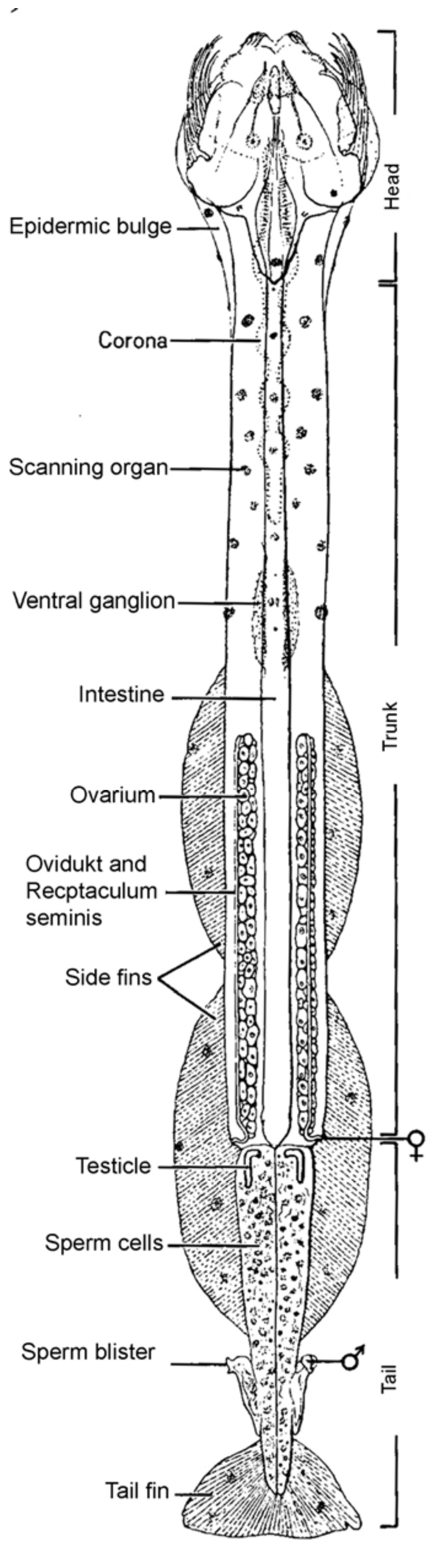

Figure 5.1.16. *Sagitta bipunctata*, scheme of body morphology (modified from Storch & Welsch, 1999).

Appendicularia
(Plate 5.3.29)

Appendicularia are the exclusively holoplanktonic tunicates (Chordata, Tunicata). They are also called Larvacea because of their apparent retention of ascidian larval organisation. Appendicularians are tiny solitary animals with peculiar anatomy and unique filter feeding system.

The two types of larvacean which are commonly found in plankton samples can be readily separated by reference to the shape and size of their body and tail. Members of the Oikopleuridae have a relatively compact body and linear tail (Fig. 5.1.17 a), while Fritillaridae have a more delicate body and thin, broad tail (Fig. 5.1.17 b).

Oikopleura dioika is one of the most common appendicularians, rather abundant in the Baltic Sea (Plate 5.3.29). It looks like one of the tadpole ascidian larvae, but the prominent tail with notochord and nerve cord is persistent. It is positioned below the trunk, perpendicular to the long axis of the animal and is five times longer than the trunk, reaching 3 mm.

In the plankton samples commonly only these “naked” animals will be observed. Meanwhile actually in the sea they live inside of a mucous construction, the so called “house” (Fig. 5.1.18). This construction is almost spherical in shape; it consists of a number of intercommunicating chambers, funnels, filters, intake openings and outlets, and functions as a complicated filtration system. Even very small particles (below 0.5 µm) can be trapped from the water by this system, accumulated and transported to the mouth of the appendicularian. Interestingly enough, it was here in the appendicularian house that the presence of nanoplankton organisms in the sea was first demonstrated by the filtering activity of these animals (Larink & Westheide, 2006).

Water is moved through the appendicularian house by the pressure generated by the beats of the tail. When the tail beats slowly, the animal hardly moves through the water, and filtering is optimal. If particles are few, the tail beats more rapidly; then water is ejected in a greater quantity and thus a jet effect propels the house forward.

The fragile construction of the house is secreted by gland cells (oikoblasts) in the epidermis. When filters are clogged with particulate matter, the animal deserts the house. This also happens when it is captured by plankton net or disturbed otherwise. Before leaving the house, the appendicularian builds (secretes) a new proto-house which can be inflated within few seconds; 4 to 16 new houses can be secreted by one appendicularian every day.

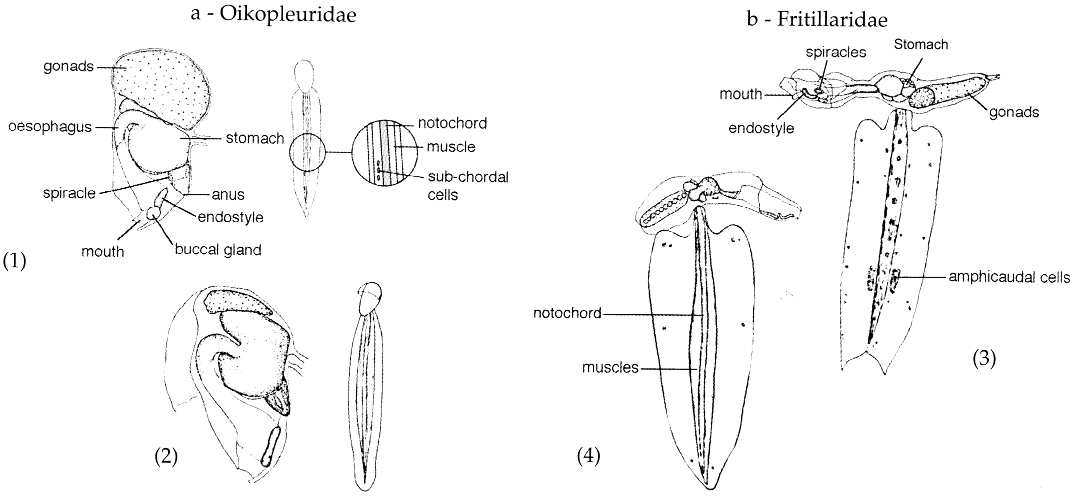

Figure 5.1.17. Appendicularia, schematic lateral view: a – Oikopleuridae, b – Fritillaridae; 1 – *Oikopleura dioika*, detail of body, whole animal and diagrammatic magnification of tail; 2 – *O. longicauda*, detail of body, whole animal; 3 – *Fritillaria megachile*; 4 – *F. haplostoma* (from Fenaux, 1967, cited after Gibbons, 1997, with modification).

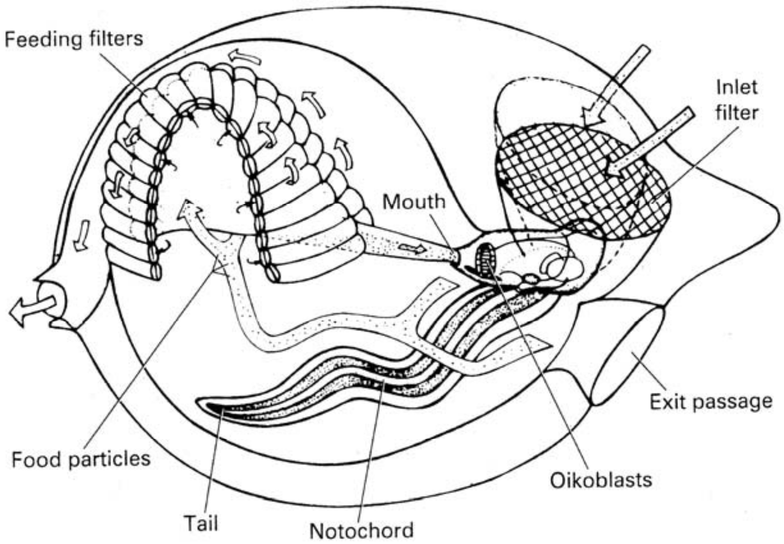

Figure 5.1.18. *Oikopleura dioika* in its "house" (modified from Larink & Westheide, 2006).

Polychaeta
(Plates 5.3.33 and 5.3.34)

Polychaetes are the basal group of the segmented worms (Annelida). The group comprises ca. 9,000 species distributed almost exclusively in the marine environment. They occur in the pelagial (a) as larval stages lasting a few hours to several weeks, (b) as modified swimming stages of mature males or females (epitokes, heteronereids), or (c) as transparent pelagic holoplanktonic species, the latter belonging to seven families.

Among the enormous diversity of reproductive modes in polychaetes, **epitoky** is the most striking. Epitokous planktonic stages are mature individuals of mainly the benthic species, which have undergone morphological, physiological and behavioural modifications that enable them to leave the bottom and to swim and broadcast their gametes in the water column.

These metamorphosed sexually mature worms are produced by two processes.

(A) The whole animal is transformed into a swimming epitoke, and once the gametes are released after a short pelagic existence this animal dies, or sometimes reverts to the atokous state (epigamy).

(B) The (mostly) posterior part of the mature worm is modified, usually equipped with a new head, and then becomes detached as a free-swimming gamete-baring stolon. Whereas this stolon dies after the release of gametes, the unchanged anterior benthic stock of the worm continues to live for further reproductive activity by multiple stolonisation (=schizogamy).

The **trochophora** (Fig. 5.1.19, left) is a typical larva of polychaetes. Often it follows a spherical **prototrochophore** that is entirely covered with short cilia. The trochophore is characterised by a ciliary band, the **prototroch**, which encircles the body anterior to the mouth and is used for locomotion and feeding. Another parallel circumferential band of cilia is **metatroch**, which posteriorly borders the mouth region. A ciliated region between the two bands is called the food groove.

Early larvae with only a few segments often are called **metatrochophores** or polytrochous larvae if they possess additional ciliary bands. In metatrochophore I, parapodia are not yet developed. Larvae with additional outer segmental structures are called metatrochophore II. Segmented larvae with functioning parapodia and prominent bundles of chaetae are called **nectochaetae** (Fig. 5.1.19, right, 5.1.20).

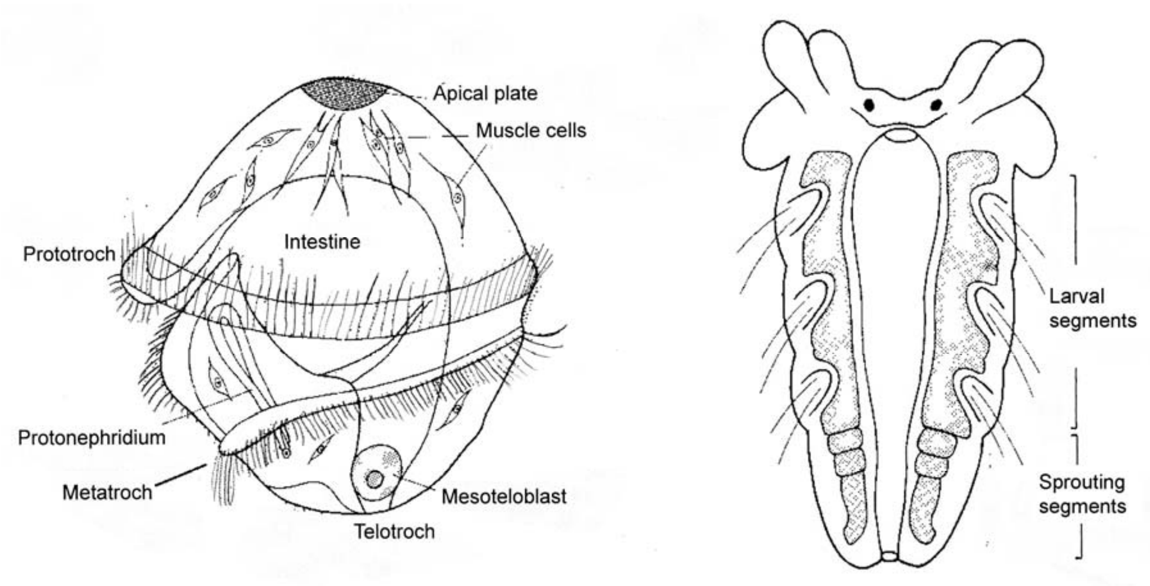

Figure 5.1.19. Larvae of Polychaeta: trochophora (left) and nectochaeta (right) (from Westheide & Rieger, 1996, with modification).

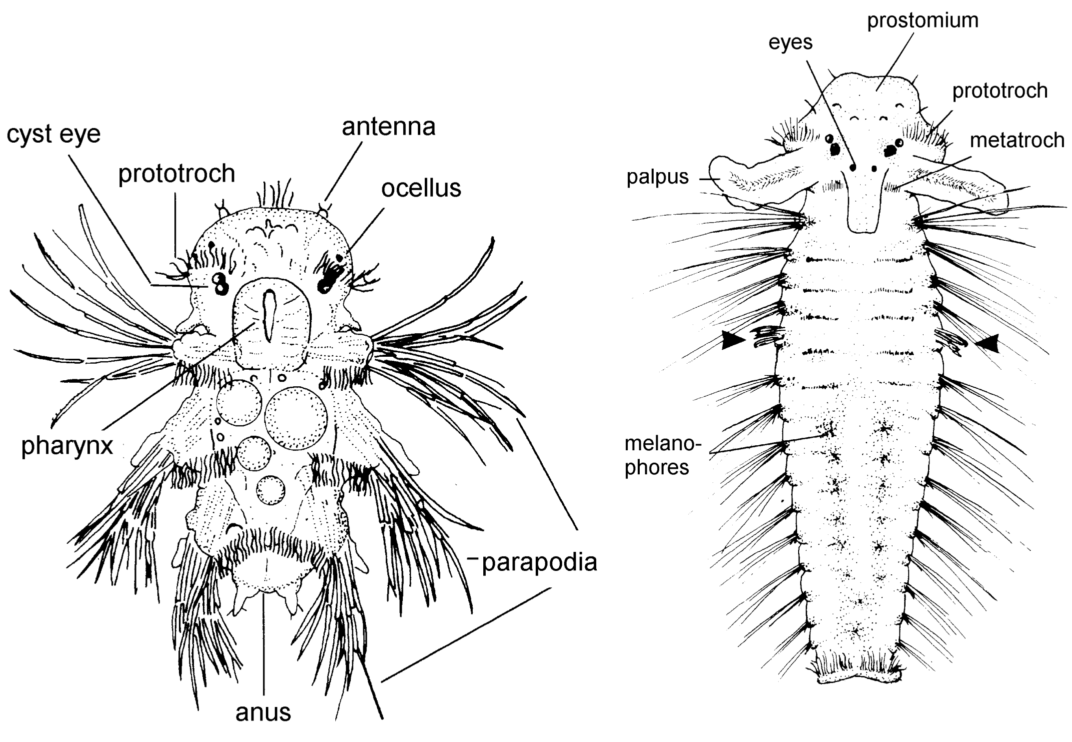

Figure 5.1.20. Larvae of Polychaeta: nectochaeta at different stages of development (from Storch & Welsch, 1999, with modification).

Trochophores and metatrochophores of polychaetes are easily confused with each other and are difficult to assign to a specific genus, or even to a family taxon.

Larvae of Spionidae (palp worms, the largest group of benthic, sessile of hemi-sessile polychaetes) are generally the most common developmental stages of polychaetes that can be found in plankton throughout the year. High abundance of polychaetes larvae in the pelagial of the sea is not only due to the large number of species in the coastal areas but also to the often long-lasting periods of their development.

5.2. Checklist of meso- and macrozooplankton of the open Baltic Sea

The checklist of zooplankton organisms larger than 200 µm inhabiting different regions of the open Baltic Sea is given in Table 5.2.1.

In most cases only valid species names and their most common synonyms are included in the table, without indication of the names of subspecies, forms or varieties which sometimes may be numerous within a certain species, like for example in the rotifer genera *Brachionus* and *Keratella*.

Basing on our previous publications on the estuarine zooplankton of the Baltic Sea (Telesh & Heerkloss, 2002, 2004) we admit that a number of zooplankton species which are not indicated in this checklist or those mentioned as "not present" in a certain area in fact may occur there but they either have not been found/sampled yet or were identified only by the genus name.

In total, 217 taxa of holoplanktonic animals (Cnidaria – 15, Ctenophora – 5, Turbellaria – 1, Rotifera – 83, Cladocera – 37, Copepoda – 63, Pteropoda – 1, Polychaeta – 7, Chaetognatha – 3, Copelata – 2) generally belonging to meso- and macrozooplankton are included in the checklist, and more than 30% of these organisms are illustrated by the photographs (see Chapter 5.3).

Rotifers are conventionally considered here as mesozooplankters, contrary to ciliates that are representing microzooplankton; the latter were described above in a separate Chapter 4.

Species names and synonymy of Rotifera are given after Kutikova (1970) and Koste (1978), names and synonyms of other species – according to the Integrated Taxonomic Information System (ITIS, http://www.itis.gov), the European Register of Marine Species (ERMS, http://www.marbef.org/data/erms.php), and the World Register of Marine Species (WoRMS, http://www.marinespecies.org).

Table 5.2.1.

Species composition of meso- and macrozooplankton species of the open Baltic Sea: **BP** – Baltic Proper; **WBS** – Western Baltic Sea; **NBS** – Northern Baltic Sea, **SBS** – Southern Baltic Sea; **EBS** – Eastern Baltic Sea ("+" present; no sign = species not found). Species **in bold** are illustrated by photographs.

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
	Cnidaria, Anthomedusae (Plates 5.3.38, 5.3.39)					
1	***Euphysa aurata*** **Forbes, 1848** (Syn.*: *Corymorpha aurata* (Forbes, 1848); *Dahlgrenella farcta* Miles, 1937; *Heteractis aurata* (Forbes, 1848))	+	+			
2	*Euphysa tentaculata* Linko, 1905 (Syn.: *Corymorpha tentaculata* (Linko, 1905))	+	+			
3	*Halitholus cirratus* Hartlaub, 1913	+	+		+	
4	*Hybocodon prolifer* L. Agassiz, 1862	+	+			
5	*Rathkea octopunctata* (M. Sars, 1835) (Syn.: *Cytaeis octopunctata* M. Sars, 1835)	+	+			
6	*Sarsia tubulosa* (M. Sars, 1835) (Syn.: *Coryne tubulosa* (M. Sars, 1835); *Oceania tubulosa* M. Sars, 1835)	+	+			
	Cnidaria, Trachymedusae					
7	*Aglantha digitalis* (O.F. Müller, 1776) (Syn.: *Aglantha digitale* (O. F. Müller, 1776))	+	+			
	Cnidaria, Scyphomedusae					
8	*Aurelia aurita* (Linnaeus, 1758) (Syn.: *Aurelia coerulea* von Lendenfeld, 1884)	+	+	+	+	
9	*Clytia hemisphaerica* (Linnaeus, 1767) (Syn.: *Phialidium hemisphaericum* (Linnaeus, 1767)	+				
10	*Cyanea capillata* (Linnaeus, 1758)		+			
11	*Haliclystus auricula* O. Fabricius, 1780 (Syn. : *Lucernaria auricula* Rathke, 1806)		+			
12	*Lucernaria quadricornis* O.F. Müller, 1776		+			
13	*Rhizostoma octopus* (Mayer, 1910) (Syn.: *Rhizostoma pulmo octopus* Mayer, 1910)		+			

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
	Cnidaria, Leptomedusae (Plate 5.3.38)					
14	*Melicertum octocostatum* (M. Sars, 1835) (Syn.: *Oceania octocostata* M. Sars, 1835)	+	+		+	
15	***Obelia geniculata*** (Linnaeus, 1758)	+	+		+	
	Ctenophora (Plates 5.3.36, 5.3.37)					
16	*Beroe cucumis* Fabricius, 1780	+	+			
17	*Beroe gracilis* Künne, 1939	+	+			
18	*Bolinopsis infundibulum* O.F. Müller, 1776 (Syn.: *Bolinopsis alata* (L. Agassiz, 1860); *B. septentrionalis* (Mertens, 1933))	+	+			
19	***Mnemiopsis leidyi*** A. Agassiz, 1865	+	+	+	+	
20	*Pleurobrachia pileus* (O.F. Müller, 1776)	+	+	+	+	+
	Turbellaria (Plate 5.3.34)					
21	***Alaurina composita*** Metschnikow, 1865		+			
	Rotifera (Plates 5.3.1 – 5.3.5)					
22	*Anuraeopsis fissa* (Gosse, 1851)			+		
23	*Asplanchna* sp.			+		
24	*Asplanchna brightwelli* Gosse, 1850		+			+
25	***Asplanchna priodonta*** Gosse, 1850 (Syn.: *Asplanchna krameri* De Guerne, 1888; *A. priodonta pelagica* Zacharias, 1892)		+		+	+
26	*Asplanchna seiboldi* (Leydig, 1854)		+			
27	Bdelloidea indet.			+		+
28	*Brachionus* sp.			+	+	+
29	***Brachionus angularis*** Gosse, 1851 (Syn.: *Brachionus testudo* Ehrenberg, 1853; *B. syennensis* Schmada, 1859; *B. papuanus* Daday, 1897; *B. urceolaris f. angulatus* Seligo, 1900)		+	+	+	+
30	***Brachionus calyciflorus*** Pallas, 1766 (Syn.: *Brachionus longispinus* Schrank, 1803; *B. pala* Eghrenberg, 1838; *B. bicornis* Bory de St. Vincent, 1826; *B. dicipiens* Plate, 1886)		+	+	+	+
31	***Brachionus plicatilis*** O.F. Müller, 1786 (Syn.: *Brachionus hepatotomeus* Gosse, 1851; *B. muelleri* Ehrenberg, 1834; *B. orientalis* Rodewald, 1937)		+			

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
32	***Brachionus quadridentatus*** Hermann, 1783 (Syn.: *Brachionus bakeri* Müller, 1786; *B. capsuliflorus* Pallas, 1766; *B. quadricornis* Schrank, 1803; *B. octodentatus* Bory de St. Vincent, 1826)		+	+		
33	*Brachionus rubens* Ehrenberg, 1838 (Syn.: *Brachionus urceus rubens* Ehrenberg, 1838)		+			
34	***Brachionus urceus*** (Linnaeus, 1758) (Syn.: *Tubipora urceus* Linnaeus, 1758; *Vorticella urceolaris* Linnaeus, 1767; *B. bursarius* Barrois & Daday, 1894; *B. sericus* Rousselet, 1907; *B. urceus* Von Hofsten, 1909; *B. urceolaris* Voigt, 1956, Rudescu, 1960)		+	+		
35	*Cephalodella catellina* (O.F. Müller, 1786))		+			
36	*Cephalodella megalocephala* (Glasscott, 1893)		+			
37	*Collotheca* sp.					+
38	*Collotheca mutabilis* (Hudson, 1885)		+			
39	*Collotheca ornata* (Ehrenberg, 1832)		+			
40	*Collotheca pelagica* (Rousselet, 1893)					+
41	*Colurella* sp.		+	+		+
42	***Conochilus unicornis*** Rousselet, 1892 (Syn.: *Conochilus leptopus* Forbes, 1893; *C. limneticus* Stenroos, 1898; *C. norvegicus* Burckhardt, 1943)				+	
43	*Dicranophorus* sp.				+	
44	*Encentrum pachypus* Remane, 1949		+			+
45	*Euchlanis* sp.					+
46	***Euchlanis dilatata*** Ehrenberg, 1832 (Syn.: *Euchlanis hipposideros* Gosse, 1851)		+	+	+	+
47	*Filinia brachiata* (Rousselet, 1901)			+		
48	***Filinia longiseta*** (Ehrenberg, 1834) (Syn.: *Triarthra longiseta* Ehrenberg, 1834)		+	+	+	+
49	***Filinia terminalis*** (Plate, 1886) (Syn.: *Filinia maior* (Colditz) Carlin, 1943; *Triarthra terminalis* Plate, 1886)		+	+	+	
50	*Hexarthra fennica* (Levander, 1892) (Syn.: *Pedalia fennica* (Levander, 1892))		+			+
51	***Kellicottia longispina*** (Kellicott, 1879) (Syn.: *Anuraea longispina* Kellicott, 1879; *Anuraea spinosa* Imhof, 1883; *Notholca longispina* Hudson & Gosse, 1889)			+	+	+

