The global diversity and biogeography of Isopoda

Lena Hartebrodt

A thesis submitted in fulfilment of the requirements for the degree of

Doctor of Philosophy in Marine Science

The University of Auckland

May 2024

Abstract

The order Isopoda is a species-rich and morphologically diverse taxon of peracarid crustaceans. They not only occur in marine environments but have also successfully conquered freshwaters and the terrestrial realm. This thesis examines the global diversity and biogeographic patterns of isopods within all three environments. After summarising what is known about their natural history, distribution, and ecological and economic importance, attention is drawn to the taxonomic diversity of isopods and their rate of description. A global list of accepted species names, including their authorities and the year of first description, was gathered from the World Register of Marine Species (WoRMS). Since the first formal description of an isopod species by Linnaeus in 1758, 10,687 isopod species in 1,557 genera and 141 families have been described by a cohort of 755 first authors. Although the number of authors has increased many-fold over time, the average number of species described per first author has declined, and the description rate has slowed down. Shifting the attention to global biodiversity patterns within isopods, the latitudinal diversity gradient (LDG) in species richness is first examined, followed by bioregionalisations within aquatic and terrestrial environments. To do so, a dataset of worldwide occurrence records was compiled from the Ocean Biogeographic Information System (OBIS) and the Global Biodiversity Information Facility (GBIF). Isopods exhibit a bimodal LDG with higher species richness in the southern hemisphere for marine isopods and in the northern hemisphere for non-marine isopods. Sampling bias in the data does not significantly affect the overall shape of the LDG. However, when accounted for, the location of diversity peaks tends to move towards tropical latitudes compared to the observed species richness, which is highest in mid-latitudes. A clustering algorithm was employed to identify distinct biogeographic regions based on their species composition. It delineated 33 marine, 28 terrestrial, and 23 freshwater bioregions – all with high endemicity – many of which agree with other biogeographical frameworks in the respective environments. The analysis also revealed substantial geographical gaps in the data, especially in Africa, Asia, Indonesia, South America, and offshore marine areas, which future research should aim to close.

Acknowledgements

I would like to thank my former main supervisor, Prof. Dr. Mark J. Costello, for providing me with the opportunity to start this PhD back in 2018. Thank you for always giving quick feedback, letting me know that you appreciate my work, and for not giving up on me when I was forced to suspend my PhD for an indefinite amount of time.

I am also grateful to my current main supervisor, Dr. Richard Taylor, who “adopted” me and my PhD project when the need arose to find a new supervisory team. Thank you for giving me the opportunity to finish what I started. In this regard, too, I must thank Dr. Brendon Dunphy for taking on the role of my co-supervisor. Thank you for acknowledging the hardship I went through.

I appreciate the company and friendship of many colleagues who came and went during my prolonged PhD journey. Thank you for enjoyable lunchtime conversations, the occasional after-work drinks, and the warm welcome you gave me when I could finally continue my work.

Many thanks to my family for letting me go my own way even though you could not comprehend it. Thank you for still being proud of me and not forcing me to give up my dream when my whole world crashed. Mum, even with all the consequences I had to face, I am grateful that I could be with you at the end. I love you, and I miss you terribly! I wish I could let you know that, eventually, I got the chance to finish my PhD.

Finally, I am most grateful to my partner Jonathan. You have been an enormous emotional support, and you stood by my side when I went through the worst. Thank you for staying through the bad days! Now it is time for the good days to return and hopefully to last … for a while.

Table of Contents

List of Tables

List of Figures

1. General Introduction

1.1. The world's biodiversity

Biodiversity can be measured at different scales, from genes to ecosystems. At the global level, a commonly used metric is species richness: the number of species that exist on Earth. But how many species are there? (See section 3.1. for a few examples of global estimates.) There is not only the known biodiversity, i.e. species that have been described and documented in the scientific literature, but also an unknown component of yet undescribed and undiscovered species. So, how can we estimate Earth's biodiversity? Estimates are based mainly on the following methods: expert opinion polls, proportions of undescribed species in samples, and extrapolation from past description rates (Appeltans et al., 2012). Expert opinion and proportions of undescribed species tend to extrapolate regional knowledge and data to a global scale, which is problematic since species richness varies between regions; therefore, regional data should not be used to estimate global species diversity (Poore & Wilson, 1993). For marine isopods, for instance, high species richness with considerable proportions of undescribed species is documented for Australian waters (Poore et al., 1994, 2015) and for the Southern Ocean (Brandt, Brix, et al., 2007), while the deep-sea of the Norwegian and Greenland seas yields only low species numbers in comparison (Poore & Wilson, 1993). Extrapolations of species richness of either one of these regions would lead to a highly biased global estimate. Ideally, extrapolations should be based on a global dataset to get a more comprehensive estimate of the overall species richness and buffer regional biases. Today, centralised, continuously updated and curated databases like the World Register of Marine Species (WoRMS, https://www.marinespecies.org) provide the opportunity to perform global analyses with a data-driven approach. However, extrapolations based on past description rates may be associated with large margins of error unless a taxon's species inventory is at least 90% complete (Bebber et al., 2007). A few studies have previously included the isopod data compiled in WoRMS in their analyses (marine isopods: Appeltans et al., 2012; Costello et al., 2012; parasitic isopods: Costello, 2016). However, at the time of these studies, the isopod inventory in WoRMS was still highly incomplete, and no meaningful estimate of global species diversity could be made. Furthermore, none of these works incorporated data for the whole order Isopoda, including freshwater and terrestrial species. No comprehensive and detailed review of the known and estimated biodiversity of aquatic and terrestrial Isopoda has been done to date. Such an assessment is performed in Chapter 3.

1.2. Global biogeographic patterns of species richness

Biodiversity is not uniformly distributed across the globe, with some regions being exceptionally rich in species while others are notably lacking in diversity. Biologists have aimed to describe and understand general biodiversity patterns for two centuries. Early naturalists who pondered broad-scale relationships between organisms and the environment include Alexander von Humboldt, Charles Darwin and Alfred Russel Wallace. Wallace was among the first to divide the terrestrial realm into distinct biogeographic regions based on the animal communities they harbour (Wallace, 1876). Similar attempts to structure the marine realm were made much later, and in the early stages, boundaries between oceanic biogeographic regions were thought to be much more blurry than on land (Briggs, 1974; Ekman, 1953). Since then, countless other studies have dealt with various gradients in diversity and scrutinised the geographical distributions of animals and plants on multiple spatiotemporal scales.