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
52	***Keratella cochlearis*** (Gosse, 1851) (Syn.: *Anuraea cochlearis* Gosse, 1851; *A. longistyla* Schmarda, 1859; *A. longispina* Imhof, 1883; *A. intermedia* Imhof, 1885)	+	+	+	+	+
53	***Keratella cochlearis baltica*** (Sokolova, 1927)	+	+	+	+	+
54	*Keratella cochlearis recurvispina* (Jägerskiöld, 1894)	+		+		+
55	***Keratella cochlearis tecta*** (Gosse, 1851)		+	+	+	+
56	***Keratella cruciformis eichwaldi*** (Levander, 1894) (Syn.: *Anuraea tecta* Plate, 1890; *A. eichwaldi* Levander, 1894; *A. cruciformis* var. *eichwaldi* Levander, 1911; *K. cruciformis* var. *eichwaldi* Remane, 1929; *K. cruciformis eichwaldi* Kutikova, 1970)	+	+	+	+	+
57	***Keratella quadrata*** (O.F. Müller, 1786) (Syn.: *Brachionus quadratus* (Müller, 1786); *Kerona octoceros* (Abildgaard, 1793); *Vaginaria squamula* (Schrank, 1803); *Anourella squamula* (Bory de St. Vincent, 1826); *Anuraea squamula* (Ehrenberg, 1832); *A. aculeata* (Ehrenberg, 1832); *A. octoceros* (Ehrenberg, 1834))	+	+	+	+	+
58	***Keratella quadrata platei*** (Jägerskiöld, 1894) (Syn.: *Keratella platei)*	+		+		+
59	*Lecane* sp.			+		+
60	*Lecane lamellata* (Daday, 1893)		+			
61	***Lecane luna*** (O.F. Müller, 1776) (Syn.: *Lecane jobloti* (Bory de St. Vincent, 1827); *L. emarginata* (Eichwald, 1847); *L. luna balatonica* (Varga, 1945); *L. submagna* (De Ridder, 1960); *L. dorsicalis* (Arora, 1965))					+
62	*Lecane lunaris* (Ehrenberg, 1832) (Syn.: *Lecane quennerstedti* (Bergendal, 1892); *L. constricta* (Murray, 1913); *L. acus* (Harring, 1913); *L. crenata* (Harring, 1913); *L. sylvatica* (Harring, 1913); *L. virga* (Harring, 1914); *L. perplexa* (Ahlstorm, 1938); *L. scutata* (Pejler, 1962))		+			
63	*Lepadella* sp.			+		
64	*Monommata* sp.			+		
65	*Mytilina mucronata* (O.F. Müller, 1773)		+			
66	*Notholca* sp.				+	+
67	***Notholca caudata*** Carlin, 1943 (Syn.: *Notholca acuminata* Skorikov, 1905)			+		

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
68	***Notholca acuminata*** (Ehrenberg, 1832) (Syn.: *Anuraea acuminata* Ehrenberg, 1832; *Anuraea striata* Ehrenberg, 1838)	+	+			+
69	*Notholca acuminata extensa* Oloffsson, 1918		+			
70	*Notholca acuminata marina* Focke, 1961		+			
71	*Notholca labis* Gosse, 1887					+
72	***Notholca squamula*** (O.F. Müller, 1786) (Syn.: *Brachionus squamula* (Müller, 1786); *Anuraea striata* (Ehrenberg, 1838); *Notholca scapha* (Gosse, 1886); *N. polygona* (Gosse, 1887); *N. jugosa* (Gosse, 1887))					+
73	*Notholca squamula salina* Focke, 1961		+			
74	*Notholca striata* (O.F. Müller, 1786)	+	+	+		
75	*Philodina* sp.			+		
76	***Ploesoma truncatum*** (Levander, 1894) (Syn.: *Gastroschiza truncata* (Levander, 1894))			+		+
77	*Polyarthra* sp.				+	+
78	***Polyarthra dolichoptera*** Idelson, 1925 (Syn. *Polyarthra platyptera* var. *dolichoptera* Idelson, 1925)	+	+	+	+	+
79	*Polyarthra major* Burckhardt, 1900			+		
80	*Polyarthra remata* Skorikov, 1896			+		
81	***Polyarthra vulgaris*** Carlin, 1943		+	+		+
82	*Pompholyx sulcata* Hudson, 1885			+		
83	*Proales* sp.		+			+
84	*Proales reinhardti* (Ehrenberg, 1834)			+		
85	***Synchaeta*** sp.	+		+	+	+
86	*Synchaeta baltica* Ehrenberg, 1834	+	+	+	+	+
87	*Synchaeta cecilia* Rousselet, 1902		+	+		
88	*Synchaeta curvata* Lie-Pettersen, 1905	+	+	+		+
89	*Synchaeta fennica* Rousselet, 1909	+	+		+	+
90	*Synchaeta grimpei* Remane, 1929		+			
91	*Synchaeta gyrina* Hood, 1887	+				+
92	*Synchaeta littoralis* Rousselet, 1902		+	+		+
93	*Synchaeta monopus* Plate, 1889	+	+	+	+	+
94	*Synchaeta pectinata* Ehrenberg, 1832		+			
95	*Synchaeta triophthalma* Lauterborn, 1894	+	+			
96	*Synchaeta vorax* Rousselet, 1902					+
97	*Testudinella clypeata* (O.F. Müller, 1786)		+			+
98	*Trichocerca* sp.		+	+	+	
99	***Trichocerca capucina*** (Wierzejski et Zacharias, 1893)			+		+
100	*Trichocerca dixon-nutalli* Jennings, 1903		+			
101	*Trichocerca marina* (Daday, 1890)		+	+		+

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
102	***Trichocerca pusilla*** (Lauterborn, 1898)					+
103	*Trichocerca (Diurella) similis* (Wierzejski, 1893)			+		
104	*Trichotria pocillum* (O.F. Müller, 1776)					+
	Cladocera (Plates 5.3.6 – 5.3.10)					
105	*Alona* sp.			+		
106	*Alona intermedia* G.O. Sars, 1862		+			
107	***Alona quadrangularis*** (O.F. Müller, 1776)				+	+
108	*Alona rectangula* Sars, 1861 (Syn.: *Alona rectangulata* Sars, 1861)		+			
109	*Alonopsis elongata* Sars, 1862				+	
110	***Bosmina crassicornis*** P.E. Müller, 1867					+
111	***Bosmina longirostris*** (O.F. Müller, 1776)	+	+	+	+	+
112	*Bythotrephes* sp.					+
113	***Cercopagis pengoi*** (Ostroumov, 1891)	+		+	+	+
114	*Ceriodaphnia* sp.			+	+	+
115	*Ceriodaphnia laticaudata* P.E. Müller, 1867		+			
116	***Ceriodaphnia pulchella*** G.O. Sars, 1862					+
117	***Ceriodaphnia quadrangula*** (O.F. Müller, 1785)					+
118	*Ceriodaphnia reticulata* (Jurine, 1820)		+			
119	***Chydorus sphaericus*** (O.F. Müller, 1785)		+	+	+	+
120	*Cornigerius maeoticus* Pengo, 1879					+
121	*Daphnia* sp.				+	+
122	***Daphnia cristata*** G.O. Sars, 1861			+		+
123	***Daphnia cucullata*** G.O. Sars, 1862	+		+	+	+
124	*Daphnia galeata* G.O. Sars, 1864		+			
125	***Daphnia longispina*** (O.F. Müller, 1785)		+		+	+
126	*Daphnia magna* Straus, 1820		+			
127	***Diaphanosoma brachyurum*** (Liévin, 1848)		+	+	+	+
128	*Diaphanosoma mongolianum* Ueno, 1938		+			
129	***Eubosmina coregoni*** Baird, 1857 (Syn.: *Bosmina coregoni* Baird, 1857; *B. coregoni coregoni* Baird, 1857)	+	+	+	+	+
130	***Eubosmina longispina*** (Leidig, 1860) (Syn.: *Bosmina longispina* Leidig, 1860; *Bosmina coregoni maritima* sensu Purasjoki, 1958)	+	+	+	+	+
131	***Eubosmina maritima*** (P.E. Muller, 1867) (Syn.: *Bosmina maritima* P.E. Müller, 1867)	+	+	+	+	+
132	*Eurycercus lamellatus* (O.F. Müller, 1776)		+			
133	*Evadne anonyx* G.O. Sars, 1897				+	+
134	***Evadne nordmanni*** Lovén, 1836	+	+	+	+	+
135	*Evadne spinifera* P.E. Müller, 1867		+			

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
136	***Leptodora kindtii*** (Focke, 1844) (Syn.: *Leptodora kindti* (Focke, 1844))			+	+	+
137	*Pleopsis polyphemoides* (Leuckart, 1859) (Syn.: *Podon polyphemoides* (Leuckart, 1859))	+	+	+	+	+
138	*Podon intermedius* Lilljeborg, 1853	+	+	+	+	+
139	***Podon leuckartii*** (G.O. Sars, 1862)	+	+	+	+	+
140	*Polyphemus pediculus* (Linnaeus, 1761)					+
141	*Sida crystallina* (O.F. Müller, 1776)					+
	Copepoda, Calanoida (Plates 5.3.11 – 5.3.22, 5.3.28)					
142	***Acartia bifilosa*** (Giesbrecht, 1881)	+	+	+	+	+
143	*Acartia clausi* Giesbrecht, 1889	+	+	+		+
144	***Acartia discaudata*** (Giesbrecht, 1882)	+	+		+	
145	***Acartia longiremis*** (Lilljeborg, 1853)	+	+	+	+	+
146	***Acartia tonsa*** Dana, 1849	+	+	+	+	+
147	*Calanus finmarchicus* (Gunner, 1765) (Syn.: *Calanus tonsus* Brady, 1883)	+	+		+	+
148	*Calanus hyperboreus* Krøyer, 1838		+			
149	*Candacia armata* (Boeck, 1872)		+			
150	*Centropages chierchiae* Giesbrecht, 1889		+			
151	***Centropages hamatus*** (Lilljeborg, 1853)	+	+	+	+	+
152	*Centropages typicus* Krøyer, 1849		+		+	
153	*Diaptomus* sp.				+	
154	*Eudiaptomus gracilis* (G.O. Sars, 1862)		+			+
155	***Eurytemora affinis*** (Poppe, 1880)	+	+	+	+	+
156	*Eurytemora hirundoides* (Nordquist, 1888)	+	+	+	+	+
157	*Eurytemora hirundo* Giesbrecht, 1881	+	+	+	+	+
158	*Eurytemora lacustris* (Poppe, 1887)				+	+
159	*Eurytemora velox* (Lilljeborg, 1853)		+			+
160	*Limnocalanus grimaldii* (De Guerne, 1886)	+	+	+	+	+
161	***Limnocalanus macrurus*** (G.O. Sars, 1863)	+	+	+	+	+
162	*Metridia lucens* Boeck, 1865		+			
163	*Microcalanus pusillus* G.O. Sars, 1903		+			
164	***Paracalanus parvus*** (Claus, 1863)	+	+		+	+
165	*Paraeuchaeta norvegica* (Boeck, 1872)	+	+			
166	*Pareucalanus attenuatus* (Dana, 1849) (Syn.: *Eucalanus attenuatus* (Dana, 1849))		+			
167	*Pseudocalanus acuspes* (Giesbrecht, 1881)	+	+			
168	***Pseudocalanus elongatus*** (Boeck, 1865) (Syn.: *P. minutus elongatus* Farran & Vervoort, 1951)	+	+	+	+	+
169	*Pseudocalanus minutus* (Krøyer, 1845)		+			
170	***Temora longicornis*** (O.F. Müller, 1785)	+	+	+	+	+

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
	Copepoda, Cyclopoida (Plates 5.3.23 – 5.3.27)					
171	*Acanthocyclops robustus* (G.O. Sars, 1863)		+			
172	*Acanthocyclops vernalis* (Fischer, 1853) (Syn.: *Acanthocyclops plattensis* Pennak and Ward, 1985)		+			
173	*Cyclops* sp.	+		+	+	+
174	*Cyclops strenuus* Fischer, 1851		+			
175	***Cyclops vicinus*** Uljanin, 1875		+			
176	*Cyclopina gracilis* Claus, 1863		+			
177	*Cyclopina kieferi* Schäfer, 1936		+			
178	*Cyclopina norvegica* Boeck, 1864		+			
179	*Diacyclops bicuspidatus* (Claus, 1857)		+			+
180	*Diacyclops bisetosus* (Rehberg, 1880)		+			
181	*Eucyclops graciloides* Lilljeborg, 1888		+			
182	*Eucyclops macrurus* (G.O. Sars, 1863)					+
183	***Eucyclops serrulatus*** (Fischer, 1851)		+			+
184	*Eucyclops speratus* (Lilljeborg, 1901)		+			
185	*Halicyclops affinis* (G.O. Sars, 1863)		+			
186	*Halicyclops magniceps* (Lilljeborg, 1853)		+			
187	*Halicyclops neglectus* Kiefer, 1935		+			
188	*Macrocyclops albidus* (Jurine, 1820)					+
189	***Megacyclops viridis*** (Jurine, 1820)		+	+		+
190	*Mesocyclops* sp.	+				
191	*Mesocyclops hyalinus* (Rehberg, 1880)		+			
192	***Mesocyclops leuckarti*** (Claus, 1857)		+	+		+
193	*Oithona atlantica* Farran, 1908		+		+	
194	***Oithona similis*** Claus, 1866	+	+		+	+
195	*Paracyclops* sp.	+				
196	***Thermocyclops oithonoides*** Sars, 1863		+	+		+
	Copepoda, Monstrilloida					
197	*Cymbasoma rigidum* Thompson, 1888		+			
198	*Cymbasoma thompsoni* (Giesbrecht, 1892)		+			
199	*Monstrilla helgolandica* (Claus, 1863)		+			
	Copepoda, Harpacticoida (Plate 5.3.28)					
200	*Canthocamptus staphylinus* (Jurine, 1820)		+			+
201	*Ectinosoma melaniceps* Boeck, 1865		+			
202	*Halectinosoma curticorne* (Boeck, 1872)	+	+			
203	**Harpacticoida** indet.	+	+	+	+	+
204	*Microsetella norvegica* (Boeck, 1865)		+			+

No	Taxa	BP[1]	WBS[2]	NBS[3]	SBS[4]	EBS[5]
	Pteropoda					
205	*Limacina retroversa* (Fleming, 1823) (Syn.: *Spiratella retroversa* Fleming, 1823)	+	+			
	Polychaeta (Plates 5.3.33, 5.3.34)					
206	***Bylgides sarsi*** (Kinberg in Malmgren, 1865) (Syn.: *Harmothoe (Antinoella) sarsi sarsi* (Kinberg, 1865); *Antinoella sarsi* (Kinberg in Malmgren, 1865); *Antinoe sarsi* Kinberg in Malmgren, 1865)	+	+	+		+
207	*Harmothoe imbricata* (Linnaeus, 1769) (Syn.: *Aphrodita imbricata* Linnaeus, 1767)		+			
208	*Harmothoe impar* (Johnston, 1839) (Syn.: *Polynoe impar* Johnston, 1839)		+			+
209	*Nephtys* sp.		+			+
210	*Nereis diversicolor* O.F. Müller, 1776		+	+		
211	***Pygospio elegans*** Claparède, 1863	+	+	+		+
212	*Tomopteris helgolandica*	+	+			
	Polychaeta, larvae	+	+	+	+	+
	Chaetognatha (Plate 5.3.30)					
213	***Parasagitta elegans*** (Verrill, 1873) (Syn.: *Sagitta elegans* Verrill, 1873)	+	+		+	+
214	***Parasagitta setosa*** (Mueller, 1847) (Syn. *Sagitta setosa* Mueller, 1847)	+	+			+
215	*Sagitta bipunctata* Quoy & Gaimard, 1828		+			
	Copelata (Plates 5.3.29)					
216	***Fritillaria borealis*** Lohmann, 1896	+	+	+	+	+
217	***Oikopleura dioica*** Fol, 1872	+	+		+	+
	Larvae of Bivalvia (Mollusca) (Plate 5.3.31)	+	+	+	+	+
	Larvae of Gastropoda (Mollusca) (Plate 5.3.31)	+	+	+	+	+
	Larvae of Cirripedia (Crustacea) (Plate 5.3.32)	+	+	+	+	+
	Larvae of Bryozoa (Plate 5.3.35)	+	+	+	+	+
	Larvae of Echinodermata (Plate 5.3.35)	+	+	+	+	

[1]**BP, Baltic Proper:** after Ackefors (1965, 1969), Ostenfeld (1931), Mankowski (1948b, 1950b, 1951, 1959), Siudzinski (1965), Mielck & Künne (1932-1935);
[2]**WBS, Western Baltic Sea** (Kieler Bight, Mecklenburg Bight): after Remane (1940), Gerlach (2000), Kube et al. (2007a, b);
[3]**NBS, Northern Baltic Sea** (Archipelago Sea, Bothnian Sea): after Vuorinen (pers. com.), Lindquist (1959), Ostenfeld (1931);
[4]**SBS, Southern Baltic Sea** (Gdansk Basin): after Mankowski (1948a, 1950a, b);
[5]**EBS, Eastern Baltic Sea** (Gulf of Riga; Gulf of Finland, *excluding* the freshwater Neva Bay): after Purasjoki (1958), Flinkman (pers. com.), Telesh (pers. com), Silina (1997), Rodionova et al. (2005), Rodionova & Panov (2006).

(*) Synonyms

5.3. Photo plates: meso- and macrozooplankton of the open Baltic Sea

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Plate 5.3.1

Rotifera. 1, *Asplanchna priodonta*, live female, lateral view; **2,** *A. priodonta*, preserved female, head withdrawn into body, with resting egg; **3, 4,** *A. priodonta*, live female, lateral view with a newborn; **5,** *Brachionus calyciflorus amphiceros*, female, dorsal view, body length up to 300 µm; **6,** *Brachionus calyciflorus dorcas*, female, dorsal view; **7,** *Brachionus calyciflorus spinosus,* female, dorsal view; **8**, *Brachionus calyciflorus calyciflorus*, female, dorso-lateral view with extended foot; **9**, *Trichocerca pusilla*, contracted female, lateral view, body length 70-115 µm; **10**, *Trichocerca capucina*, female, lateral view, body length 240-300 µm (after Telesh & Heerkloss, 2002).

Note: Numbers below/near the scale bars indicate length of scale bars in micrometers.

Plate 5.3.1

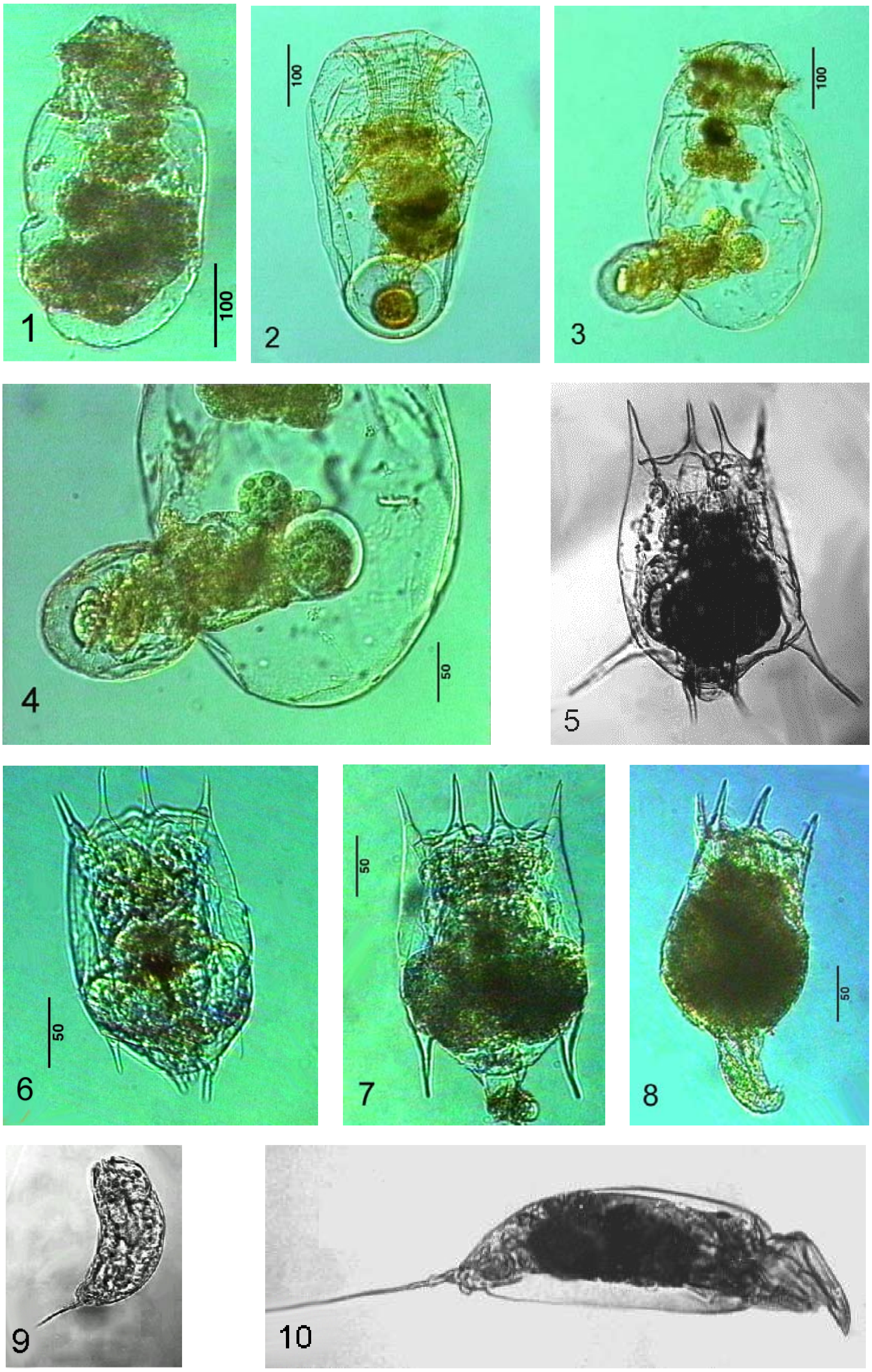


Plate 5.3.2

Rotifera. 1, *Brachionus angularis*, female with two eggs, dorsal view, body length 80-200 µm; **2**, *B. angularis*, female with one egg, dorsal view; **3**, *B. angularis*, female, ventral view; **4**, *Brachionus plicatilis*, female, ventral view; **5**, *Brachionus urceus*, female with egg, ventral view; **6**, *Euchlanis dilatata*, female, lateral view, body length up to 320 µm; **7**, *E. dilatata*, live female, dorsal view; **8**, *Conochilus unicornis*, semi-contracted female with fused ventral antenna (arrow); **9**, *C. unicornis*, female, lateral view with extended foot; **10**, *C. unicornis*, partly destroyed big colony of females with resting eggs (dark masses); **11**, *C. unicornis*, small colony; **12**, *Filinia longiseta*, dorsal view, head well seen at left side (after Telesh & Heerkloss, 2002).

Plate 5.3.2

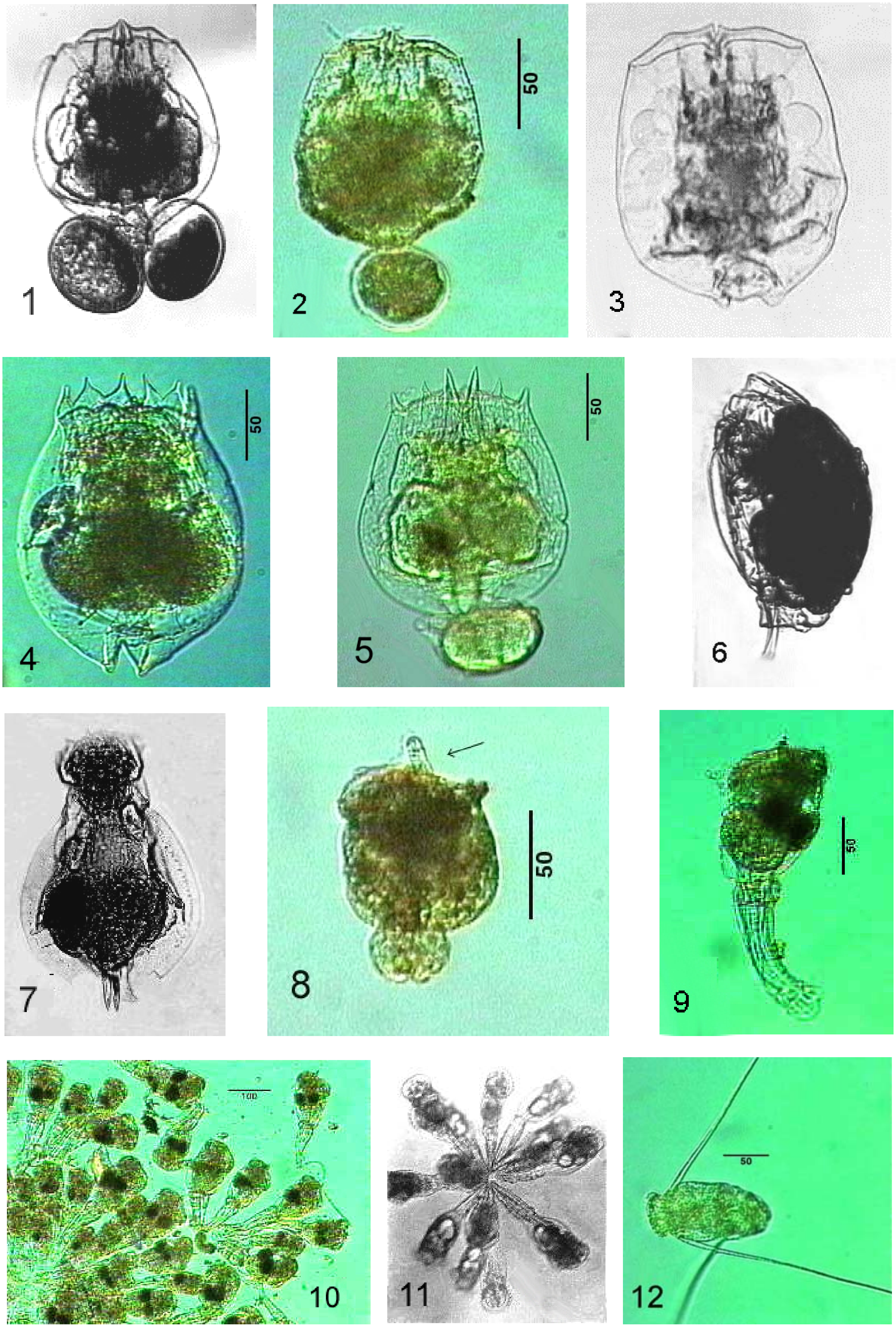


Plate 5.3.3

Rotifera. 1, 2, 3, *Filinia longiseta*, female, lateral view; **4**, *Kellicottia longispina*, female, lateral view, with egg; **5**, *K. longispina*, female, ventral view; **6**, *Keratella cochlearis typica*, female, lorica with long spine, dorsal view; **7**, *K. cochlearis typica*, female, lorica with short spine, dorsal view; **8**, **9**, *Keratella cochlearis baltica,* female, lateral view, with egg; **10**, *K. cochlearis baltica,* female, ventro-lateral view; **11**, **12**, **13**, *Keratella cruciformis eichwaldi*, female, dorsal view (after Telesh & Heerkloss, 2002).