1.2.1. The latitudinal diversity gradient

One of the most outstanding and extensively studied patterns in biogeography is the latitudinal gradient in species richness. For a long time, it was considered a unimodal bell shape with increasing diversity from the poles toward the equator (e.g., Stehli et al., 1969). This pattern holds true for some terrestrial organisms (Clarke & Crame, 1997; Dantas & Fonseca, 2023) and a few marine groups (e.g., Boltovskoy & Correa, 2017). However, the latitudinal diversity gradient is taxon-specific, and in recent years, evidence has accumulated showing that for most taxa, the gradient is indeed bimodal with a dip at or near the equator (Cerezer et al., 2022; Chaudhary et al., 2016). Claims that the observed bimodal pattern stems from insufficient sampling of equatorial regions (Fernandez & Marques, 2017) could be refuted, with several studies showing that sampling bias does not affect the overall shape of the gradient (e.g., Chaudhary et al., 2017; Rivadeneira & Poore, 2020). Although the generality of the pattern itself is widely accepted (Hillebrand, 2004a), no consensus about the underlying causes that form and maintain the latitudinal diversity gradient has yet been reached (Currie et al., 1999). Numerous hypotheses have been presented, which can be roughly grouped into evolutionary and ecological processes. Some researchers propose that the modern latitudinal diversity gradient has been formed due to varying speciation and extinction rates (Allen & Gillooly, 2006; Crame, 2023; Krug et al., 2007). Others have shown strong correlations between latitudinal species richness and environmental variables like temperature, salinity, and precipitation (Boltovskoy & Correa, 2017; Cruz-Motta et al., 2020; Dantas & Fonseca, 2023) or else an effect of predation intensity (Ashton et al., 2022; Freestone et al., 2021) or the seasonality of primary productivity (Crame, 2020; S. J. Culver & Buzas, 2000). Many agree, however, that there is no single underlying cause and that local and regional processes also play an important role (Condamine et al., 2012; Cruz-Motta et al., 2020; Gaston, 2000). While early broad-scale studies on biogeographic patterns in isopod species richness often did not find a clear latitudinal gradient within the group or broader region they examined (Kussakin, 1973; G. D. F. Wilson, 1998), current research points to a well-formed bimodal latitudinal diversity gradient both in the ocean and on land (Rivadeneira & Poore, 2020; Saeedi et al., 2022; Sfenthourakis & Hornung, 2018). Rivadeneira and Poore (2020) performed the most comprehensive analysis of the latitudinal diversity gradient of marine isopods in spatial and taxonomic scales and the amount of data so far. They, too, used occurrence records obtained from the Ocean Biogeographic Information System (OBIS, https://obis.org) as is done in this thesis. However, the data their analysis was based on are several years old now and include only a bit over half the number of marine species in this thesis' analysis. Moreover, no comprehensive study of latitudinal patterns in species richness of the whole order Isopoda has been done yet. Here, data obtained from OBIS are complemented with occurrence records from the Global Biodiversity Information Facility (GBIF, https://www.gbif.org) to include freshwater and terrestrial isopod species. Based on such an extensive dataset, a global analysis of latitudinal diversity gradients of and within Isopoda is performed in Chapter 4.

1.2.2. Bioregionalisation

Since Wallace's famous division of the terrestrial realm into six zoogeographic regions (Wallace, 1876), abundant biogeographic studies encompassing a growing body of empirical data have been conducted on various spatiotemporal and taxonomic scales. For instance, Holt et al. (2013) analysed the distributions and phylogenetic relationships of more than 21,000 vertebrate species to refine Wallace's biogeographic world map. They identified 20 distinct zoogeographic regions, further grouped into 11 larger realms. Before that, Olsen et al. (2001) had developed a detailed map of the world's terrestrial ecoregions (nested within biomes and realms), which was refined in 2017 (Dinerstein et al., 2017). Abell et al. (2008) developed a similar map of the world's freshwater ecoregions with the same aim as its terrestrial counterpart in acting as a base map for conservation planning. Similar frameworks for the marine realm have been produced through meta-analysis by Spalding and colleagues focusing on shallow coastal waters (Spalding et al., 2007) and pelagic surface waters (Spalding et al., 2012), respectively. Further biogeographic maps of the world's oceans were drawn for the mesopelagic zone (Sutton et al., 2017) and the deep sea (Watling et al., 2013). In contrast to approaches that were mainly based on oceanographic features, the marine biogeographic realms (both continental-shelf and offshore deep-sea realms) delineated by Costello et al. (2017) were based on the analysis of empirical distribution data of 65,000 marine species. Recently, a growing number of studies have been conducted on the geographic diversity patterns of specific taxa regionally and globally. Examples include global studies on bioregionalisation in brittle stars (Bribiesca-Contreras et al., 2019; Victorero et al., 2023), deep-sea anthozoans (Watling & Lapointe, 2022), polychaetes (Pamungkas et al., 2021), and amphipods (Arfianti & Costello, 2020), as well as regional research on polychaetes and isopods from the northwestern Pacific Ocean and adjacent Arctic Ocean areas (Saeedi et al., 2022) or shallow-water isopods from the East Pacific (del Carmen Espinosa-Pérez & Hendrickx, 2006). Plenty of regional-scale studies, often with a taxonomic focus on a specific isopod family or genus, can be found in the scientific literature. However, a comprehensive global study of bioregionalisation in isopods is still lacking and is performed in Chapter 5 for marine, freshwater and terrestrial realms.

1.3. The taxon of interest: Isopoda Latreille, 1816

Isopods are a species-rich group of benthic peracarid crustaceans. A detailed overview of their natural history, biodiversity and geographic distribution is presented in Chapter 2. Here, only a general summary of the taxon shall be given.