Plate 5.3.3

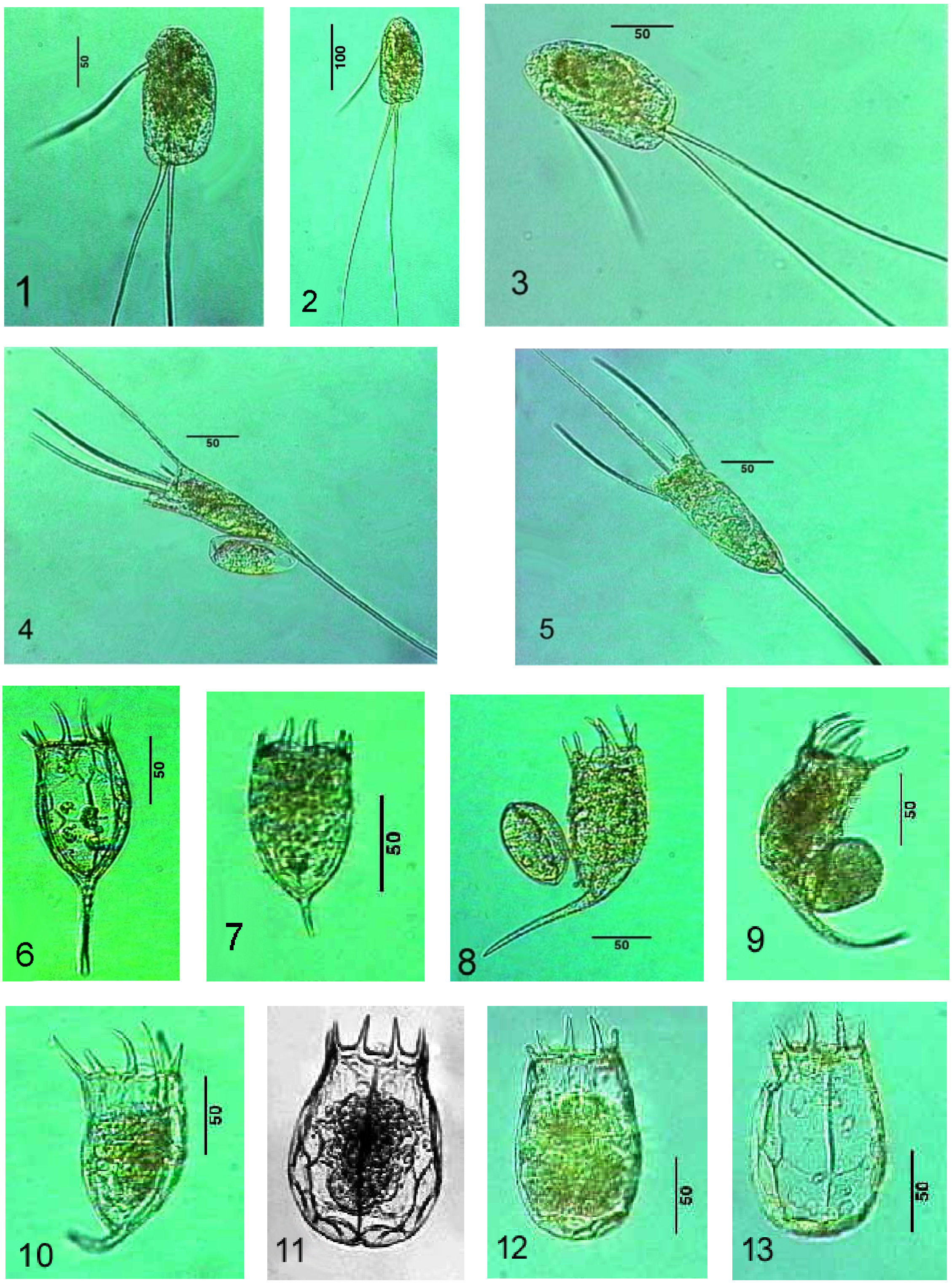


Plate 5.3.4

Rotifera. 1, *Keratella quadrata platei*, female, dorsal view, body length up to 350 µm (photo courtesy of P. Snoeijs); **2**, *K. quadrata platei*, female, dorsal view, corona well seen at the right side (photo courtesy of P. Snoeijs); **3**, *Keratella quadrata*, live female with egg, ventral view; **4**, *K. quadrata*, live female, dorsal view; **5**, *Synchaeta* sp., live female, semi-contracted, body length up to 6oo µm (photo H. Sandberg); **6**, *Notholca acuminata*, female, dorsal view; **7**, *Notholca caudata,* female, dorsal view; **8**, *Notholca squamula*, female, dorsal view, body length 120-190 µm; **9**, *Lecane luna*, female, ventral view; **10**, *Ploesoma truncatum*, female, dorso-lateral view; **11**, *Polyarthra vulgaris*, female, lateral view with ventral finlet (arrow); **12**, *Polyarthra dolichoptera*, female, ventral view with coronal antennae (arrows) (**3, 4, 6-12** after Telesh & Heerkloss, 2002).

Plate 5.3.4

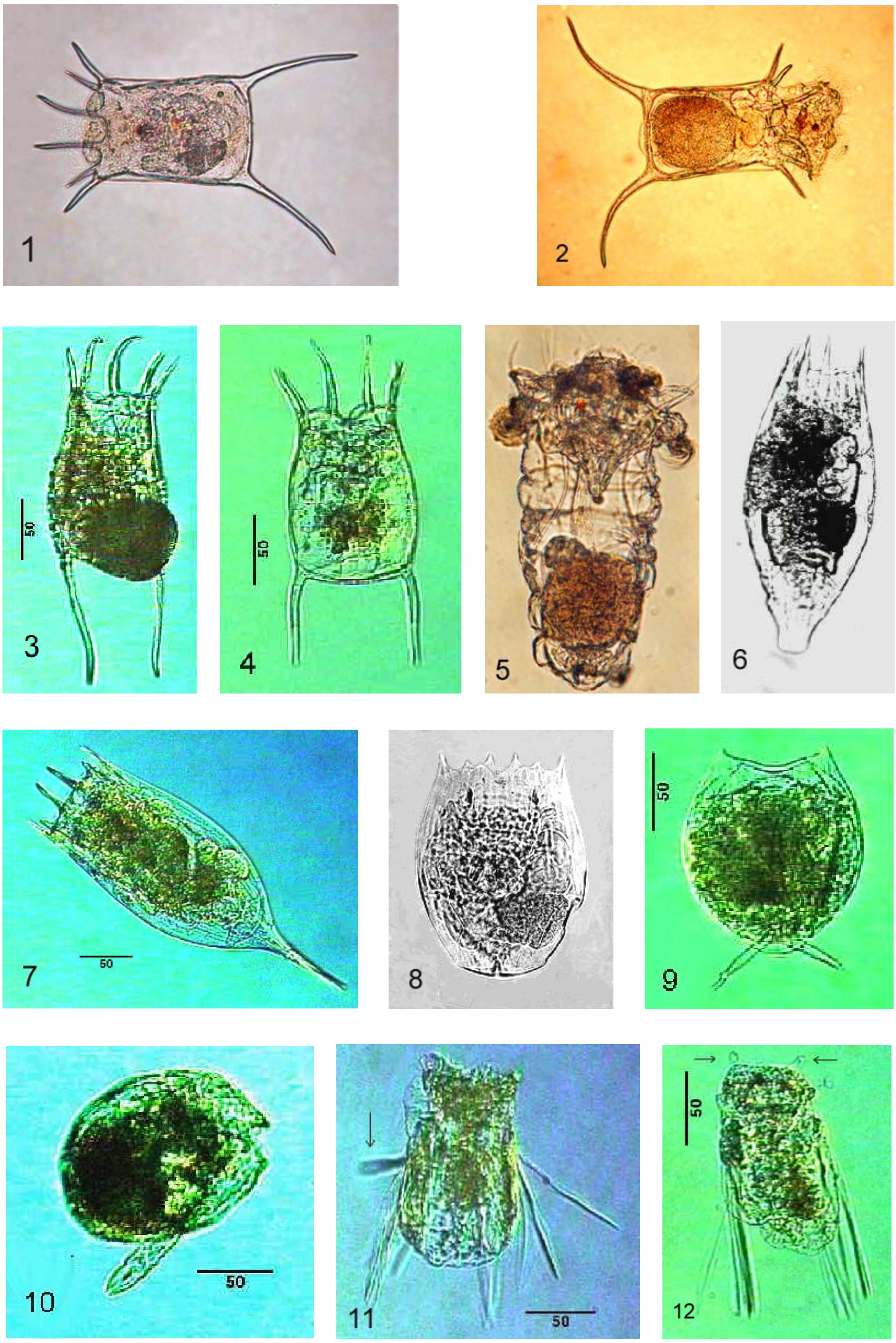


Plate 5.3.5

Rotifera. 1, 2, *Synchaeta* spp. (photo H. Sandberg); **3**, **4**, *Brachionus quadridentatus*, female with egg(s), different shape of lorica seen, length of lorica ca. 300 µm; **5**, *Filinia terminalis*, female, dorsal view, head withdrawn into body (after Telesh & Heerkloss, 2002).

Plate 5.3.5

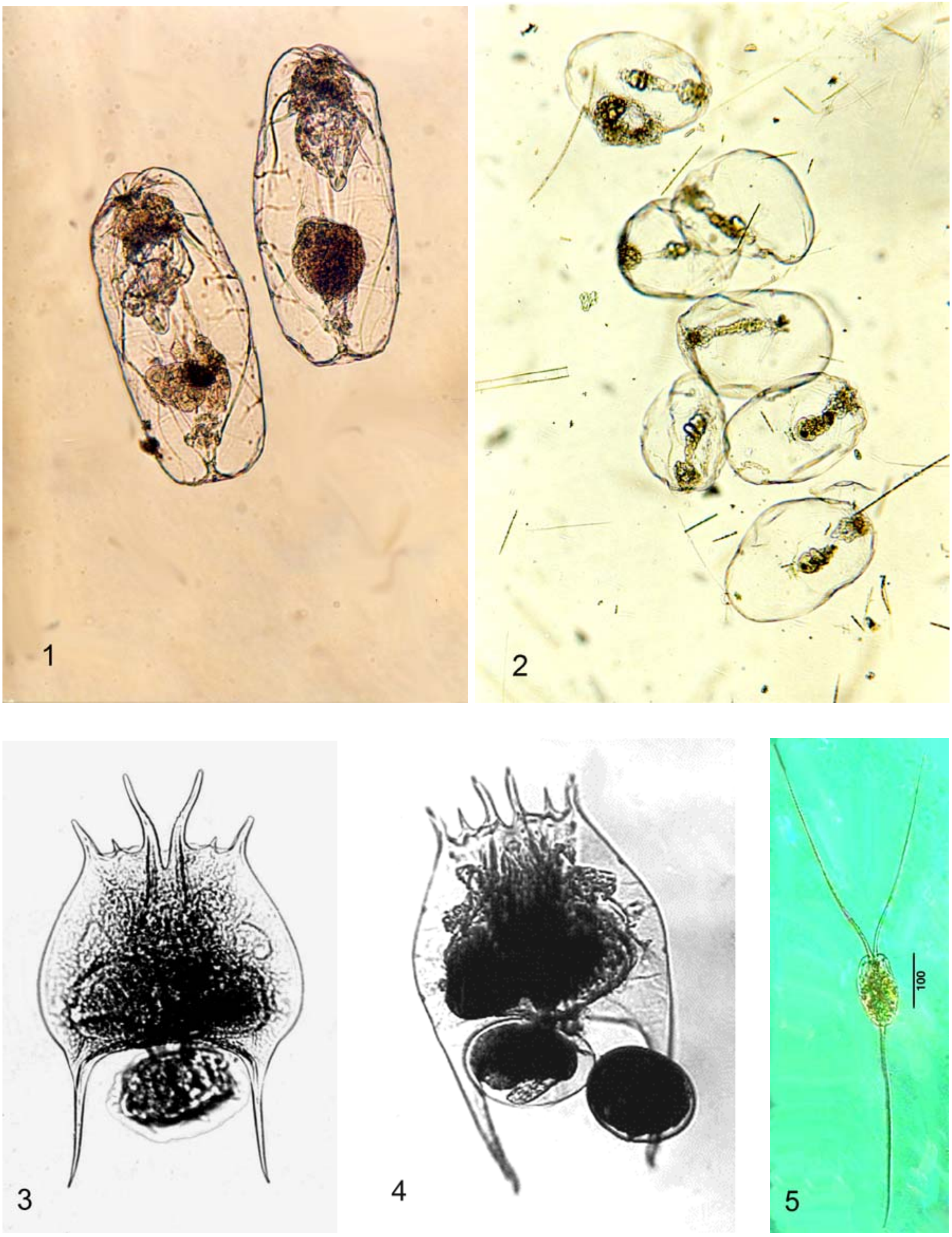


Plate 5.3.6

Cladocera. **1**, *Alona quadrangularis*, female, lateral view; **2**, Embryons of *Bosmina* sp., length ca. 180 µm; **3**, *Eubosmina maritima*, female with an embryo, lateral view, body length 250-620 µm (photo courtesy of H. Sandberg); **4**, *Eubosmina maritima*, female with eggs, lateral view (photo courtesy of H. Sandberg); **5**, *B. longirostris*, male, lateral view; **6**, *Bosmina longirostris curvirostris*, female with an embryo in the brood chamber, lateral view (after Telesh & Heerkloss, 2004).

Plate 5.3.6

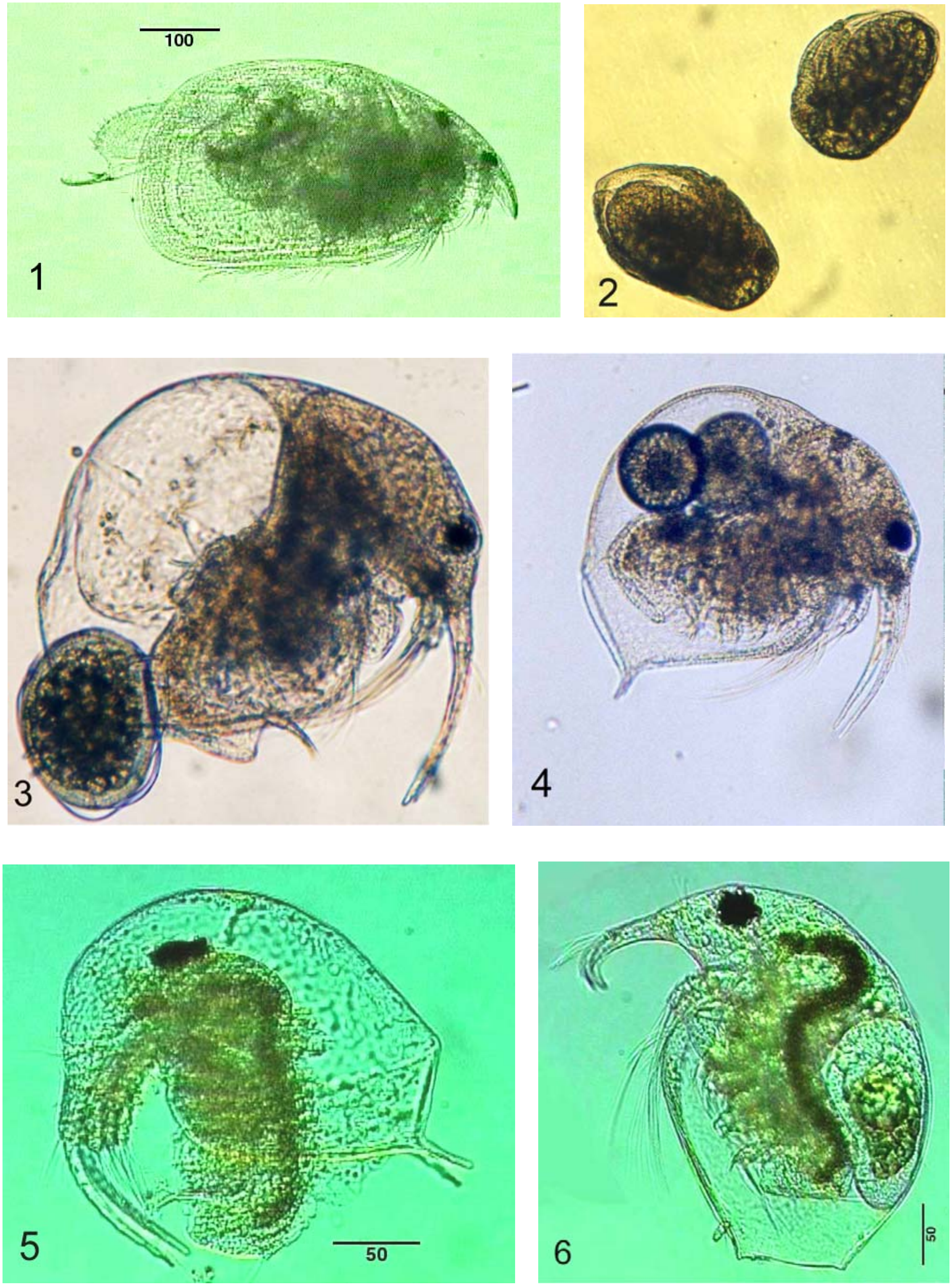


Plate 5.3.7

Cladocera. **1**, *Eubosmina coregoni gibbera*, female with embryos, lateral view; **2**, *Eubosmina coregoni thersites*, female with resting egg, lateral view; **3**, *Bosmina crassicornis*, female with eggs, lateral view; **4**, *Eubosmina longispina*, young female, lateral view; **5**, *E. longispina*, juvenile, lateral view; **6**, *E. longispina*, male, lateral view, body length 400-600 µm, photo courtesy of P. Snoeijs (**1-5** after Telesh & Heerkloss, 2004).

Plate 5.3.7

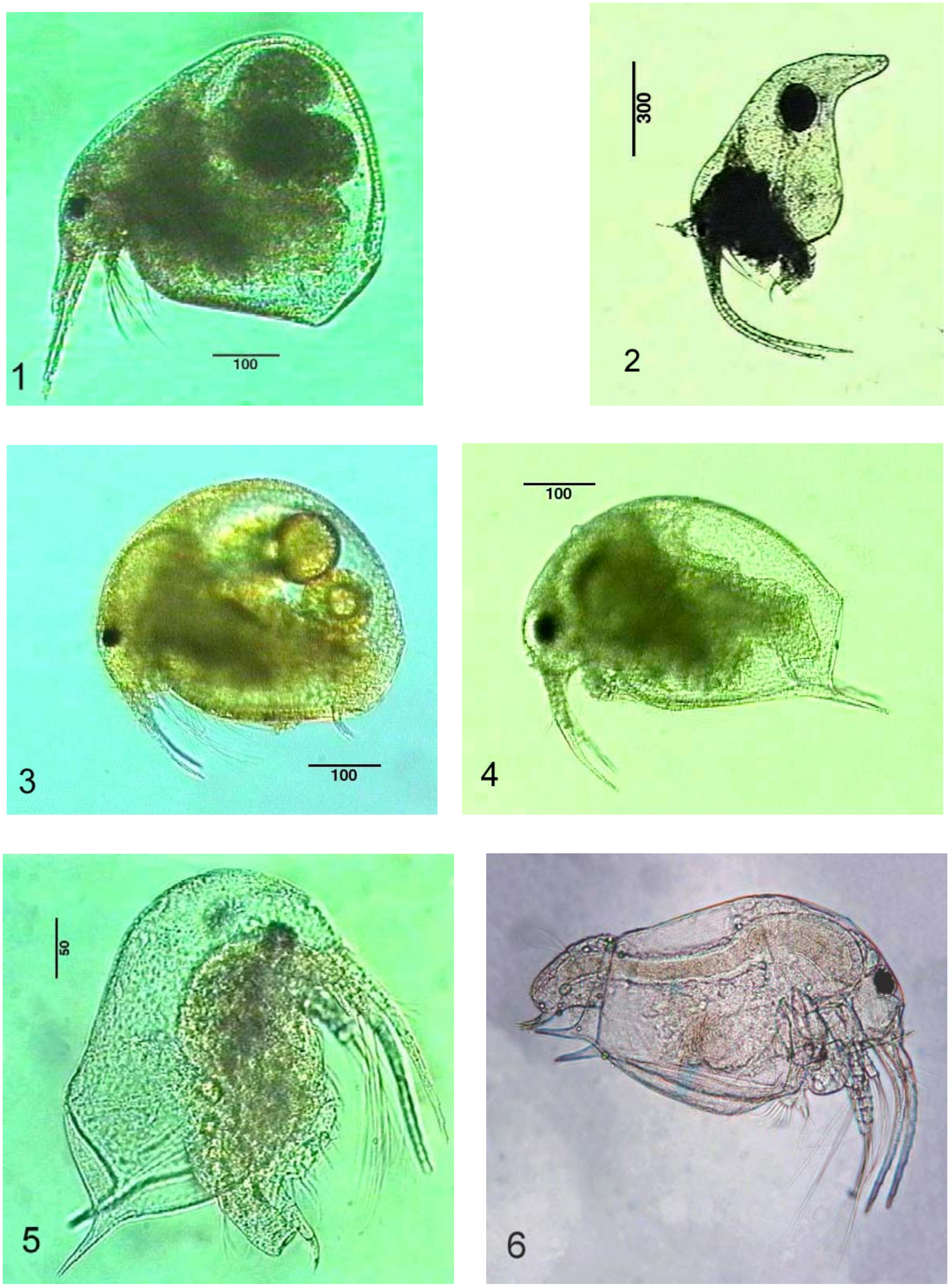


Plate 5.3.8

Cladocera. **1**, *Ceriodaphnia quadrangula*, female, lateral view; **2**, *Ceriodaphnia pulchella*, female with eggs, lateral view; **3**, *Chydorus sphaericus*, female with egg, lateral view with extended postabdomen; **4**, *Daphnia longispina*, female with embryos, lateral view; **5**, *Daphnia cristata*, female with egg, lateral view; **6**, **7**, *Daphnia cucullata*, females with eggs, lateral view, difference in helmet morphology is due to cyclomorphosis; **8**, *Daphnia cucullata procurva*, female, lateral view (after Telesh & Heerkloss, 2004).

Plate 5.3.8

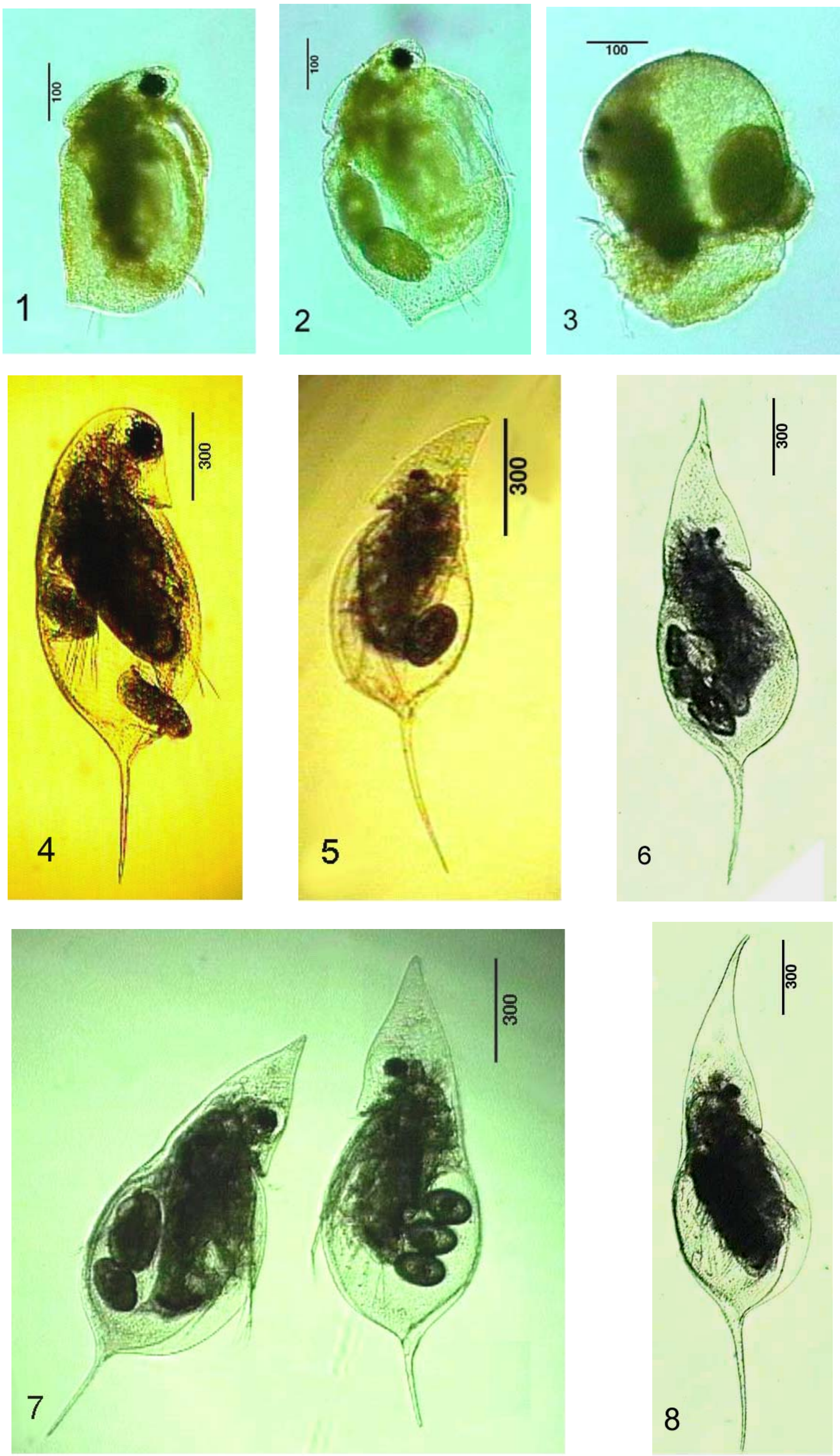


Plate 5.3.9

Cladocera. **1**, *Evadne nordmanni*, young female, lateral view (photo courtesy of P. Snoeijs); **2**, *E. nordmanni*, female with resting egg, lateral view, body length ca. 700 µm (photo H. Sandberg); **3**, *E. nordmanni*, female with embryos, lateral view; **4**, *E. nordmanni*, male, body length ca. 500 µm, lateral view (photo H. Sandberg); **5**, *Podon leuckartii*, female with eggs, lateral view; **6**, *Podon leuckartii*, female with resting egg, lateral view; **7**, *Leptodora kindtii*, female, ventral view; **8**, *L. kindtii*, female and an egg, lateral view (**3, 5-8** after Telesh & Heerkloss, 2004).

Plate 5.3.9

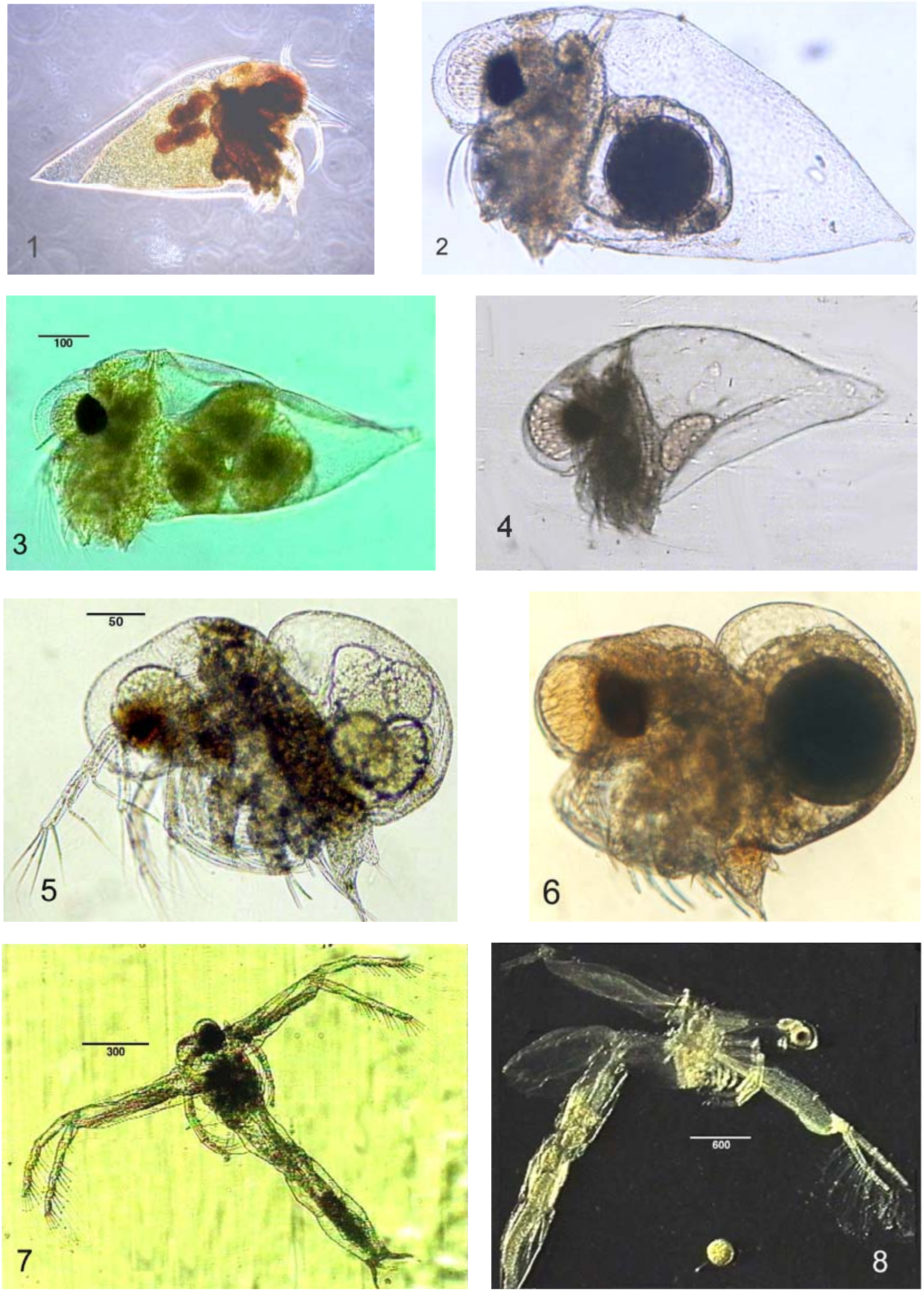


Plate 5.3.10

Cladocera. **1**, *Cercopagis pengoi*, general view of a female at stage II (with 2 claws) with resting egg, lateral view; **2**, *C. pengoi*, body of a female with embryos in brood chamber, lateral view; **3**, *C. pengoi*, body of a female with resting egg, lateral view; **4**, **5**, *Diaphanosoma brachyurum*, female, lateral view; **6**, *D. brachyurum*, male, dorso-lateral view (after Telesh & Heerkloss, 2004).

Plate 5.3.10

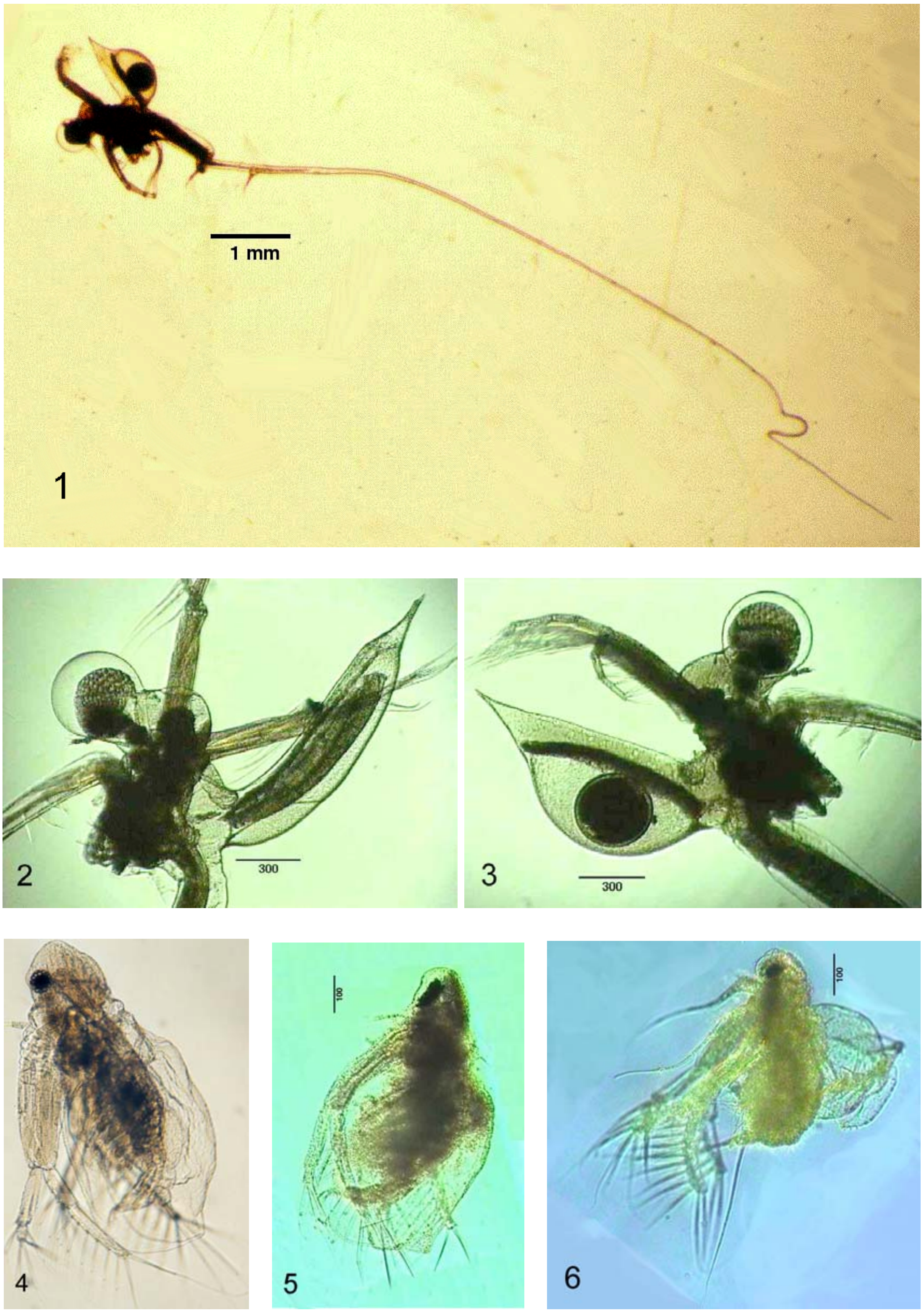


Plate 5.3.11

Copepoda. **1**, *Acartia tonsa*, female ventrally; **2**, *A. tonsa*, male ventrally; **3**, *A. tonsa*, female laterally, prosome* length ca. 620 µm, arrow shows P5; **4**, **5**, *A. tonsa*, male urosome; **6**, *A. tonsa*, P5 of female (after Telesh & Heerkloss, 2004; **3**, **5**, photos H. Sandberg).

* In calanoid copepods, prosome is considered as cephalothorax plus fifth thoracic segment (CPHT + TH5); in cyclopoid copepods prosome is equal to cephalothorax.

Plate 5.3.11

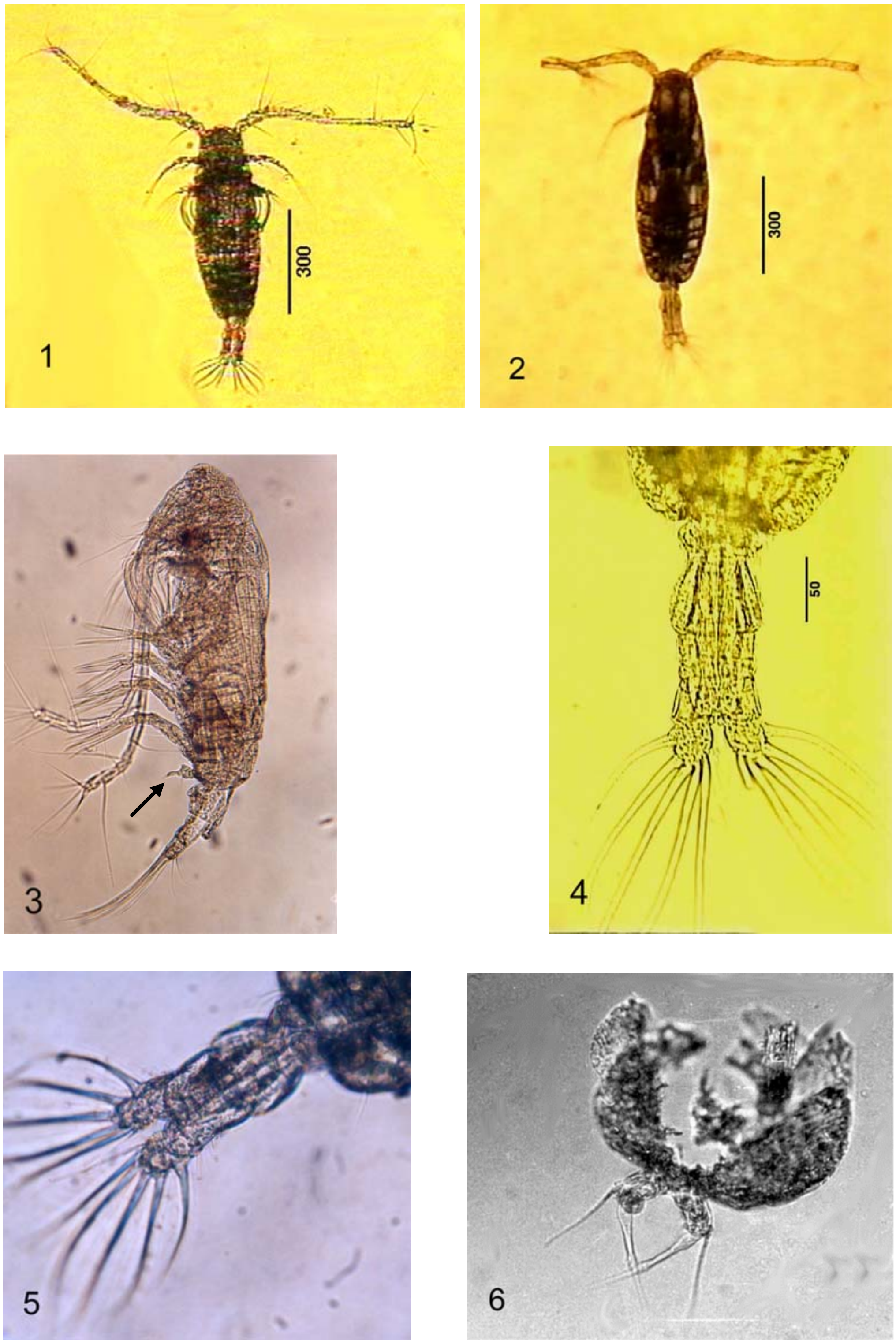


Plate 5.3.12

Copepoda. **1**, *Acartia tonsa*, abdomen and P5 of female, lateral view; **2**, *A. tonsa*, posterior part of male laterally, arrow shows P5; **3**, *Acartia longiremis,* copepodite C5, lateral view, prosome length ca. 520 µm; **4**, *A. longiremis,* copepodite C3, lateral view, prosome length ca. 400 µm; **5**, *A. longiremis,* copepodite C4, ventral view, prosome length ca. 460 µm; **6**, nauplius of *Acartia* sp., length ca. 180 µm (**1**, **2**, photos H. Sandberg; **3-6**, photo courtesy of P. Snoeijs).

Plate 5.3.12

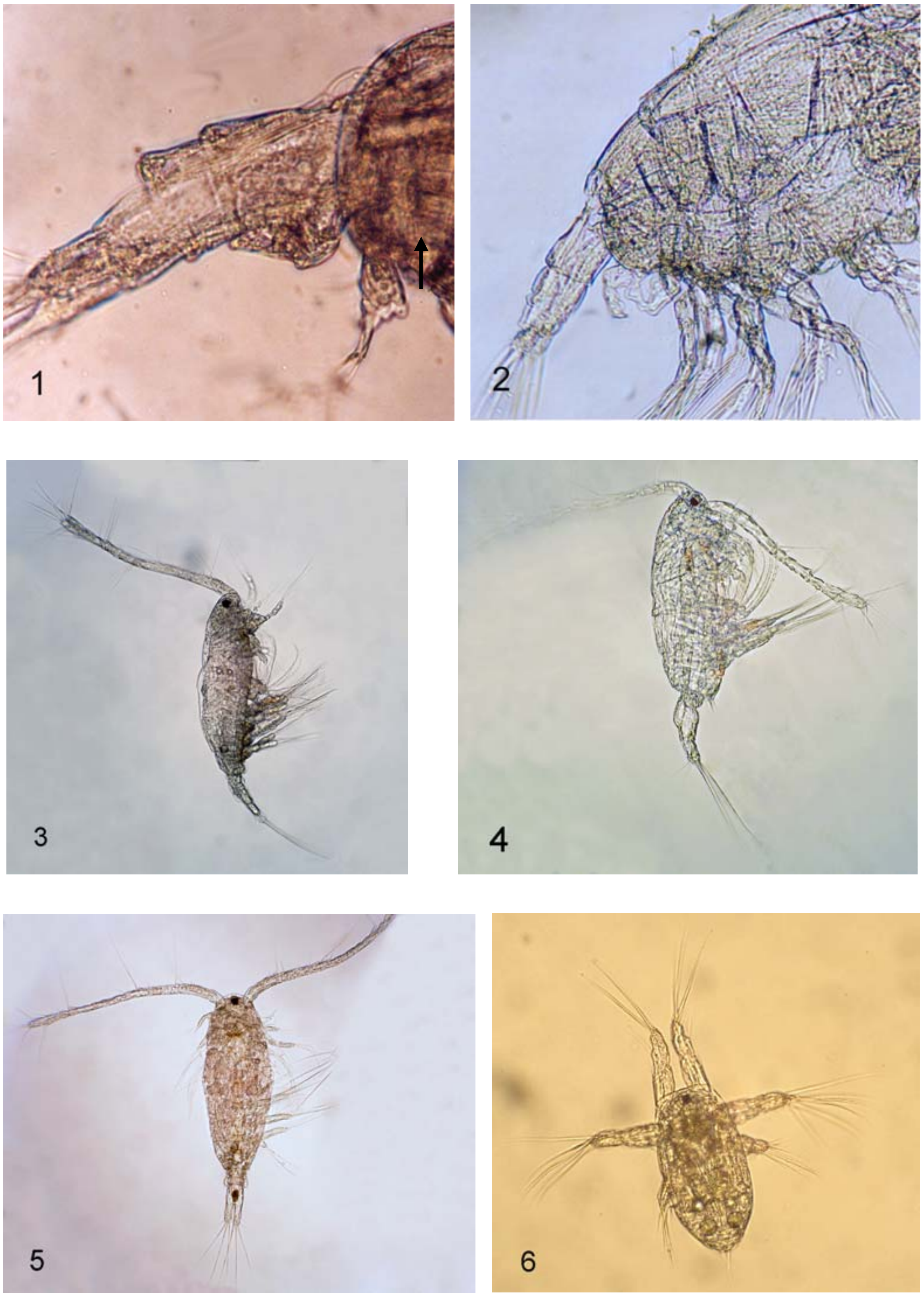


Plate 5.3.13

Copepoda. 1, *Acartia longiremis,* female, lateral view, prosome length ca. 650 µm; **2**, *A. longiremis,* copepodite C5, lateral view, prosome length ca. 520 µm, arrow shows characteristically long setae on the antennae; **3**, *A. longiremis,* female with epibionts (arrow), ventral view; **4**, *A. longiremis,* copepodite C5, lateral view; **5**, *A. longiremis,* copepodite C4, ventral view, prosome length ca. 460 µm; **6**, *A. longiremis,* copepodite C3, ventral view, prosome length ca. 400 µm (photo courtesy of P. Snoeijs).

Plate 5.3.13

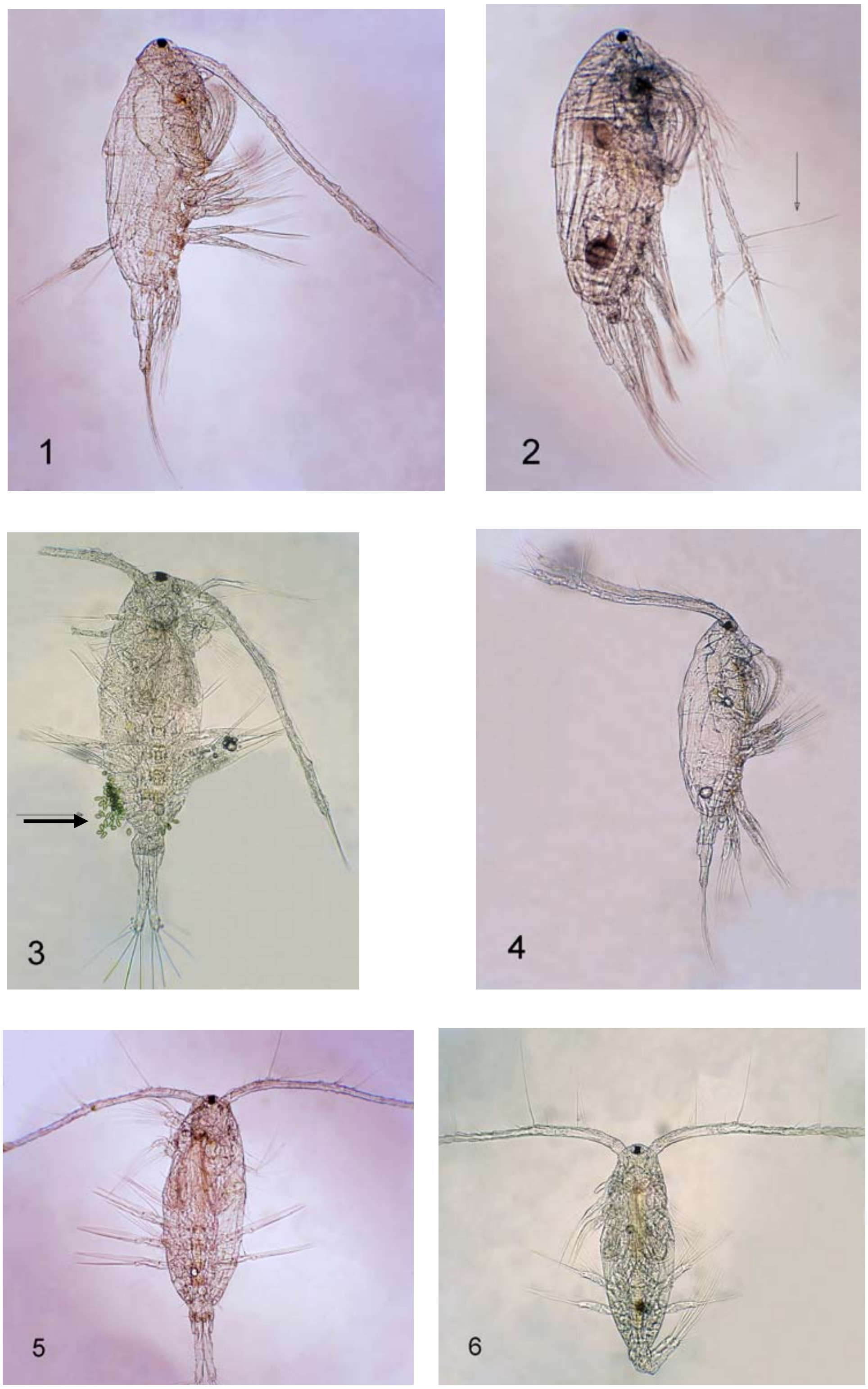


Plate 5.3.14

Copepoda. 1, *Acartia longiremis,* female, ventral view, prosome length ca. 650 µm; **2**, *A. longiremis,* two females, arrow shows a characteristic spine; **3**, *A. longiremis,* urosome of female; **4**, *A. longiremis,* female, short arrow shows a characteristic spine, long arrow shows P5; **5**, *A. longiremis,* male, ventral view, prosome length ca. 600 µm, arrow shows a characteristic spine; **6**, *Acartia* sp., nauplii, body length ca. 200 µm (photos H. Sandberg).