In Isopods a carapace is absent and oostegites form a ventral brood pouch, in which the eggs are deposited and undergo a direct development into a juvenile manca stage (Ax, 2000). The taxon is morphologically diverse (see Fig. 1.1), but generally, they have a segmented, dorsoventrally flattened body with usually seven pairs of pereopods commonly employed for walking and crawling. Even though some isopod species have evolved the ability to swim, walking is their preferred method of locomotion, with swimming acting more as an escape mechanism (Hessler, 1993; Hessler & Strömberg, 1989). Given their weak dispersal abilities, isopods are expected to show distinct biogeographic patterns with high endemicity. The ubiquity of isopods in all of the world's oceans and on almost every continent makes them an ideal taxon for the analysis of basic global diversity patterns.

Figure 1.1. Examples of diverse isopod morphologies. **(a)** *Neastacilla tuberculata* (Arcturidae), with long antennae and an elongated, cylindrical body; **(b)** *Paranthura punctata* (Paranthuridae), another example of a long and slender body outline; **(c)** An egg-bearing, distorted female of a species of Bopyridae; **(d)** *Cirolana* sp. (Cirolanidae), ventral view, displaying the typical elliptical body shape of an isopod; **(e)** *Idotea metallica* (Idoteidae), oval body shape with a distinct square telson; **(f)** *Paraserolis polita* (Serolidae), with a trilobite-like appearance; **(g)** *Scyphax ornatus* (Scyphacidae), with styliform uropods and pereopods with many stout setae; **(h)** *Plakarthrium typicum* (Plakarthriidae), ventral view, extensions of coxae and peduncular articles of antennae forming oval outline; **(i)** *Gnathia* sp. (Gnathiidae), male with distinctive protruding mandibles, scale bar: 1 mm; **(j)** *Gnathia* sp. (Gnathiidae), gravid female carrying about 30 eggs, scale bar: 1 mm; **(k)** *Munna neozelanica* (Munnidae), with long, slender pereopods. Photos are from museum specimens. After Hartebrodt (2020).

1.4. Thesis objectives and structure

One can find plenty of local and regional studies in the published literature that report on the geographical distribution of either selected isopod species or a genus, a family, or an ecological group of isopods that has been studied in the area. Only a few review articles attempted to summarise the diversity and/or biogeography of isopods on a global scale. However, these reviews only dealt with certain suborders, families, or isopods occurring in a specific environment. So far, no comprehensive study has been published detailing the diversity and biogeography of the whole order Isopoda. This species-rich order consists of marine, freshwater and terrestrial species, and this thesis aims to objectively analyse the number of species in this taxon and how this diversity is distributed globally.

The following chapters aim to give an overview of the global biodiversity and biogeography of the order Isopoda:

Chapter 2: The superdiverse order Isopoda (Crustacea: Peracarida) – what is known about their natural history, diversity and distribution?

This chapter gives an overview of the species-rich crustacean order Isopoda. It summarises their natural history, including the fossil record and the general morphology of isopods. Further, it details the various habitats in which isopods occur and reviews the extent of biogeographic work conducted on numerous groups of the order. Finally, it outlines the ecological and economic importance of Isopoda.

A condensed version of this chapter, which focused on marine isopods only, has been published as an encyclopaedia article in Elsevier's "Encyclopedia of the World's Biomes": https://doi.org/10.1016/B978-0-12-409548-9.11682-3.

Chapter 3: Progress in the discovery of isopods – is the description rate slowing down?

The objective of this chapter is to review the progress in the formal description of isopod species and outline their biodiversity. From data available in the World Register of Marine Species (WoRMS), a global list of accepted species names, including their authorities and year of description, was compiled. Changes in the rate of description over time are examined in relation to the number of taxonomists involved. Additionally, by applying a statistical model, predictions about the number of new species descriptions until the end of the 21st century are provided.

This work has been published in PeerJ: https://doi.org/10.7717/peerj.15984.

Chapter 4: Isopod taxonomic diversity is bimodal with latitude

In this chapter, the pattern of the latitudinal diversity gradient in species richness is analysed for the order Isopoda as a whole as well as for various environmental and ecological subgroups. A global dataset of occurrence records compiled from the Ocean Biogeographic Information System (OBIS) and the Global Biodiversity Information Facility (GBIF) is used to examine patterns in alpha and gamma diversity. Additionally, Hill numbers are employed to account for sampling bias in the data and to explore how it affects the shape of the gradient.

Chapter 5: Global bioregionalisation and endemicity of isopods

This chapter explores the global biogeographic structuring of isopods. Making use of the interactive web application “Infomap Bioregions” and occurrence data available in the Ocean Biogeographic Information System (OBIS) and the Global Biodiversity Information Facility (GBIF), bioregions within the marine, terrestrial, and freshwater realms are identified based on their species composition. Additionally, rates of endemicity are calculated for each resulting bioregion.

2. The superdiverse order Isopoda (Crustacea: Peracarida) – what is known about their natural history, diversity and distribution?

2.1. Systematics and fossil record

The speciose peracarid order Isopoda Latreille, 1816 is one of the morphologically and ecologically most diverse taxa within the subphylum Crustacea. The over 10,600 described isopod species have been classified into 12 suborders, just over 140 families and more than 1,550 genera (WoRMS, 2023). Such extensive taxonomic diversity has attracted the attention of many experts trying to resolve the questions of isopod systematics, though they often do not reach a consensus (Martin & Davis, 2001). While there is broad agreement that Isopoda are a monophyletic group (Brusca & Wilson, 1991), the argument about their sister taxon still needs to be settled. Based on morphological characters many experts favour the hypothesis that Amphipoda is the sister group to isopods (Poore, 2005; Wills, 1998), whereas other studies, some of which include molecular data to corroborate their results, do not find evidence for this claim and instead suggest a sister-group relationship between Tanaidacea and Isopoda (Richter & Scholtz, 2001; G. D. F. Wilson, 2009). The suborders Phreatoicidea (which is confined to freshwater environments) and Asellota (a very species-rich group that comprises many marine and some freshwater species) are generally considered as the oldest isopod lineages (Brusca & Wilson, 1991; Dreyer & Wägele, 2002). The Oniscidea, a group of almost entirely terrestrial isopods, are also seen as one of the earliest derived isopod lineages (Brusca & Wilson, 1991), but there are still arguments about the monophyly of this taxon (Dimitriou et al., 2019; Dreyer & Wägele, 2002; Lins et al., 2017). Phylogenetic analyses continue to reshuffle isopod systematics. For example, a long-standing taxon, the Flabellifera, is now widely disregarded (Brandt & Poore, 2003; Brusca & Wilson, 1991), while new evidence supports the reinstatement of Epicaridea as a suborder (Yu et al., 2018). Numerous taxonomic revisions keep changing the classification of isopod species by transferring them into new genera, placing them into synonymy, etc. (e.g., Boyko & Williams, 2023). With many new species described each year that shed new light on isopod taxonomy and phylogeny, there is still a long way to go until the systematics and taxonomy of Isopoda are fully resolved.