Plate 5.3.14

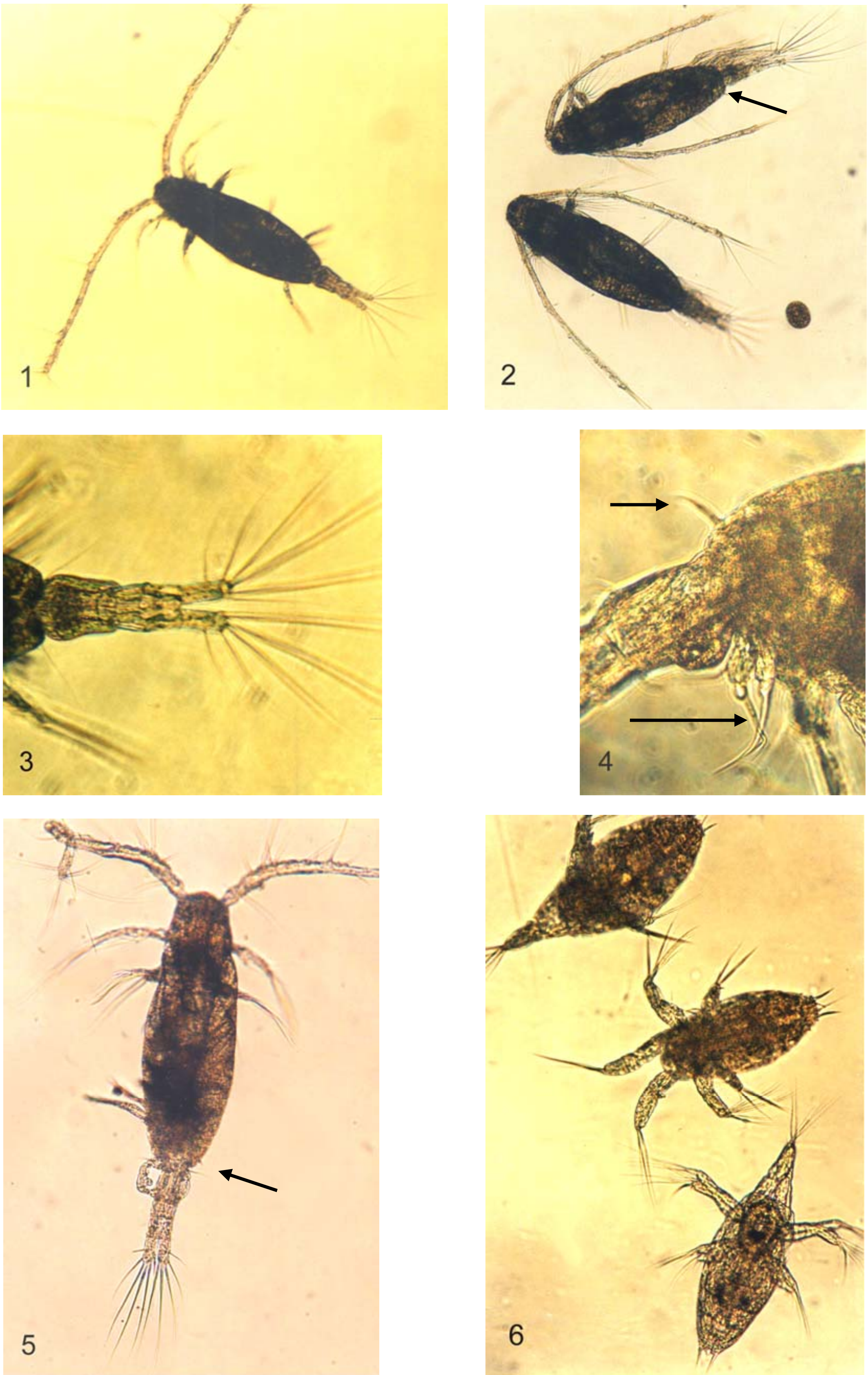


Plate 5.3.15

Copepoda. 1, *Acartia bifilosa*, male, ventral view, total length ca. 1000 µm, prosome length 775.8 ± 10.5 µm (Postel et al., 2007); **2**, *A. bifilosa*, female P5, prosome length of a female 785.5 ± 11.9 µm; **3**, *A. bifilosa*, male, ventro-lateral view; **4**, *Acartia discaudata*, female ventrally, prosome length 759 ± 14 µm (Conover, 1959); **5**, *A. discaudata*, female, lateral view; **6**, *A. discaudata*, male, ventral view, total length ca. 900 µm (photos H. Sandberg).

Plate 5.3.15

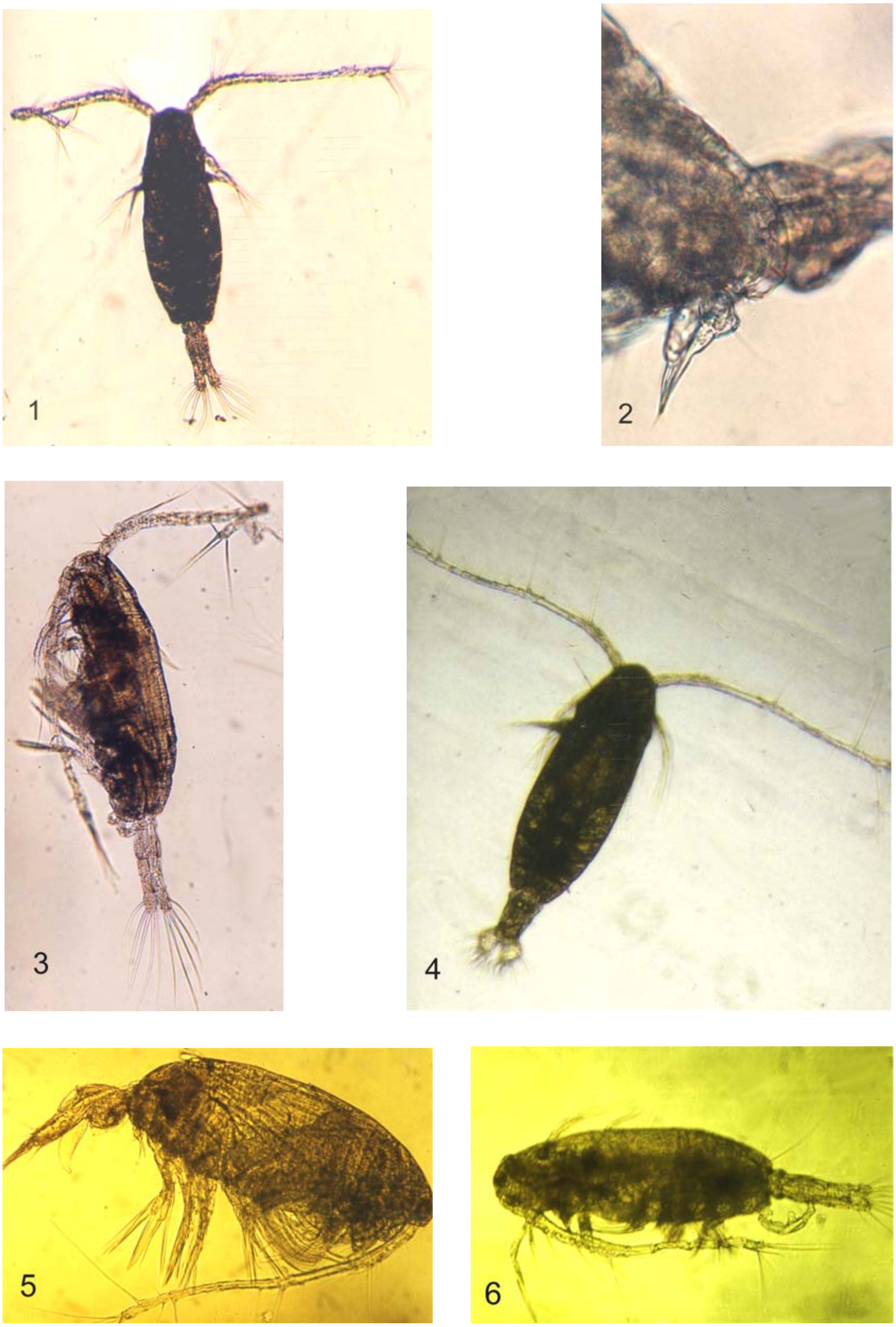


Plate 5.3.16

Copepoda. 1, *Centropages hamatus*, male, dorsal view, total length ca. 1400 µm, CPHT length 802.8 ± 8.6 µm (Postel et al., 2007); **2**, *C. hamatus*, abdomen of female, with spermatophore (arrow); **3**, *C. hamatus*, abdomen of male, P5 seen at left side (arrow); **4**, *C. hamatus*, copepodite C4, lateral view, length of prosome 655.8 ± 12.1 µm (Postel et al., 2007); **5**, **6**, *C. hamatus*, copepodite C2, ventral view, length of prosome 478.4 ± 16.4 µm (Postel et al., 2007) (**1**-**3**, photos H. Sandberg; **4-6**, photo courtesy of P. Snoeijs).

Plate 5.3.16

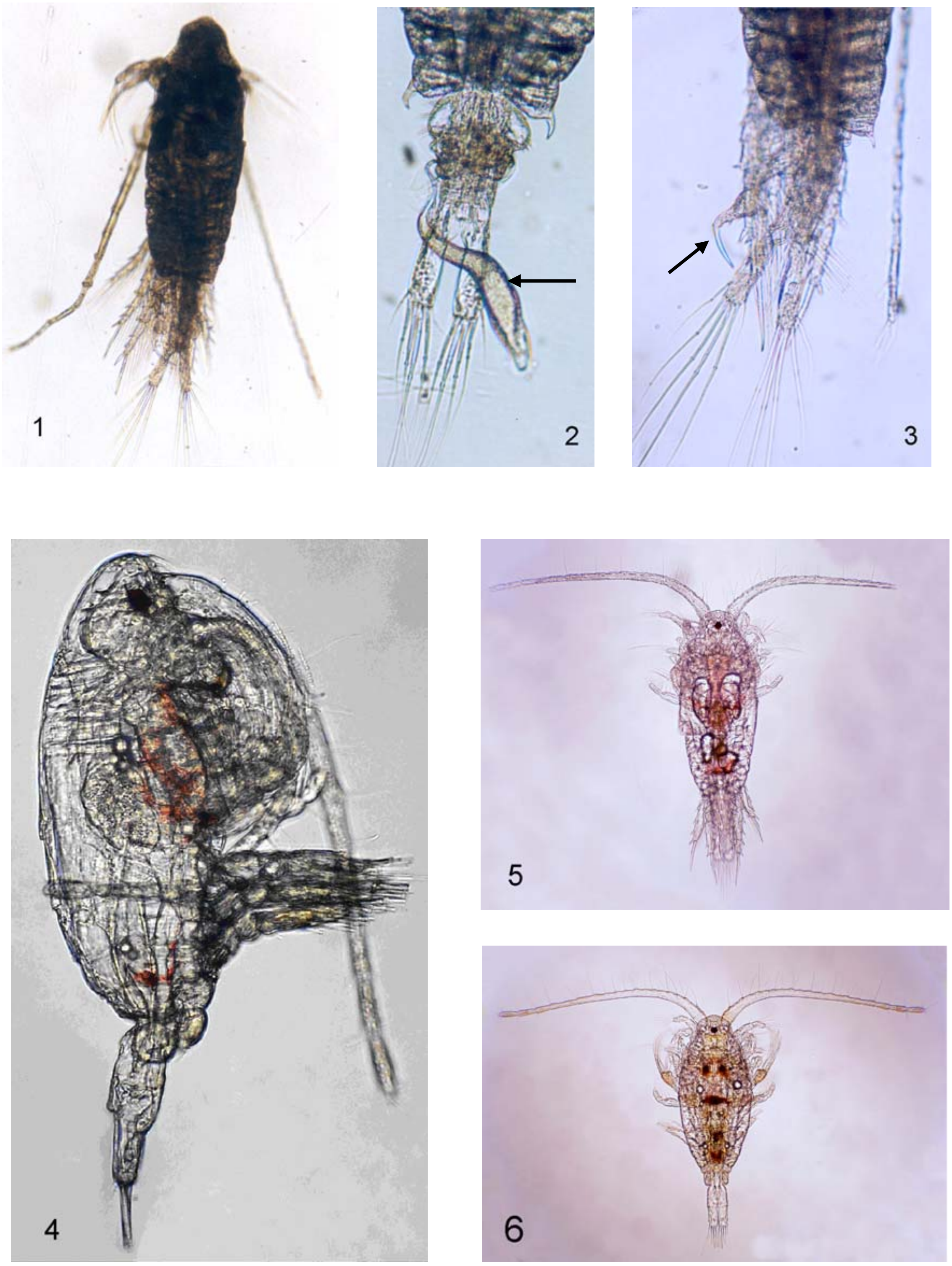


Plate 5.3.17

Copepoda. 1, *Eurytemora affinis*, female with egg sack, ventral view, prosome length ca. 650 µm; **2**, *E. affinis*, male, lateral view; **3**, *E. affinis*, nauplius N6 ventrally, body length 260 µm; **4**, *E. affinis*, male, articulation of the antenna; **5**, *E. affinis*, P5 of male; **6**, **7**, *E. affinis*, P5 of female; **8**, *E. affinis*, posterior end of female, with eggs; **9**, *E. affinis*, female with few loose eggs; **10**, *E. affinis*, male, lateral view, arrow shows the articulated antenna (after Telesh & Heerkloss, 2004; **1, 3, 4, 9**, photos H. Sandberg).

Plate 5.3.17

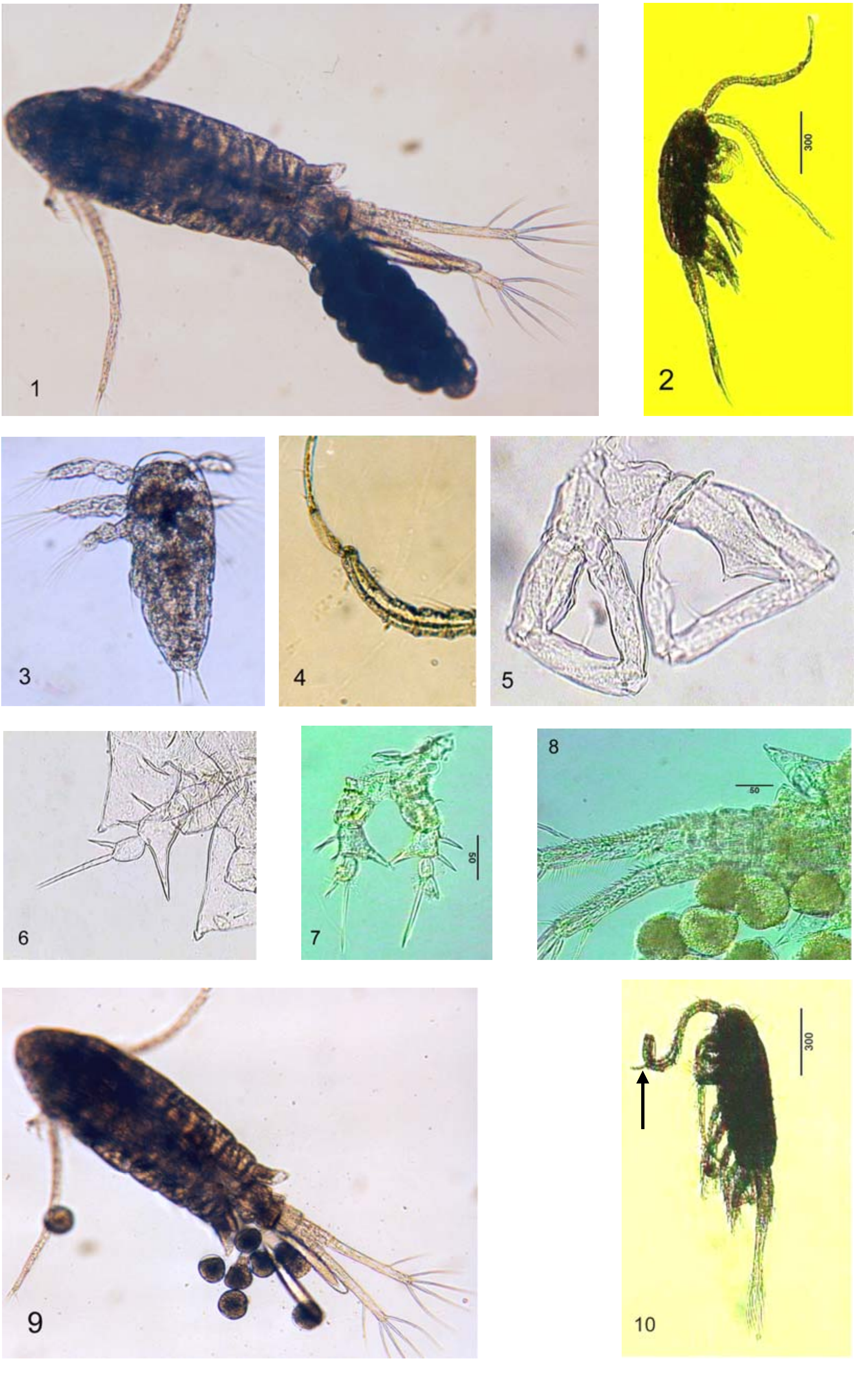


Plate 5.3.18

Copepoda. 1, *Eurytemora affinis*, male, lateral view, prosome length ca. 600 µm, arrow shows articulation of the antenna; **2**, *Limnocalanus macrurus*, male, dorso-lateral view, prosome length 1.7-1.8 mm (Hernroth, 1985), **3**, *L. macrurus*, female, lateral view, total length 2.4 – 2.9 mm, prosome length 1.7-1.9 mm (Czaika, 1982; Balcer et al., 1984; Hernroth, 1985) (photos H. Sandberg).

Plate 5.3.18

Plate 5.3.19

Copepoda. 1, 2, *Paracalanus parvus*, female, lateral, body length ca. 1000 µm, arrow shows P5; **3**, *P. parvus*, male, ventral view, prosome length 789.8 ± 10.4 µm (Postel et al., 2007); **4**, *P. parvus*, male, lateral view, arrow shows P5; **5**, *Pseudocalanus elongatus*, female, dorsal view, prosome length 887.0 ± 9.5 µm (Postel et al., 2007); **6**, *P. elongatus*, copepodite C3, lateral view, prosome length 573.5 ± 41.9 µm (Postel et al., 2007) (photos H. Sandberg).

Plate 5.3.19

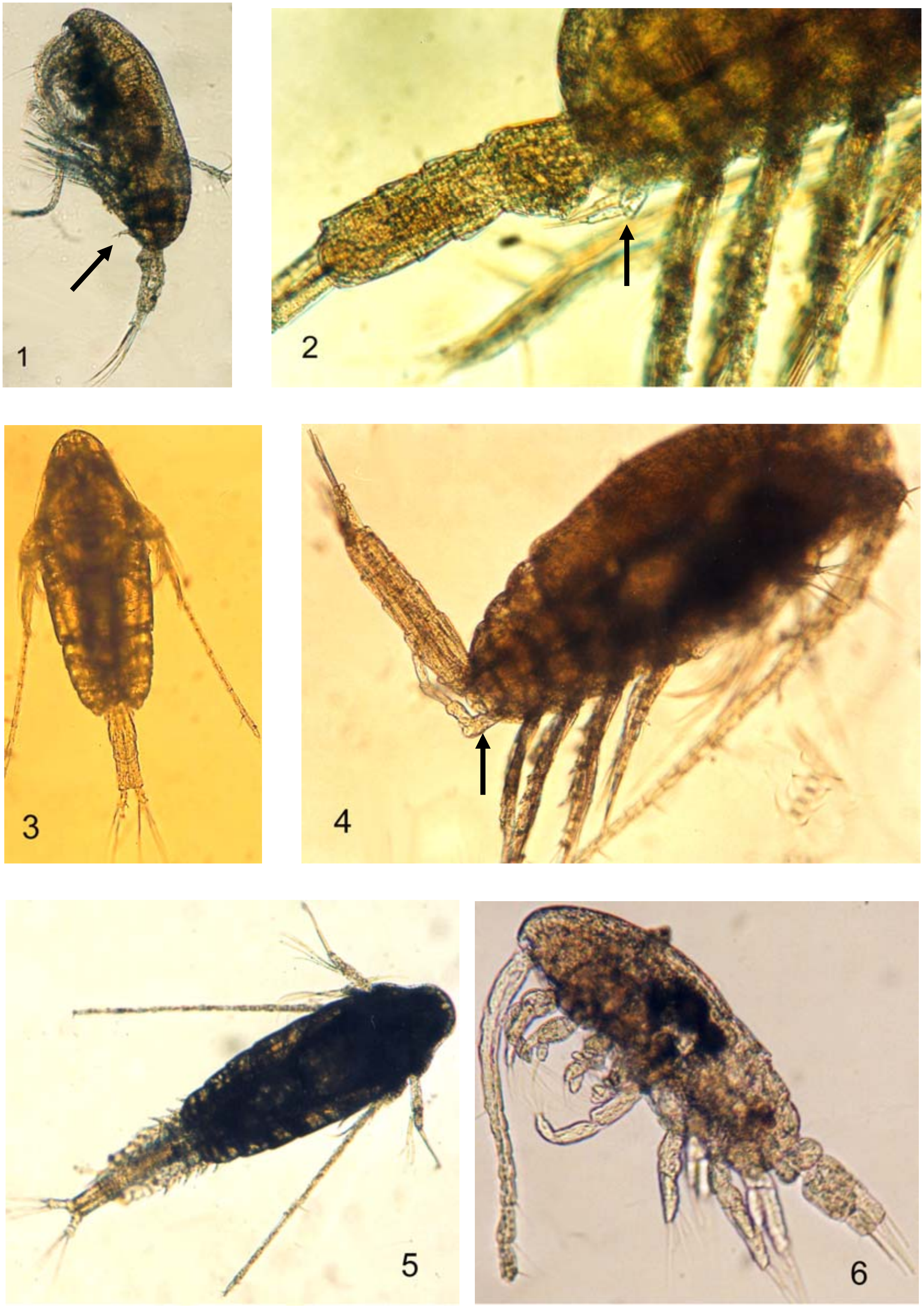


Plate 5.3.20

Copepoda. 1, *Pseudocalanus elongatus*, female, abdomen, lateral view, vertical arrow shows genital segment, horizontal arrow shows spermatophores; **2**, *P. elongatus*, P5 of male, lateral view; **3**, *P. elongatus*, copepodite C4, lateral view, length 716.9 ± 24.4 µm (Postel et al., 2007), red inclusions – lipids stocked for diapausing; **4**, *P. elongatus*, nauplius ventrally, length 306.9 ± 14.0 µm (Postel et al., 2007); **5**, *P. elongatus*, nauplii at different stages in the sample (**1, 2, 4, 5**, photos H. Sandberg; **3**, photo courtesy of P. Snoeijs).

Plate 5.3.20

Plate 5.3.21

Copepoda. 1, *Temora longicornis*, copepodite C3, lateral view, length 425.4 ± 9.6 µm (Postel et al., 2007); **2, 3**, *T. longicornis*, female, lateral view, prosome length 709.3 ± 6.7 µm (Postel et al., 2007); **4**, *T. longicornis*, male, lateral view, prosome length 690.8 ± 6.0 µm (Postel et al., 2007) (photo courtesy of P. Snoeijs).

Plate 5.3.21

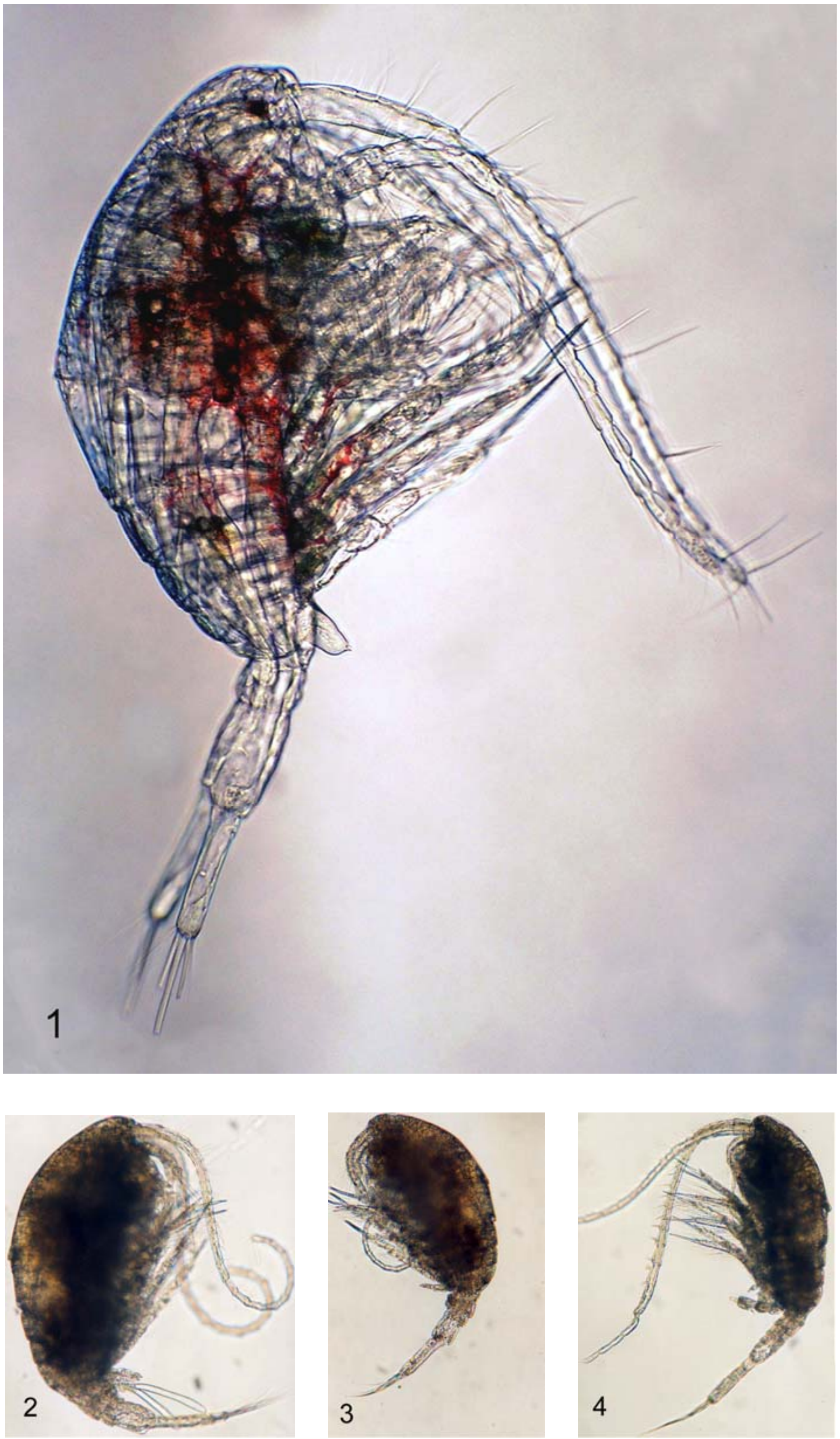


Plate 5.3.22

Copepoda. 1-6, *Temora longicornis*, copepodites at different stages: **1**, C4, prosome length 575.4 ± 8.0 µm (Postel et al., 2007); **2**, C3; **3-6**, C1 and C2 (photo courtesy of P. Snoeijs).

Plate 5.3.22

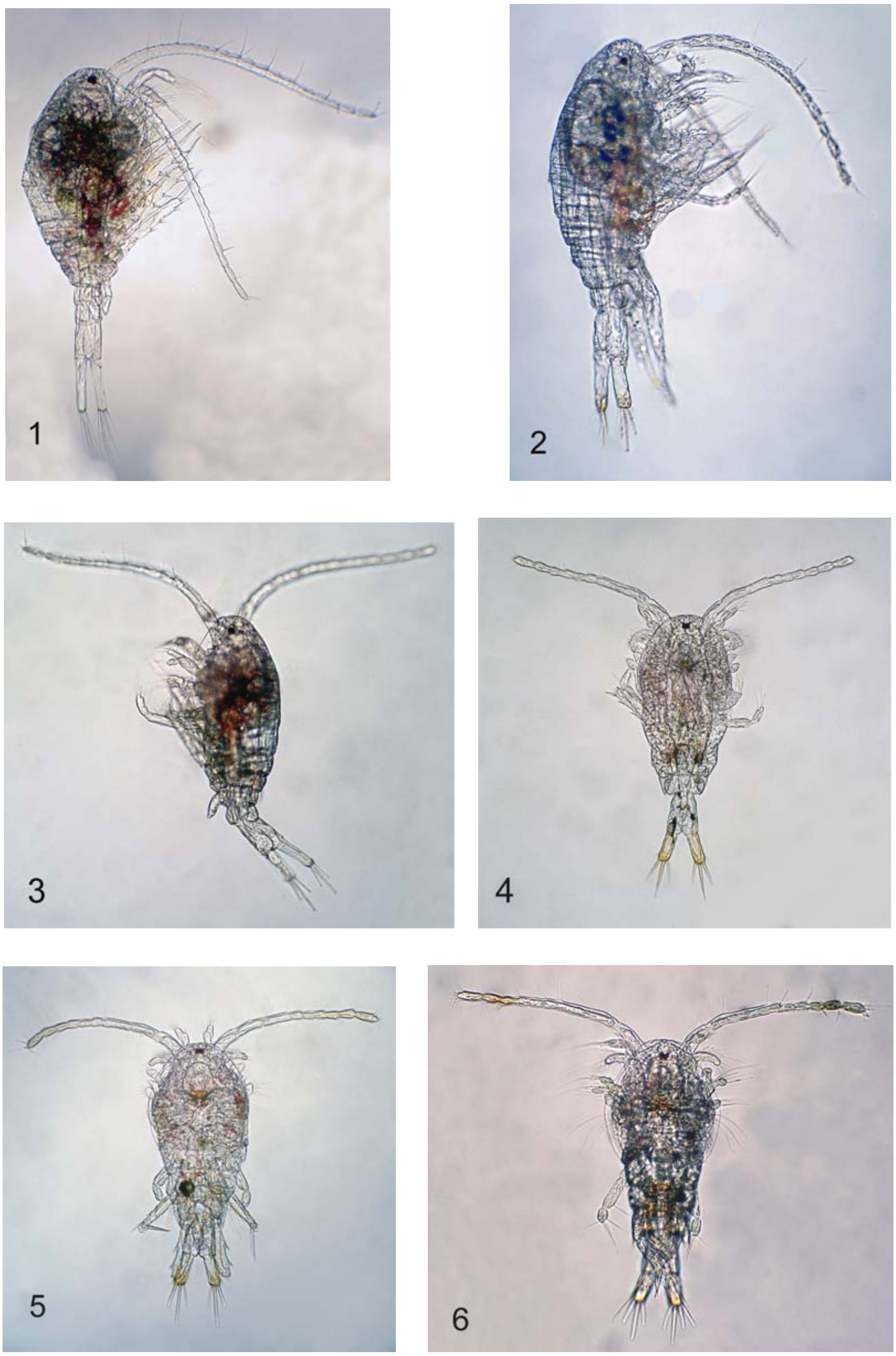


Plate 5.3.23

Copepoda. 1-3, *Cyclops vicinus*, female; **4**, *C. vicinus*, copepodite C4, CPHT length 550.4 ± 68.1 µm (Postel et al., 2007) (**1, 2, 4**, photos H. Sandberg; **3**, after Telesh & Heerkloss, 2004).

Plate 5.3.23

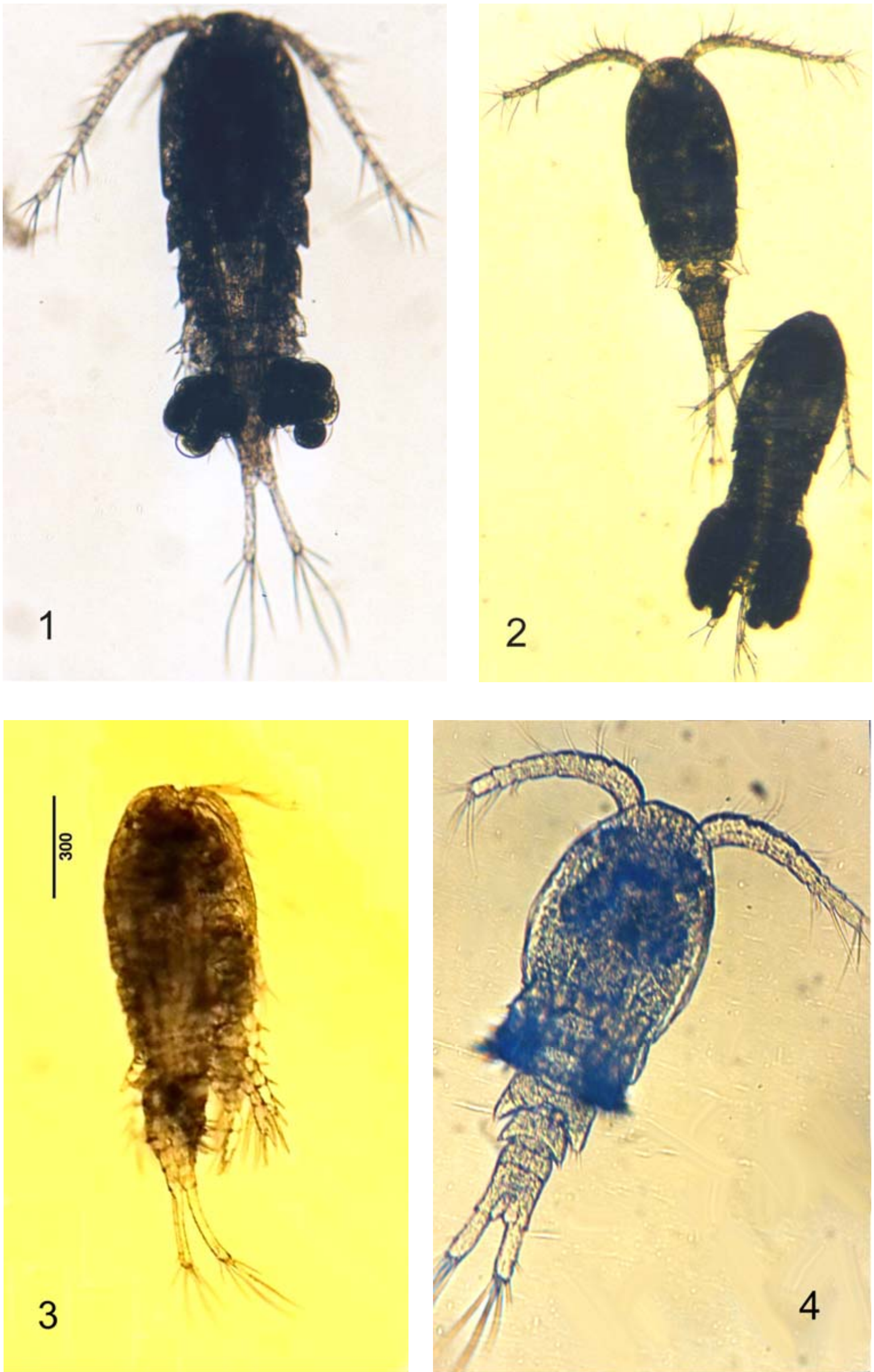


Plate 5.3.24

Copepoda. 1, *Cyclops vicinus*, head of a female, dorso-lateral view; **2**, *C. vicinus*, female A1; **3, 4**, *C. vicinus*, FU of female; **5**, *C. vicinus*, urosome of male; 6, *C. vicinus*, genital somite of male (**1,** photo H. Sandberg; **2-6**, after Telesh & Heerkloss, 2004).

Plate 5.3.24

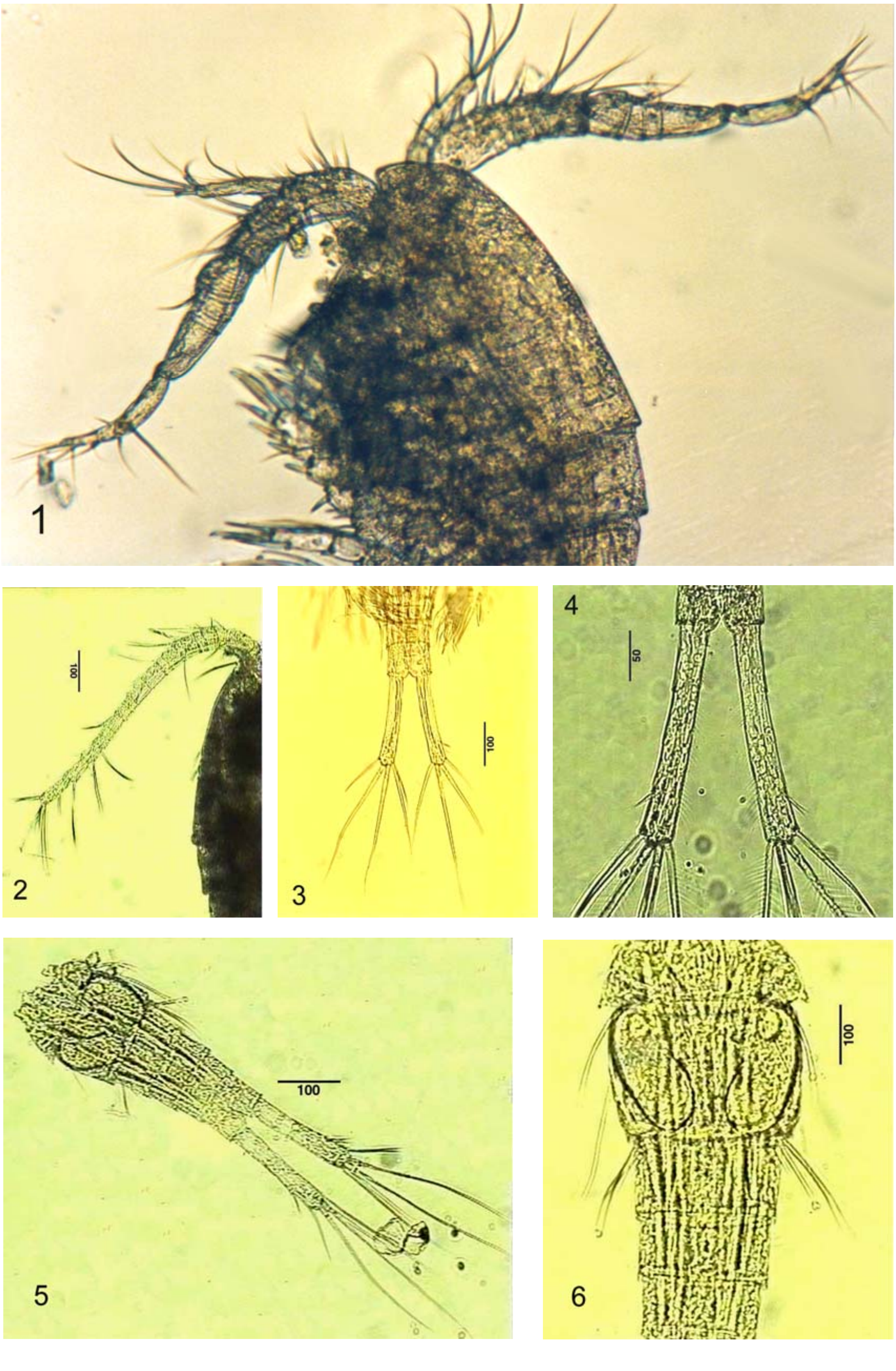


Plate 5.3.25

Copepoda. 1, *Eucyclops serrulatus*, female dorsally; **2**, *E. serrulatus*, female laterally; **3**, *E. serrulatus*, male ventrally; **4**, *E. serrulatus*, female FU and egg-sacs; **5**, *E. serrulatus*, male furca; **6-8**, *E. serrulatus*, male A1; **9**, P5 of *Eucyclops* sp. (after Telesh & Heerkloss, 2004).

Plate 5.3.25

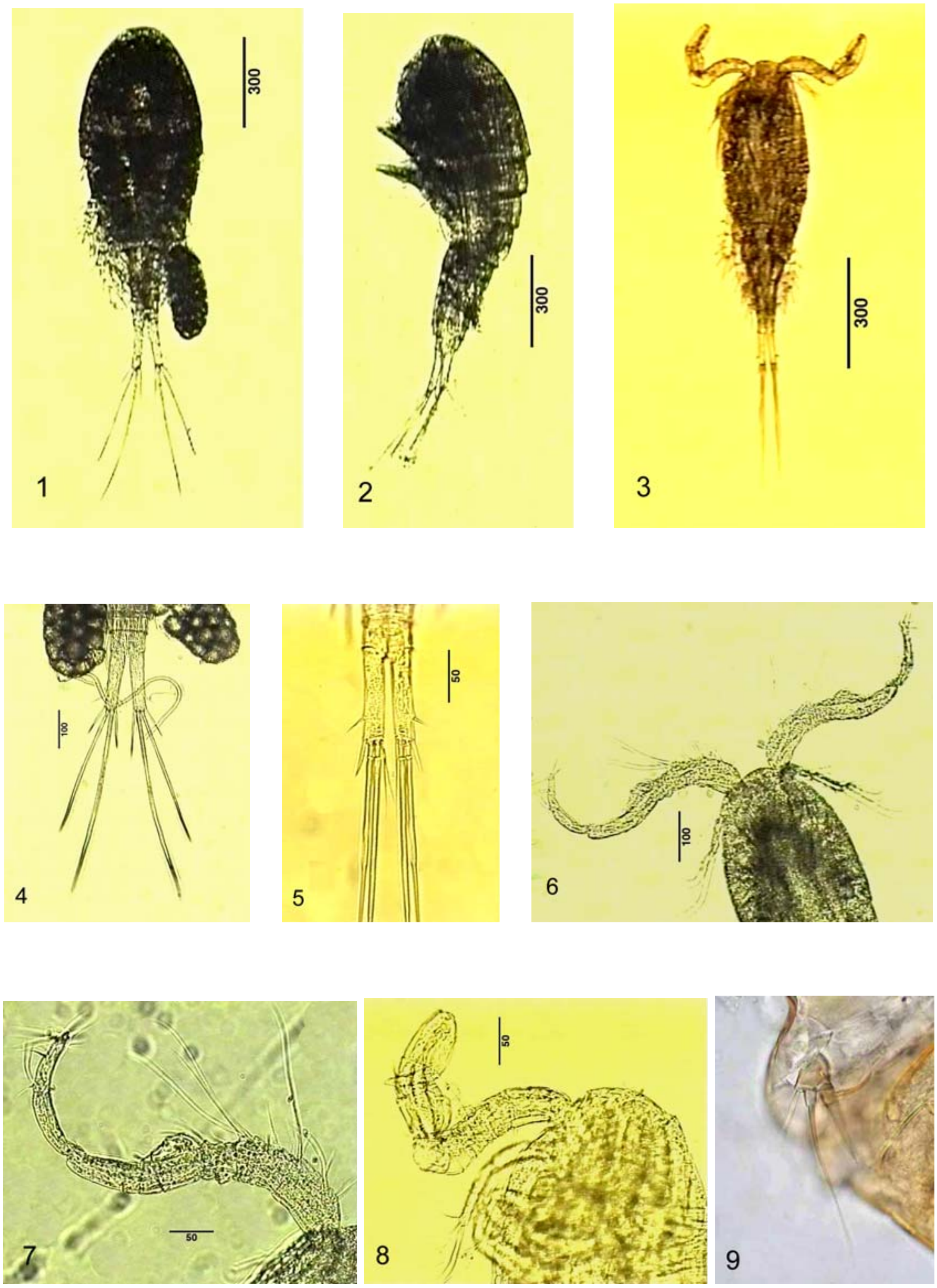


Plate 5.3.26

Copepoda. 1, *Mesocyclops leuckarti*, female dorsally; **2**, *M. leuckarti*, male; **3**, *M. leuckarti*, female UR, egg-sacs and FU; **4**, *Thermocyclops oithonoides*, female dorsally; **5**, *T. oithonoides*, female UR and eggs; **6**, *T. oithonoides*, male laterally; 7, *T. oithonoides*, male CPH and A1 (after Telesh & Heerkloss, 2004).

Plate 5.3.26

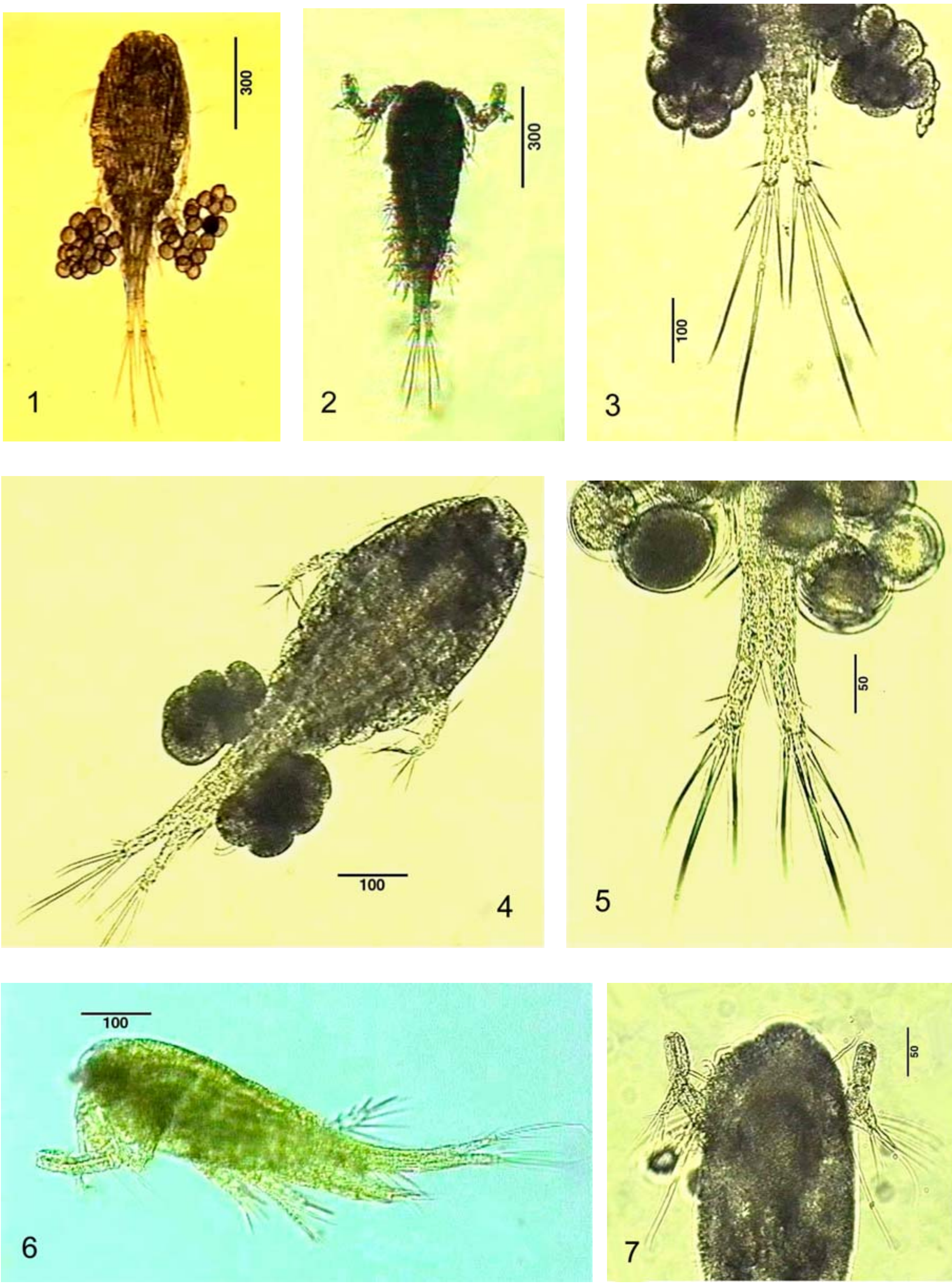
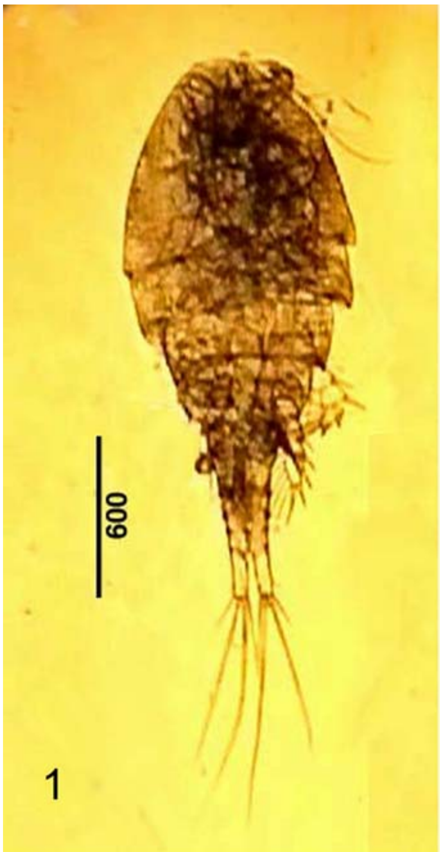


Plate 5.3.27

Copepoda. 1, *Megacyclops viridis*, female ventrally; **2**, *M. viridis*, female FU; **3**, *M. viridis*, female CPHT with A1; **4**, *M. viridis*, copepodite C5 ventrally (after Telesh & Heerkloss, 2004).