The fossil record of isopod crustaceans is rich compared to other peracarid taxa (Etter, 2014), although fossils of individual groups can be quite rare, and specimens are often incomplete. The absence of a waxy epicuticle in most isopods tends to lead to rapid decay after death and, therefore, hinders fossilisation (Girling, 1979). Nevertheless, isopods can become fossilised in a wide variety of depositional environments (Etter, 2014). The oldest known fossil isopod, *Hesslerella shermani*, was described by Schram (1970) as a representative of Phreatoicidea and dates back to the late Carboniferous more than 300 million years ago (mya). The first, and until recently only, known asellote fossil was discovered in slabs dating back to the Triassic period around 210-215 mya (Selden et al., 2016). Fossil records from the Jurassic period are more abundant and include fossilised trackways (Gaillard et al., 2005) and a well-preserved cirolanid isopod (Etter, 2014). The earliest specimens of parasitic isopods (assumed to be representatives of Gnathiidae and epicarideans) also stem from this period (Klompmaker et al., 2014; Nagler et al., 2017). Another rare, fossilised case of parasitism by a cymothoid isopod was found in deposits from the Cretaceous, showing the isopod still attached to its host fish (Stinnesbeck et al., 2022). While it is common for terrestrial isopods to be preserved in amber (Lu et al., 2023), it is very rare to find marine forms preserved in this way. Schädel et al. (2021) described such a fossil from Myanmar, having an assemblage of more than 100 epicaridean larvae or paedomorphic males enclosed in a single piece of 100-million-year-old amber. A diverse assemblage of shallow-water isopod species from the Upper Cretaceous, which can be linked to the extant suborders Cymothoida, Asellota and Valvifera, provides evidence for the existence of a fairly modern isopod fauna as early as 100 mya (G. D. F. Wilson & Morel, 2022). The fossil record also shows that all major modern families of the terrestrial Oniscidea were already present and widely distributed in the mid-Cenozoic era, about 20 mya (Broly et al., 2013; Lu et al., 2023).

2.2. Morphology

As mentioned earlier, isopods display a great diversity in body plans. Most are dorsoventrally flattened and broadly oval, whereas the body outline of others is narrow and elongate, sometimes cylindrical and rather vermiform. One outstanding characteristic that Isopoda share with Amphipoda and Ingolfiellida is the absence of a carapace (Ax, 2000; Lowry & Myers, 2017). Generally, the isopod body is divided into three sections: the cephalon, pereon and pleon. All segments of the cephalon are fused, including the first (and sometimes second) thoracomere. It bears two pairs of antennae, mandibles, two maxillae and maxillipeds, which are the appendages of the first fused thoracic segment (Menzies & Kruczynski, 1983). Most isopods have well-developed sessile compound eyes, although in subterranean, deep-sea, and some parasitic species, eyes can be greatly reduced or completely absent. The pereon usually has seven free segments, each bearing a pair of uniramous pereopods. Gnathiid isopods, most notably, differ in this regard by having only five pairs of pereopods and their respective body segments (Lawrence & Keast, 1990; Menzies & Kruczynski, 1983). The pleon comprises five segments that can be fused to various degrees and a pleotelson, which is formed by the telson and a fused sixth abdominal segment (Lawrence & Keast, 1990). This fused pleonite bears the biramous uropods, which can vary from styliform to broad and often form a tail fan with the telson (Ax, 2000; Lawrence & Keast, 1990). The anterior five pleonites each bear a pair of biramous lamellar pleopods, which perform a respiratory function and can aid swimming. It is assumed that the aforementioned gill function of the pleopods correlates with the caudal displacement of the heart in isopods (Ax, 2000). In male isopods, the second (and sometimes first) pleopods are modified to bear an appendix masculinum and assist in sperm transfer (Lawrence & Keast, 1990; G. D. F. Wilson, 1991). The location of the two penes varies from the coxae of the last pereopod pair (in Phreatoicidea) over the sternum of pereonite 7 (in most isopods) to the pleotelson (in Valvifera) (G. D. F. Wilson, 1991). Another outstanding feature of Isopoda is their biphasic moult, during which they first shed the posterior half of their body, followed by the anterior half a few hours to a few days later (Carpenter, 2021; Tait, 1917). A characteristic of adult females – the marsupium – is formed during a reproductive moult (Warburg, 1993). Such a ventral brood pouch is an aspect all Peracarida have in common. It is formed by thin, broad, medially overlapping oostegites (Ax, 2000). The number of oostegite pairs varies between species. In addition or instead of such lamellar plates, some isopods form the marsupium from anterior and/or posterior pockets. Further, a few species brood their eggs in internal pouches (Harrison, 1984).

Given the highly diverse ecology of isopods, many modifications to the general body plan can be found within the group. In cave-dwelling and deep-sea species, too, adaptations not only include the loss of eyes but often also more elongated and slender appendages equipped with many mechanoreceptory setae and chemosensors for orientation in their dark environment (Botosaneanu, 2001). A parasitic lifestyle also makes several adaptations necessary. Epicarideans are the most highly modified isopods and often show little resemblance to free-living forms, with females being hardly more than sacks of eggs (Lawrence & Keast, 1990; Williams & Boyko, 2012). Hence, sexual dimorphism can be very pronounced in parasitic groups. Gnathiidae display very different sexes, with rather slender males with large cephalons and protruding mandibles, and unimposing, juvenile-like females greatly enlarged with eggs

(Chong et al., 2015). In many free-living isopods the only notable difference between sexes is usually a size dimorphism – much less pronounced than in parasitic forms – often with males being larger than females (Ayari et al., 2021; Brook et al., 1994; Kitaura & Nunomura, 2019), as well as the other way round (Thiel, 1999). The size range within the whole order extends from less than 1 mm to the largest confirmed record of a giant isopod at 421 mm (Soto & Mincarone, 2001). However, the majority of isopods range between 3 and 20 mm (Poore & Bruce, 2012). The group also shows a striking diversity in surface ornamentation, with spines, setae, microscales, pits, tubercles, tricorns and papillae being common features (Powell & Halcrow, 1982). Overall, most isopods are cryptically coloured or patterned (Poore & Bruce, 2012). Even strong colouration like striking reds or greens in some herbivorous marine isopods aids camouflage by matching the algae these isopods feed on. A few species can have several colour morphs and, depending on the species' life history may have the ability to change colour with the next moult cycle (Lee, 1966; Lee & Gilchrist, 1972). With regards to a subterranean lifestyle in many freshwater and terrestrial isopods, depigmentation and the absence of surface ornamentation is common (Botosaneanu, 2001).

2.3. Feeding modes and life history

Keeping in line with the overall high diversity within the order, members of Isopoda display an abundance of feeding methods. Generally speaking, isopods can be considered as omnivores. Different groups specialise in different feeding strategies like filter feeding, detritus feeding, grazing, carnivory or feeding on the body fluids and tissue of host organisms as parasites. However, many isopods feed opportunistically on food items other than their main food source. For example, Brökeland et al. (2010) described the diet of four deep-sea asellote species. The most frequent food items found in gut contents were mineral particles and mucous material, but diatoms, foraminifers and their faecal pellets were also abundant. Therefore, those isopod species were classified as phytodetritus feeders. Similarly, the estuarine anthurid *Cyathura polita* (Stimpson, 1855) is known as a detritus feeder that includes diatoms in its diet. Furthermore, in laboratory settings, it has been observed preying on smaller crustaceans and scavenging on dead fish (W. D. Burbanck, 1962). In the scientific literature, many examples can be found of not only predatory but also herbivorous isopods opportunistically scavenging on carrion (e.g., Johnson, 1976; Paris, 1963). On the other hand, numerous species are primarily characterised as scavengers. In the deep sea, for instance, isopods are frequently encountered at natural as well as artificial food falls of various stages (Linse et al., 2014; McClain et al., 2019). Common food items of marine predators are polychaetes, amphipods and small bivalves (Ejdung & Elmgren, 2001; Johnson, 1976), or larger prey like fish in the case of micropredators that attach temporarily to their "host" to feed on tissue and body fluids (Delaney & Brusca, 1985). In some species, cannibalism has been observed as well. While feeding on conspecifics is quite common in laboratory settings, it occurs on smaller scales in the field. In natural settings, adults may prey on mancas or small juveniles, which in turn actively avoid the cannibalistic adults by occupying distinct microhabitats (Jormalainen & Shuster, 1997; Leonardsson, 1991). Apart from this, wood-boring limnoriid species ingest the scraped-off plant material, while boring sphaeromatids do not and are considered filter-feeders (Si et al., 2002). Most terrestrial isopods are decomposers that predominantly feed on decaying plant litter on the soil surface (Paris, 1963; Zimmer, 2002). Only a few species climb up plants to feed on living leaves (Glazier & Kleynhans, 2015) or plant seeds (Saska, 2008). Decaying leaf litter has already been colonised by microbes and fungi, which makes it more palatable and easier to digest (Zimmer, 2002). To additionally enhance nutrient assimilation, terrestrial isopods are known to practice coprophagy (Paris, 1963; Wieser, 1978; Zimmer, 2002). Horváthová and Bauchinger (2019) have shown that terrestrial isopods do not primarily rely on the cellulose content of leaves but on the biofilm associated with them. Hence, they classified this feeding type as biofilmivory. Biofilmivory, mostly along with detritus feeding, is also a prevalent feeding method in cave species (Brad et al., 2021; Ercoli et al., 2019). Moreover, Lindquist et al. (2005) reported on the association between marine isopods and episymbiotic microbes (mainly cyanobacteria) in coral reefs of Papua New Guinea. The isopods appear to farm their symbionts by actively exposing themselves on sun-lit substrates. The photosymbionts serve as a food source for the isopods and act as a defence mechanism by producing chemicals repulsive to fish. Therefore, they allow the isopods to be active during daylight (when more predators are active, too), which increases foraging opportunities and decreases competition with other nocturnal isopod species.

In addition to all the above-discussed feeding strategies, there is the parasitic feeding mode. The evolutionary origin of the parasitic lifestyle from scavenging or predatory feeding modes in several isopod families is still debated. Nagler et al. (2017) hypothesised that parasitism within isopods arose only once from a scavenging lifestyle in an early cirolanid-like ancestor. Subsequent diversification of parasitic strategies is proposed to have followed independent evolution within several clades, during which in some lineages a change of hosts from fishes to crustaceans occurred (Dreyer & Wägele, 2001; Ketmaier et al., 2008). In brief, species within Cymothoidae and Gnathiidae are ectoparasites of fishes (Cohen & Poore, 1994; Smit et al., 2014), and epicarideans are parasites of crustaceans (Williams & Boyko, 2012). Most Epicaridea are ectoparasitic as well. However, some species are endoparasites. While some parasites are highly host-specific, others have a larger pool of hosts. For instance, sequencing of blood meals from gnathiid isopods in the Caribbean revealed 70 host fish species in 27 families (Hendrick et al., 2023). However, this study also showed that even with such low host specificity, these gnathiids still have detectable host preferences, with fish species from the families Haemulidae and Lutjanidae exploited more frequently than others. There are also examples in the scientific literature of accidental infections of unusual hosts, like cephalopods or sea snakes (Pascual et al., 2002; Saravanakumar et al., 2012). Furthermore, in a few cases, isopods are parasitic on other parasites, like barnacles or bopyroids, which is termed hyperparasitism (Oanh & Boyko, 2020; van As, 2019). Usually, the parasite load of a host individual of bopyrid or cymothoid parasites is restricted to a single infestation by a female and accompanying male/s. Double infestations are rare but do occur sometimes (Cericola & Williams, 2015; Corral et al., 2019). However, Aneesh et al. (2022) reported an unusual case of simultaneous double infestation of individual host fishes by two different cymothoid species – a surface-attaching species and a buccal-infesting one.