Plate 5.3.27



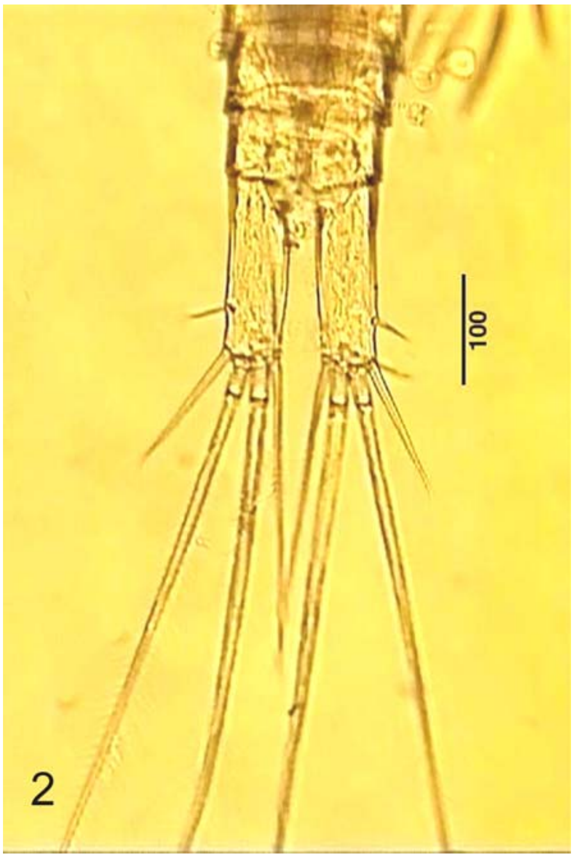


Plate 5.3.28

Copepoda. 1, 2, *Oithona similis*, female ventrally, total length ca. 800 mm, CPHT length 432.3 ± 10.0 µm (Postel et al., 2007); **3**, *O. similis*, young copepodite, CPHT length 293.8 ± 26.6 µm (Postel et al., 2007); **4**, *O. similis*, male ventrally, total length ca. 600 µm; **5**, *O. similis*, male dorsally; **6**, *O. similis*, nauplius dorsally, length 235.4 ± 1.4 µm (Postel et al., 2007); **7**, unidentified cyclopoid copepod; **8**, Harpacticoid copepod, total length ca. 800 µm, lateral view (photos H. Sandberg).

Plate 5.3.28

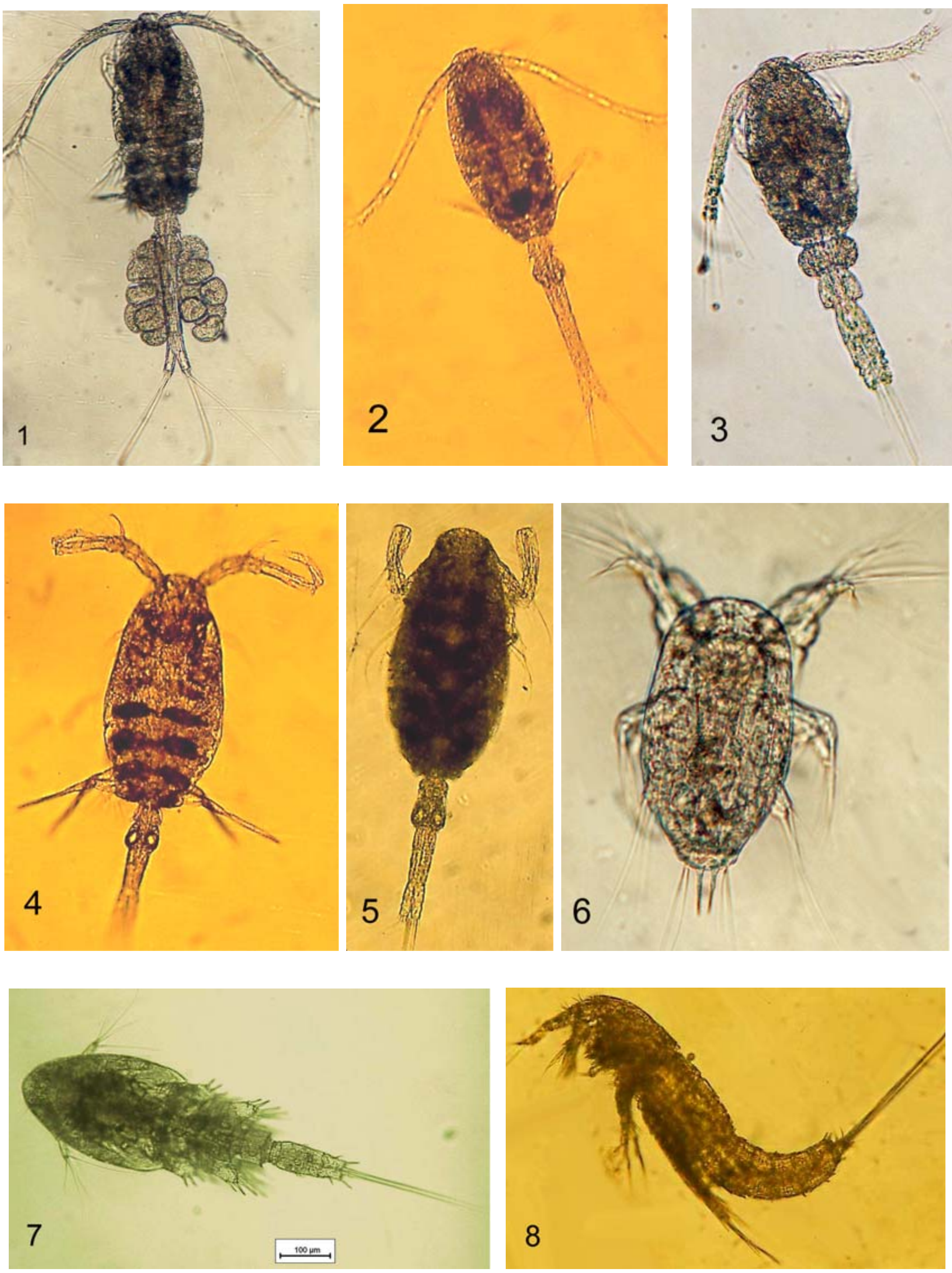


Plate 5.3.29

Copelata. 1, *Fritillaria borealis*, body length 758.5 ± 59.1 µm (Postel et al., 2007); **2**, **3**, *Oikopleura dioica*, adult with fertile gonad, total length ca. 1200 µm, body length ca. 700 µm; **4**, *O. dioica* in the zooplankton sample (photos H. Sandberg).

Plate 5.3.29

Plate 5.3.30

Chaetognatha. 1, *Parasagitta elegans*, general view, total length measured from the tip of the head to the end of the tail, excluding the tail fin, ca. 17 mm (possible range 2-22 mm, after Maciejewska & Margoñski, 2001); **2, 3,** *P. elegans*, head, width ca. 750 µm; **4**, *Parasagitta setosa*, forepart of adult specimen (photos H. Sandberg).

Plate 5.3.30

Plate 5.3.31

Mollusca, meroplanktonic larvae. **1,** Veliger larvae of Gastropoda, width ca. 220 µm; **2-4**, different larval stages of Gastropoda; **5**, young larva of a bivalve mollusc (right) and nauplius of a copepod crustacean (left); **6-9**, different larval stages of Bivalvia (**6, 7**, larvae of *Dreissena polymorpha*) (**1-4, 8, 9**, photos H. Sandberg; **5**, photo courtesy of P. Snoeijs; **6, 7**, after Telesh & Heerkloss, 2004).

Plate 5.3.31

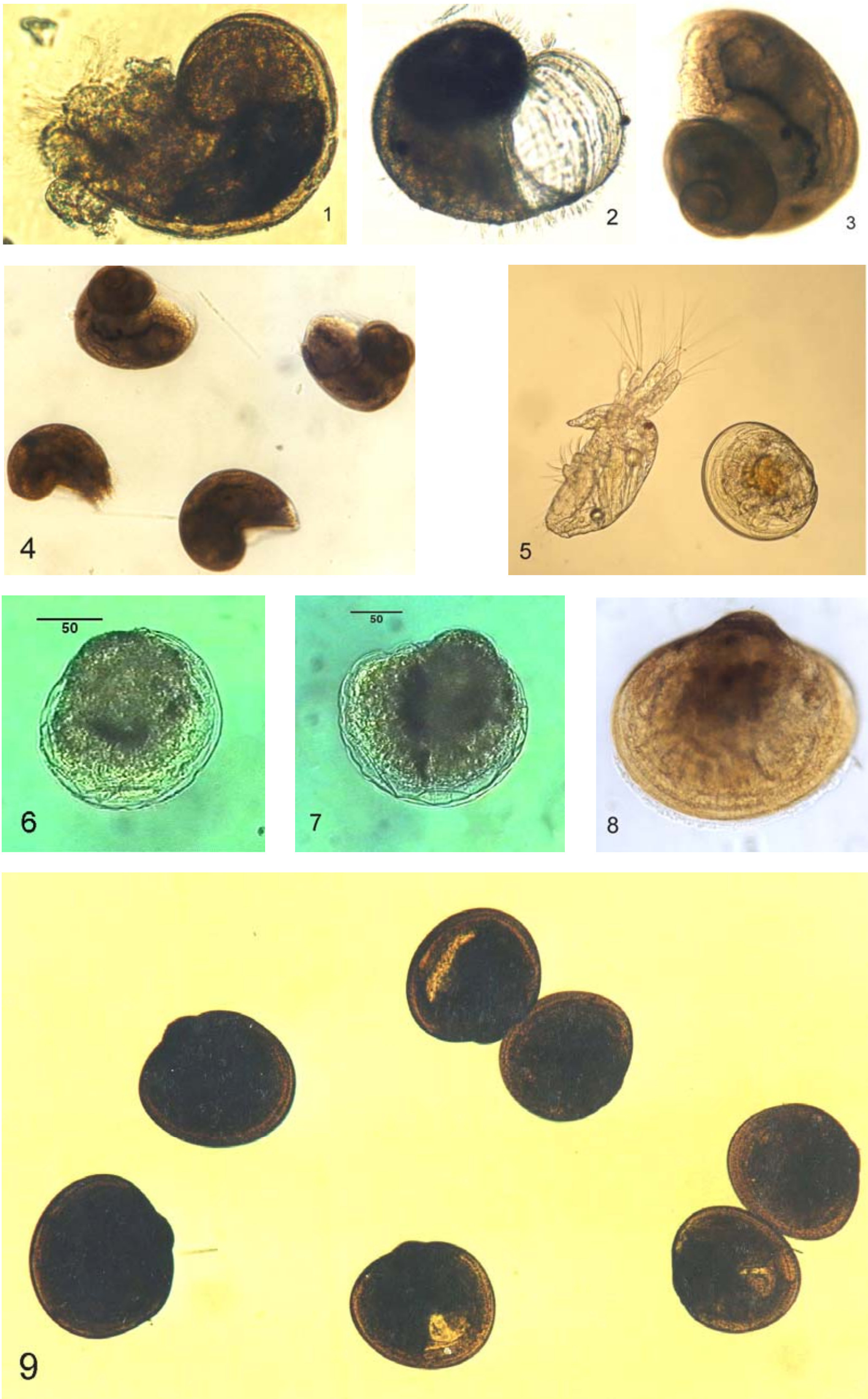


Plate 5.3.32

Crustacea, Cirripedia, larvae. 1-3, Nauplii of *Balanus improvisus*; **4-6**, Cypris stages of *Balanus* sp., lateral view, length ca. 650 µm (**1, 3-6**, photos H. Sandberg; **2**, after Telesh & Heerkloss, 2004).

Plate 5.3.32

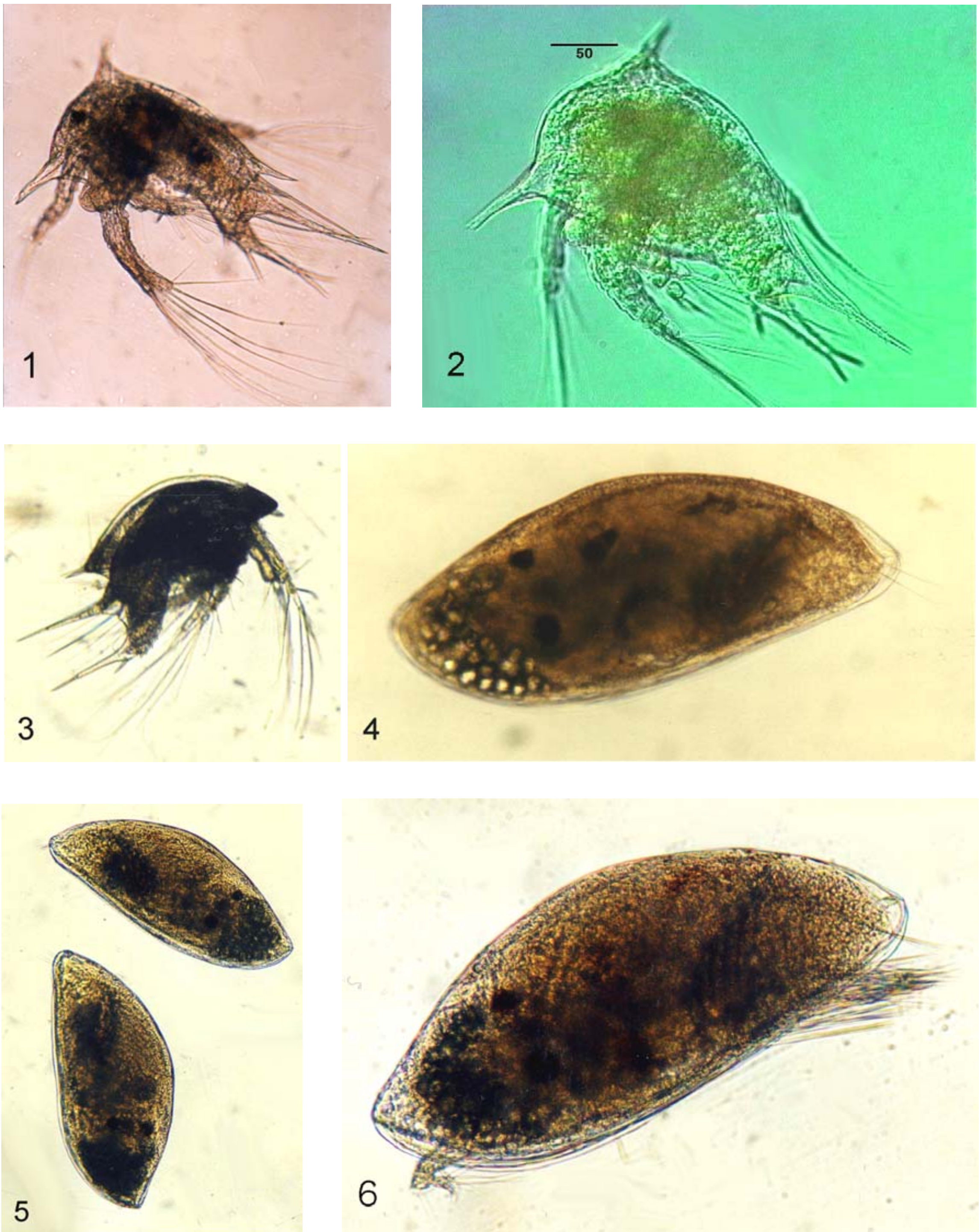


Plate 5.3.33

Polychaeta, larvae at different developmental stages. 1, **2**, Trochophore, length ca. 200 µm; **3-7**, nectochaete of different species (**5**, **6**, *Marenzelleria viridis*, after Telesh & Heerkloss, 2004); **8, 9**, larvae of unidentified polychaete species (photos H. Sandberg).

Plate 5.5.33

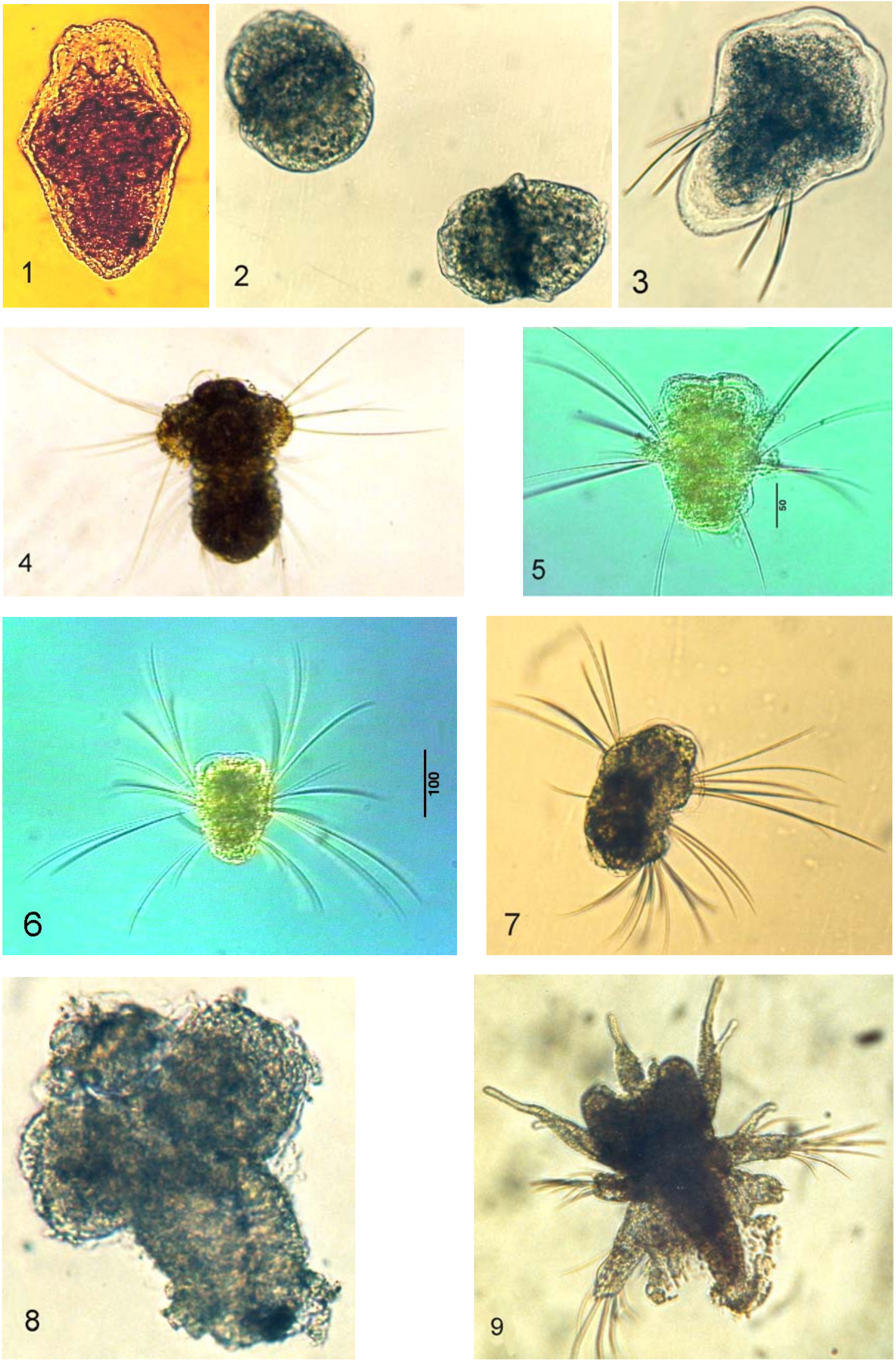


Plate 5.3.34

Polychaeta. 1, *Harmothoe* sp.**,** young specimen**,** length ca. 800 µm; **2**, *Pygospio elegans,* larvae, length 638.3 ± 55.7 µm (Postel et al., 2007). **Turbellaria**. **3**, *Alaurina composita*, adult specimen, length ca. 2.5 mm. The species is among the few holoplanktonic turbellarians of the Baltic Sea; it is forming chains of individuals (zooids) and thus reproducing asexually by transverse division (fission); predators on copepods and cladocerans; one individual may eat 4 copepods per day (Larink & Westheide, 2006) (photos H. Sandberg).

Plate 5.3.34

Plate 5.3.35

1, 2, Bryozoa, Cyphonautes larvae, width ca. 700 µm, has a long-lasting pelagic phase (4-8 weeks) and can be regularly found in plankton; **3**, **Echinodermata,** Ophiopluteus larvae of Ophiuroida, length ca. 400 µm. **4, 5**, **Protista**, unidentified species of Heliozoa, diameter with axopodia ca. 80 µm. **6, 7**, Fish egg of undetermined species, dorsal view, diameter ca. 1 mm (photos H. Sandberg).

Plate 5.3.35

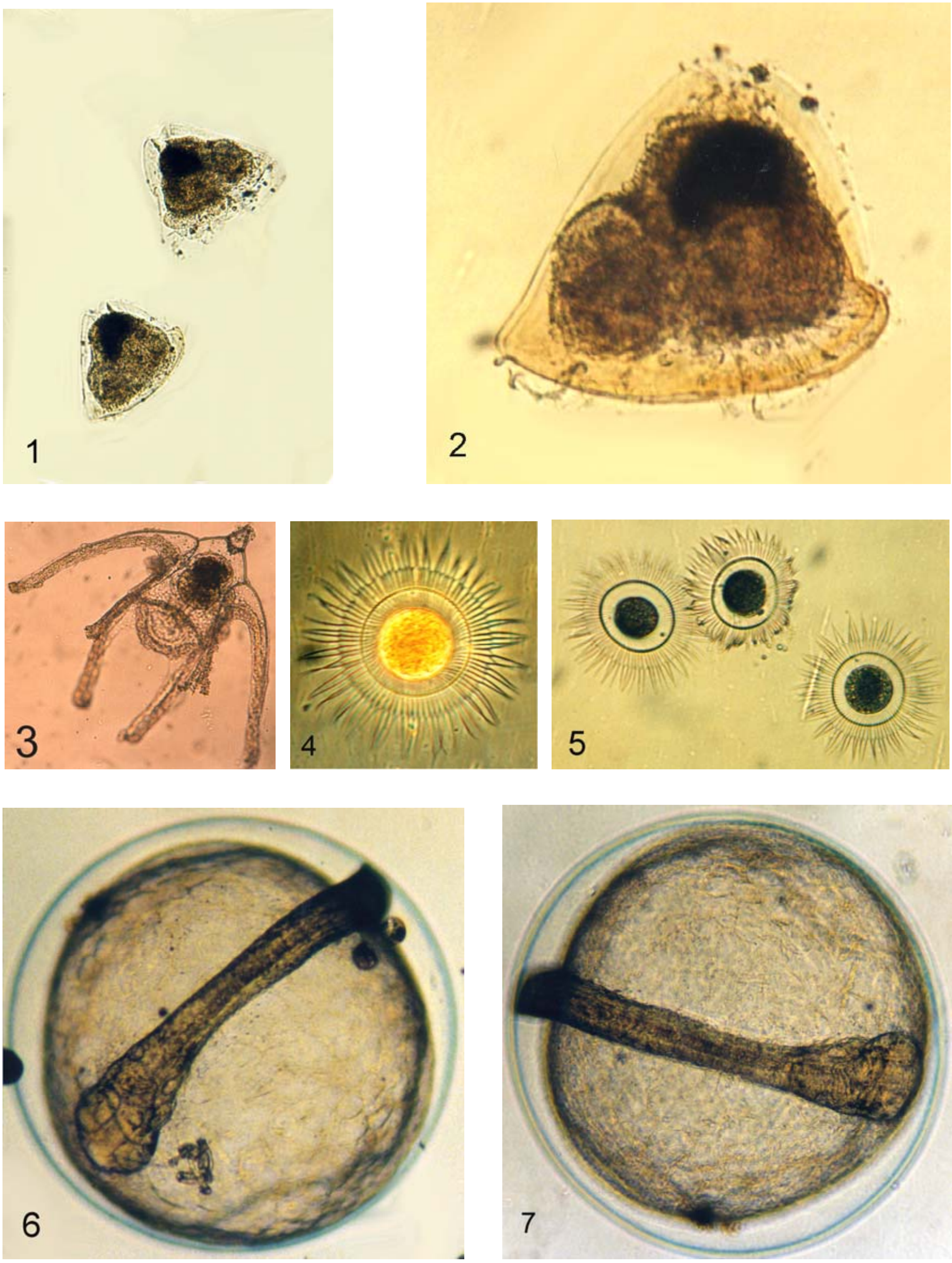


Plate 5.3.36

Ctenophora. 1, 2, *Mnemiopsis leidyi*, adult, average body length 4-6 cm (**1**, in aquarium, photo L. Postel, 2006); **2**, in the sea (photo G. Niedzwiedz, 2008).