A particularly interesting parasite-host relationship is the one between *Cymothoa exigua* Schioedte & Meinert, 1884 and its fish hosts. The feeding activity of this cymothoid isopod causes the degeneration of the host's tongue, which is then functionally replaced by the isopod itself (Brusca & Gilligan, 1983). While in this case, the host fishes appear to be in otherwise good condition, in various instances, isopod parasites can have detrimental effects on their hosts when they act as parasitic castrators. Infected hosts do not only show a reduction in growth rate and biomass, but the parasites also target their gonads or otherwise reduce their reproductive output, i.e. through feminisation of host males (modification of external secondary sex characters) (Beck, 1980; Corral et al., 2019; Fogelman et al., 2009). Many free-living isopods have to deal with their own parasites, too. For example, several, mainly terrestrial isopod species are commonly infected with *Wolbachia* strains. These maternally inherited proteobacteria cause functional feminisation of infected males (Bouchon et al., 1998). Other parasites, too, compromise the reproductive output of their isopod hosts. Larval acanthocephalans, for instance, have been shown to prevent female freshwater isopods from attaining sexual maturity (Kakizaki et al., 2003). Furthermore, an epicaridean isopod that has been found in the brood pouch of an idoteid isopod causes breeding failure by destroying the entire brood (Salemaa, 1986). Other more or less commonly encountered parasites of isopods are nematodes (Poinar Jr., 1981; Rusconi et al., 2023), copepods (Kakui et al., 2023) and nematomorphs (Kakui et al., 2021). Apart from this, a very well-known case of parasitism in terrestrial isopods is an iridovirus infection that induces a colour change to an iridescent blue-violet along with behavioural changes like slower movements and responses, as well as reduced food consumption (Lupetti et al., 2013).

Moving onward to the topics of life cycles and reproduction, isopods also display an array of diverse strategies in these areas. Mating strategies and mating systems vary widely from gonochorism to sequential hermaphroditism and from social monogamy to polygamy and promiscuity. While the majority of isopods are gonochoristic (reproducing as either a male or a female throughout their lifetime), protogynous, as well as protandrous sex change is not uncommon, especially in parasitic isopods (Aneesh & Kappalli, 2020; Brook et al., 1994; M. P. Burbanck & Burbanck, 1974; Tsai et al., 1999). Furthermore, for several parasitic isopod species a socially monogamous mating system has been suggested, for instance, for the bopyrid *Robinione overstreeti* (Adkinson & Heard, 1995) in which pairs that are closely matched in body size share the same host individual over long periods of time (Bortolini Rosales et al., 2021). In the desert isopod *Hemilepistus reaumurii* (H. Milne-Edwards, 1840), which shows highly developed social behaviour, adults form lifelong monogamous cooperative pairs and later, with their offspring, strictly closed family communities (Linsenmair, 1984). Gnathiidae, on the other hand, practice polygamy. Hayashi et al. (2020) give an example of a polygynous mating system in *Caecognathia* sp., where males mate with several females, but a female mates only once with a single male. Within Gnathiidae it is not uncommon for males to form harems of up to 43 females depending on the species (Tanaka, 2007, and references therein). In one such harem-forming species, Tanaka and Nishi (2011) noticed an alternative male form that was often found amidst the females in a harem. These smaller males have inconspicuous mandibles and are thought to be sneaker males. Similarly, Shuster (1987) described alternative male forms in the sphaeromatid *Paracerceis sculpta* (Holmes, 1904). He termed them alpha-, beta- and gamma-males and noted that all morphs have mature sperm-producing organs. The large, ornamented alpha-males attract females and guard them in spongocoels where mating and brooding occur. Intermediate-sized beta-males resemble females and gain access to spongocoels by deceiving the bigger alpha-males. Finally, gamma-males, the smallest male form, are satellite males that resemble immature individuals but have the highest gonadal investment of the three male morphs. In terrestrial isopods, instead, promiscuity is widespread. In *Armadillidium vulgare* (Latreille, 1804), for example, where both sexes mate with several partners, a single brood could be fathered by up to 5 different males (Durand et al., 2020). Polygynandry has also been suggested as a probable mating system in marine isopods. Nakamachi and Asakura (2020) observed that reproductive aggregations of an intertidal isopod were frequently composed of several males and females, sheltering together in a barnacle shell.

Isopoda, like other peracarids, are brooders with direct development that lack a planktonic larval phase. They do not have true larvae, as their young hatch from the brood pouch as miniature versions of the adults apart from lacking the last pair of pereopods (for a detailed account of the various larval types of isopods see Boyko & Wolff, 2014; Martin, 2014). As with everything in Isopoda, the morphology of the marsupium can be quite variable (see section 2.2.), and even exceptional cases of internal brooding are known (Harrison, 1984; Klapow, 1970). After release from the marsupium, isopods develop generally over three manca stages (Montesanto et al., 2012), increasing their size with each moult (Carpenter, 2021). The seventh pair of pereopods is fully developed after the moult from the third manca to a juvenile stage, and further moults are required to develop the secondary sexual characters of adults (Boyko & Wolff, 2014). Depending on the species and environmental conditions, breeding in isopods can either be seasonal (e.g., Ayari et al., 2021; R. J. Ellis, 1971; Kitaura & Nunomura, 2019) or occur throughout the year (e.g., Boos et al., 2021; Carpenter, 2021; Johnson, 1976). Lifespans vary from only several months up to more than 20 years in the case of the cave-dwelling species *Bahalana geracei* Carpenter, 1981 (Carpenter, 2021). Female isopods may be either semelparous (producing only one brood during their lifetime), or iteroparous (having multiple broods before they die). For example, females of the desert isopod *Hemilepistus reaumurii* (H. Milne-Edwards, 1840) produce a single brood within their 14-month-lifespan (Ayari et al., 2021), whereas females of the fish parasite *Anilocra pomacentri* Bruce, 1987, which have a similar life expectancy, can produce three broods (Adlard & Lester, 1995). Brood size varies widely from only 1-4 eggs in an anthurid species (Kensley, 1984) to several thousand in parasitic isopods (Bortolini Rosales et al., 2021; Cericola & Williams, 2015) and is positively correlated with female body size (e.g., Adlard & Lester, 1995; Brook et al., 1994; Carpenter, 2021). Brooding in itself is already a form of parental care, and in several species it has been observed that females create an aerating water current through the marsupium with their maxillipeds (Cericola & Williams, 2015; R. J. Ellis, 1971; Harrison, 1984). However, some isopods even provide extended parental care to their offspring, usually through cohabitating within the parents' shelter (Kitaura & Nunomura, 2019; Thiel, 1999, 2003; Zaixso et al., 2009), or in the case of a social isopod species through providing food and offering protection for weeks after hatching (Linsenmair, 1984). Many isopod species are also known to show precopulatory mate guarding behaviour (e.g., Jormalainen et al., 2000; Kitaura & Nunomura, 2019; Shuster, 1981). Males of the freshwater asellid *Caecidotea tomalensis* (Harford, 1877) seize mature females and carry them until they have completed their posterior moult, at which point the male inseminates the female and releases it (R. J. Ellis, 1971). Another quite peculiar case of mate guarding has been observed in the janirid *Iais pubescens* (Dana, 1853) (Thiel, 2002). In this species, males manipulate the marsupium of adult females about to release their young to obtain virgin juveniles. The juveniles are then carried by the adult males for approximately seven days until they moult and are then fertilised and released. It is assumed that mate guarding evolved as a response to short female receptivity to copulation. The marsupium forms during the reproductive moult and males can only pass sperm into the marsupium as long as the exoskeleton is still soft, which leaves only a short time window for copulation. In a semelparous *Caecognathia* species, where females only have one chance for mating, female larvae can prolong their larval phase if adult males are absent (Hayashi et al., 2020). Contrary to aquatic isopods, oniscideans usually do not practice mate guarding. Zimmer (2001) assumes that this is due to the loss of temporal restrictions to female receptivity since copulation can occur throughout the entire moult cycle, and therefore, costs outweigh the gains.

2.4. Habitat

Isopods are ubiquitous. They thrive in marine, brackish and freshwater environments, as well as in terrestrial habitats. More than half of all described species are marine and can be found from the intertidal zone to the deep sea. However, shallow coastal waters harbour the highest diversity of marine isopods (Poore & Bruce, 2012). Isopods inhabit soft sediments, where they frequently burrow into the sediment (Hessler & Strömberg, 1989), with some species having a completely interstitial lifestyle (W. D. Burbanck, 1962), as well as hard substrates, on which isopods tend to shelter in crevices and cracks, and – especially during low tide in the intertidal zone – hide under rocks to avoid predation (Johnson, 1976). Herbivorous aquatic isopods live on macroalgae that are also their food source (Lee & Gilchrist, 1972). In general, isopods are a cryptic component of ecosystems, often inhabiting substrates or associating with fauna and flora that offer protection. For instance, harem-forming gnathiids inhabit sponges (Shuster,

1987) or polychaete tubes (Tanaka & Nishi, 2011), while aggregations of the intertidal isopod *Dynoides dentisinus* Shen, 1929 can be found sheltering in barnacle shells (Nakamachi & Asakura, 2020). Other isopod species can sometimes be found firmly attached to the external tube wall of Ceriantharia, tube-dwelling anemones, surrounded by ptychocyst filaments and safely out of reach of the anemone's tentacles (Ceriello et al., 2020). Furthermore, Glynn (1968) reported on cases of facultative commensalism between sphaeromatid isopods and chitons. The isopods usually occurred along the pallial groove of the chitons, intercepting food scraps that the host did not ingest. Similarly, the idoteid isopod *Edotia doellojuradoi* Giambiagi, 1925 has been reported in association with a mussel (Zaixso et al., 2009). However, this once as commensalism regarded relationship is now assumed to be a case of parasitism since only mussels infested with isopods had gill damage and reduced weight. Another idoteid isopod, *Synidotea variegata* Collinge, 1917, lives as a symbiont on a sea urchin (Yesudas et al., 2021). Moreover, several marine isopods are known to live in and on scyphozoans. One such example is the deep-sea isopod *Anuropus* sp., which inhabits the bell cavity of its host (Barham & Pickwell, 1969); another is the sphaeromatid *Cymodoce gaimardii* (H. Milne-Edwards, 1840) that attaches to the external surface of the jellyfish (Browne et al., 2017). In both cases, it is not clear whether the symbiosis might be of a parasitic nature. As already mentioned above, there are groups of obligate parasites within Isopoda that infect either fish (Smit et al., 2014) or crustacean hosts (Williams & Boyko, 2012). Most of these are external parasites attaching to the skin or appendages or within the buccal and branchial cavities. Only a few, like the Entoniscidae, are endoparasitic and live in the body cavities of their hosts. While isopods are generally benthic organisms, some epicaridean larvae are sometimes observed as rare components of the zooplankton community, where they search for their intermediate copepod hosts (Williams et al., 2022). However, those larvae still tended to be found in the highest abundances near the bottom. Isopods are also some of the most abundant organisms in the deep sea, where the macrobenthic communities are often dominated by asellotes (O'Hara et al., 2020). They occur down to hadal depths in the various trenches of the world's oceans (Kniesz et al., 2018, and references therein). The deepest recorded isopod species, *Macrostylis mariana* Mezhov, 1993, was discovered in the Mariana Trench at depths below 10,000m (Mezhov, 1993). Other rather patchily distributed habitats in which isopods have been documented are seamounts (Svavarsson, 2006) and hydrothermal vent biotopes, both in shallow (Kamenev et al., 1993) and deep waters (Malyutina & Golovan, 2022). All these examples illustrate again how diverse Isopoda are and that they successfully evolved under a multitude of environmental conditions. Furthermore, many isopod species can tolerate a wide range of abiotic conditions;

for example, a lot are euryhaline and are adapted to live in brackish habitats like estuaries (Newman et al., 2007) or inland salt lakes (P. Ellis & Williams, 1970) where they experience a wide range of salinities. Some terrestrial species are adapted to live in similarly abiotically stressful habitats, like salt marshes (Dias et al., 2005). Also, a great variety of isopods inhabit subterranean habitats like caves and groundwater environments (e.g., Botosaneanu, 2001; Brad et al., 2021). Epigean freshwater species can be encountered in lotic as well as lentic habitats (G. D. F. Wilson, 2008a). Moreover, as the most successful group of truly terrestrial crustaceans, Isopoda, specifically the Oniscidea, have colonised all imaginable land habitats from the littoral zone (Campos-Filho et al., 2018) to woodlands (Achouri et al., 2008), grasslands (Paris, 1963), urban areas (Vilisics et al., 2007), and even deserts (Ayari et al., 2021) as an important component of the soil macrofauna. Additionally, a few terrestrial species are known to be myrmecophiles and live as symbionts in ant nests (Parmentier et al., 2017)