Plate 5.3.36

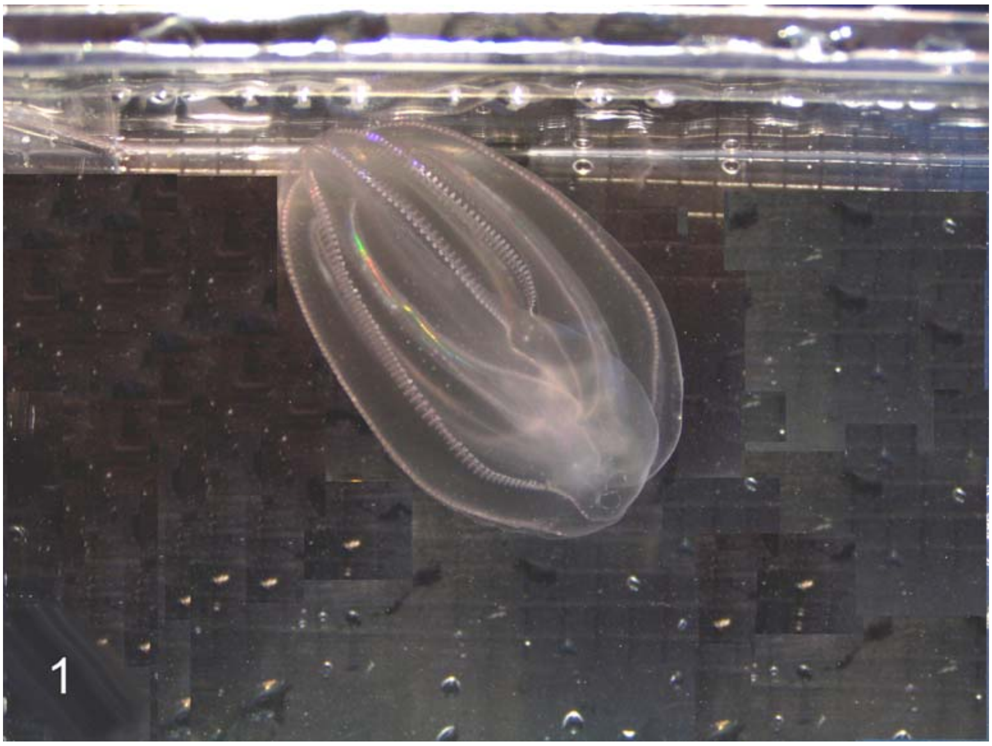


Plate 5.3.37

Ctenophora. 1, *Mnemiopsis leidyi*, juvenile, body length 1284 µm; **2,** *M. leidyi*, juvenile at cydippe stage, body length 1184 µm; **3**, *M. leidyi*, juvenile or a fragment of ctenophore body developing into the adult specimen by regeneration; **4**, *M. leidyi*, juvenile, length 2.5 cm; **5**, *M. leidyi*, adult, dorsal view (photos L. Postel).

Plate 5.3.37

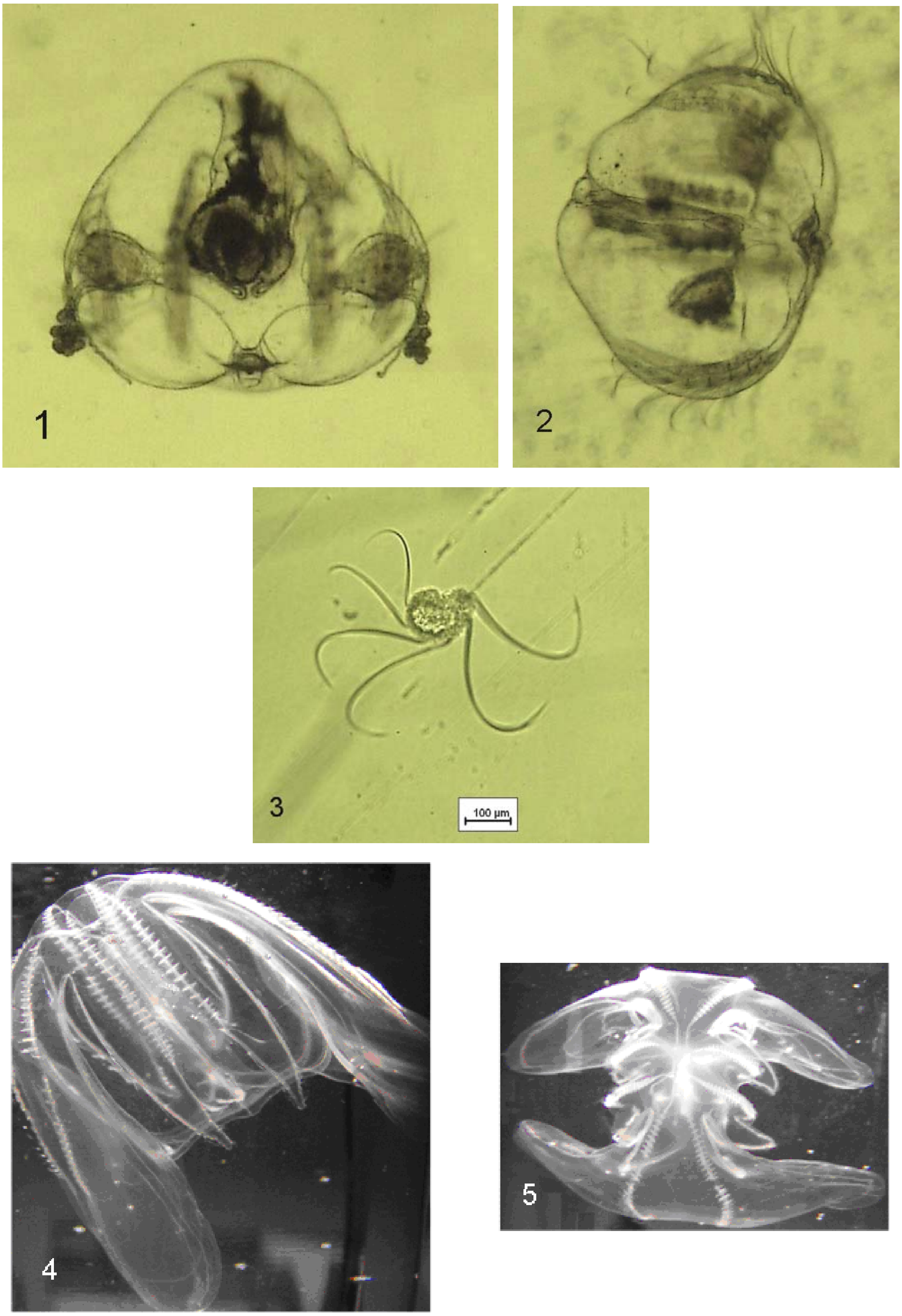


Plate 5.3.38

Cnidaria, Hydrozoa. 1, *Euphysa aurata*, body length 1186 µm; **2**, *Obelia geniculata*, free-swimming mature hydromedusae (photos L. Postel).

Plate 5.3.38

Plate 5.3.39

Cnidaria, Hydrozoa. 1, Planktonic polyp of a hydromedusa; **2, 3,** brachiolaria of a starfish (photos L. Postel).

Plate 5.3.39

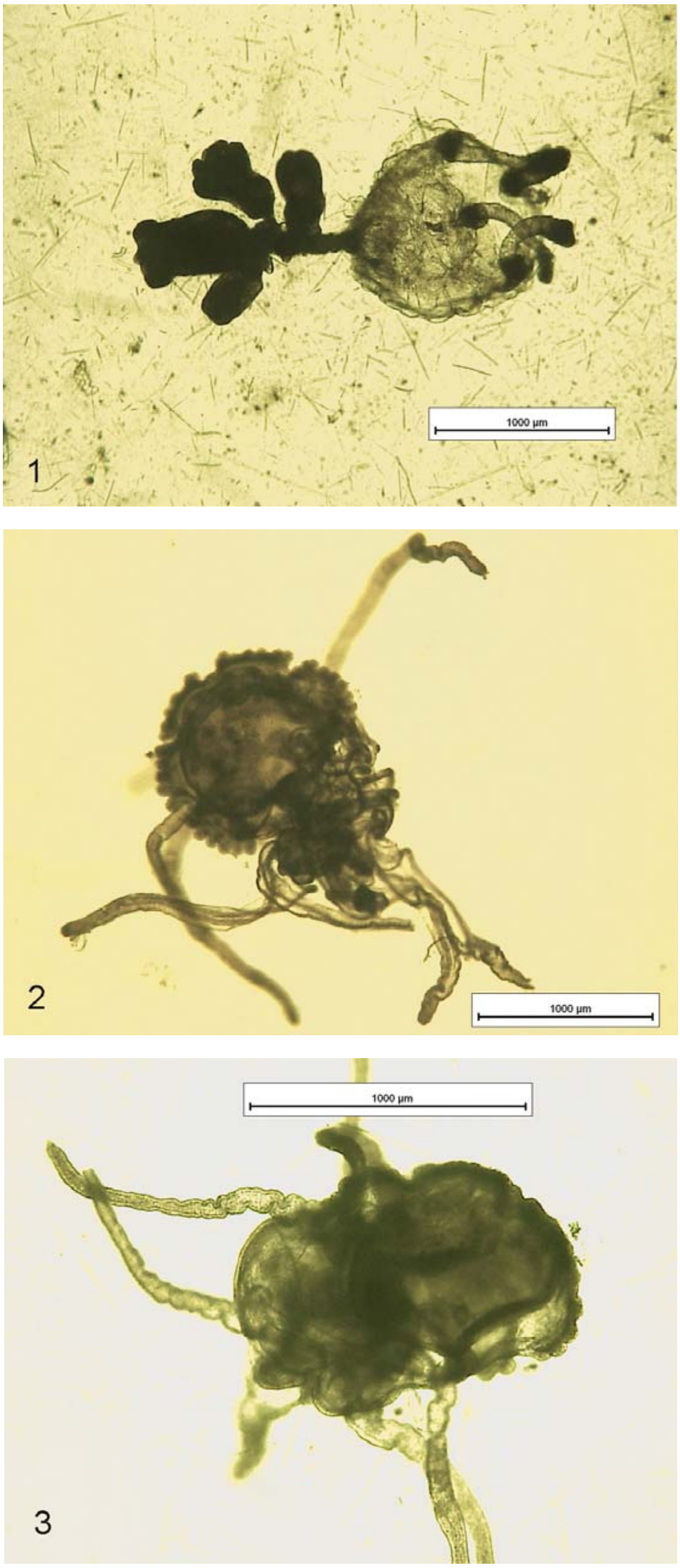


Plate 5.3.40

Epibionts on planktonic crustaceans. 1-6, Epibionts on *Acartia* spp., probably *Colacium vesiculosum* Ehrenberg (Euglenophyceae) (after Moehlenberg & Kaas, 1990), photos L. Postel.

Plate 5.3.40

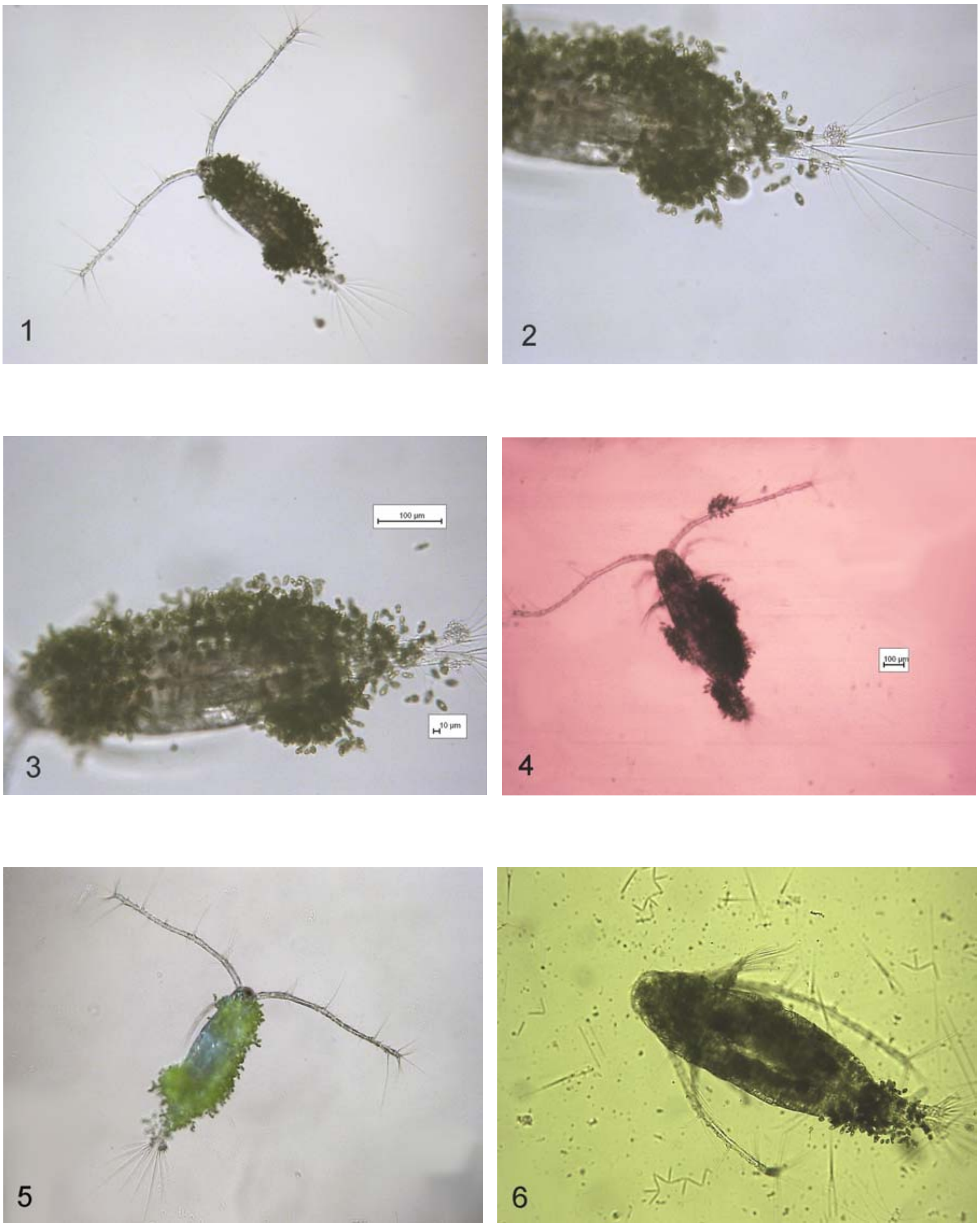


Plate 5.3.41

Epibionts on planktonic crustaceans. 1-2, Epibionts on *Acartia* sp.; **3**, epibionts on *Centropages hamatus*; **4-6**, epibionts (possibly *Ellobiopsis chattoni*) on *Pseudocalanus elongatus* (photos L. Postel).

Plate 5.3.41

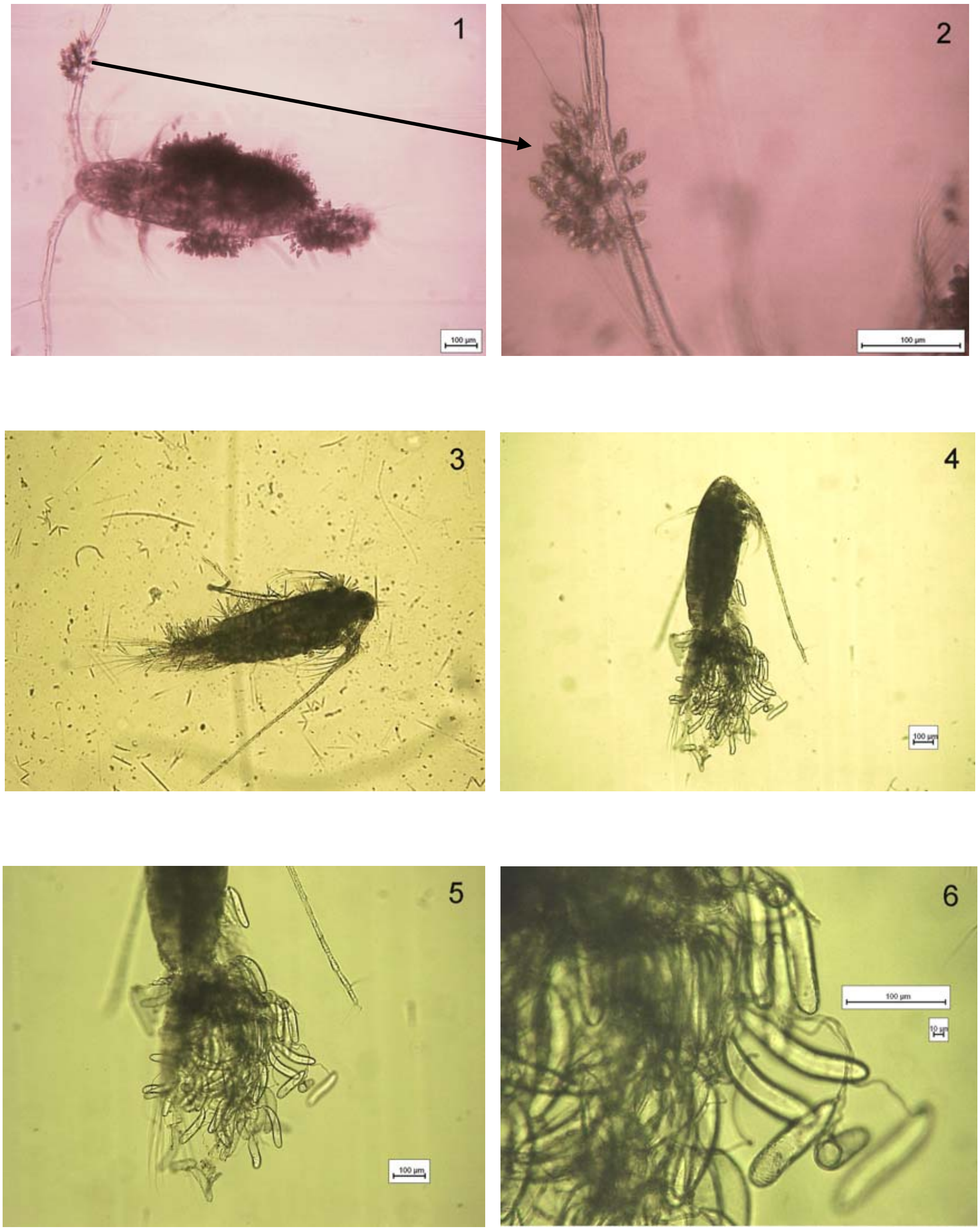


Plate 5.3.42

Epibionts on planktonic crustaceans. 1-3, Unidentified epibiont on *Temora longicornis* (photos L. Postel).

Plate 5.3.42

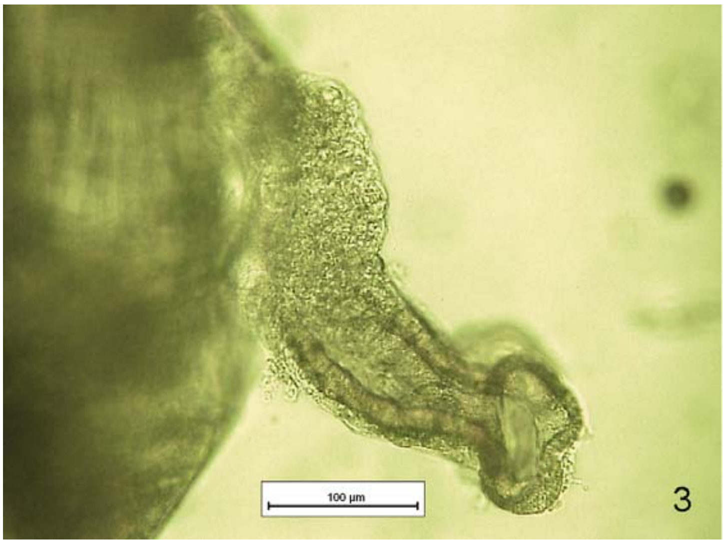
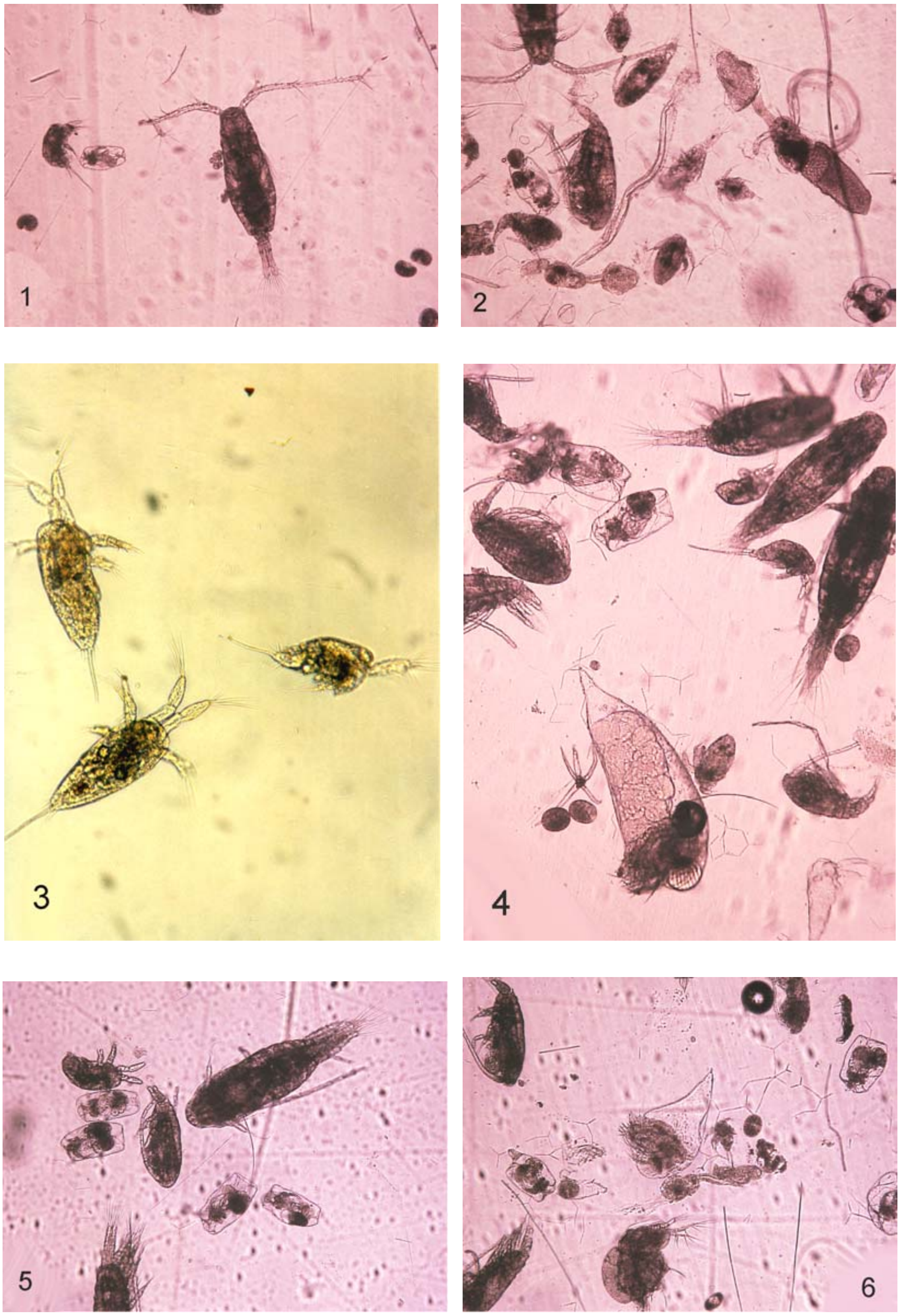


Plate 5.3.43

1-2, 4-6, General view of a zooplankton sample from the Baltic Sea; 3, *Centropages hamatus*, nauplii (photos H. Sandberg).

Plate 5.3.43



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SELECTED ZOOPLANKTON INTERNET DATA BASES

- Integrated Taxonomic Information System (ITIS):

http://www.itis.gov/

- The European Register of Marine Species (ERMS):

http://www.marbef.org/data/erms.php

- The World Register of Marine Species (WoRMS):

http://www.marinespecies.org

- ICES Identification Leaflets for Plankton:

http://www.ices.dk/products/fiche/Plankton/START.PDf

- The user-friendly guide to coastal planktonic ciliates:

http://www.liv.ac.uk/ciliate/intro.htm

- An image-based key to the zooplankton of the northeast USA:

http://cfb.unh.edu/CFBkey/html/index.html

- Zooplankton of the Great Lakes:

http://www.cst.cmich.edu/users/mcnau1as/zooplankton%20web/

- The Great Lakes water life photo gallery with a list of rotifer and crustacean sites:

http://www.glerl.noaa.gov/seagrant/GLWL/GLWLife.html

- Plankton*Net @ Roscoff:

http://planktonnet.sb-roscoff.fr/index.php

- International Code of Zoological Nomenclature:

http://www.iczn.org

- National Institute for Environmental Studies:

http://www.nies.go.jp/chiikil/protoz/identifi.htm

INDEX OF LATIN NAMES

Only <u>valid</u> species and genera Latin names are included in the Index.
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