2.5. Biogeography

As already established in the previous section, members of the order Isopoda can be found almost everywhere on Earth, with terrestrial Antarctica being the notable exception due to its present-day inhospitable conditions. Marine isopods are prevalent from the intertidal zone over sun-lit, shallow coastal waters to the deepest, lightless areas of the world's oceans. Non-marine isopods have colonised nearly every continent and can be found on continental as well as oceanic islands. They display remarkable adaptability, thriving in diverse habitats both above and below ground, and occur at various altitudes, from the shoreline to considerable elevations within mountain ranges. The current biogeographic distribution of any taxonomic group is shaped by a complex interplay of factors, such as the region's geological history, the evolutionary history of the taxon in question, and a variety of abiotic and biotic aspects. In the scientific literature, many studies document the biogeography of isopods and provide hypotheses about how those patterns arose. However, the spatial and taxonomic scales of these surveys vary widely. Some studies only investigate isopod distributions on local or small regional scales (Doti et al., 2020; Janssen et al., 2019) or focus on small taxonomic units like a specific species (Elsner et al., 2013; Recuero & Rodríguez-Flores, 2019) or a single genus (Malyutina et al., 2018; Riseman & Brusca, 2002). Nonetheless, the majority of biogeographic research deals with broad regional to continental/ocean basin scales (Bruce, 1986; Castelló et al., 2020; Kensley, 2001), either focusing on the whole isopod community within the general region (Brusca, 1987; Menzies & Glynn, 1968) or concentrating on ecological or taxonomic groups (Bruce, 1986; Brusca & Wallerstein, 1979; G. D. F. Wilson, 2008b). Only a few studies have documented the distribution of species-rich isopod families on a global scale (Cohen & Poore, 1994; Delaney, 1989; Markham, 1986).

Isopods, being predominantly benthic brooders with direct development, are considered weak dispersers, therefore endemism is expected to be high. For instance, 87% of endemism is reported for Southern Ocean isopods (Brandt, De Broyer, et al., 2007) and a similar percentage (86%) for Australian cirolanids (Bruce, 1986). Lower but still high endemism has been observed within isopods from the Galapagos region (51%) (Brusca, 1987). Moreover, in the Indian Ocean, between 46% to 79% of species in various subregions have been characterised as endemic (Kensley, 2001). Biogeographic studies with relatively big spatial scales frequently find that isopod communities of different regions tend to form well-separated clusters (Brandt et al., 2012; del Carmen Espinosa-Pérez & Hendrickx, 2006). The Southern Ocean, for instance, appears to be inhabited by a diverse and distinct isopod fauna (Brandt, De Broyer, et al., 2007; Brandt et al., 1999; Held, 2000), which probably evolved due to biogeographic isolation promoted by the Antarctic Circumpolar Current (Barker et al., 2007; Crame, 1999). In contrast, other regions like the Mediterranean Sea or the deep Sea of Japan seem to be areas of rather low diversity, likely due to extinction events during periods of low sea level or severe anoxic conditions, respectively (Cartes & Figueroa, 2020; Elsner et al., 2013). The deep sea was long thought of as a very homogeneous and rather uninhabitable environment with limited biodiversity. However, once more efficient sampling methods became available, it was discovered that diversity in the deep sea is indeed much higher than previously believed (Hessler & Sanders, 1967). Only very few, if any, deep sea peracarids are considered to be truly widespread (Brandt et al., 2012) and ridges and trenches often form effective dispersal barriers for non-swimming species (Bober et al., 2018; Johannsen et al., 2020). Isopods have colonised the deep sea on multiple occasions (Lins et al., 2012; Raupach et al., 2009) with some deep-sea families having evolved and radiated there (Hessler et al., 1979), while others invaded the deep sea more recently from high-latitude shallow waters (Held, 2000; Kussakin, 1973).

The origin of terrestrial isopods has been dated back to pre-Pangaean times in the late Paleozoic, and phylogenetic analyses suggest that the terrestrial environment has been colonised multiple times (Broly et al., 2013; Lins et al., 2017). To successfully shift from an aquatic life to an entirely terrestrial one, Oniscidea had to evolve several morphological, physiological and behavioural adaptations (Hornung, 2011; Schmidt & Wägele, 2001). Those adaptations helped them to conquer the land, spread across almost every continent, and become the most successful group of crustaceans in the terrestrial realm. Oniscidea show a great variety of distribution patterns on different spatial scales. On a fine spatial scale, their distribution is highly connected to environmental heterogeneity (Sfenthourakis & Hornung, 2018), and desiccation resistance appears to be a key feature in shaping these distributions (Csonka et al., 2018). In parallel to the marine realm, temperature is a limiting factor, too. In a cartographic analysis of isopod distributions in the former USSR, for instance, it was observed that no isopods occurred north of the isocline of 120 days per year with temperatures above 10°C (Kuznetsova & Gongalsky, 2012). A particularly well-studied area in terms of biogeographic patterns within Oniscidea seems to be the Mediterranean region. Several studies have focused on the isopod fauna of Mediterranean islands and islets. Although similarity values between some islands are often found to be relatively high (Gentile & Argano, 2005; Triantis et al., 2008), different island groups were found to form distinct clusters (Gentile & Argano, 2005; Sfenthourakis, 1996). The observed structure reflects the interconnectivity between archipelagos and the most proximate mainland and evolutionary events acting on a local scale. Rates of endemism reported for terrestrial regions seem to be lower than in the ocean. Sfenthourakis (1996) documented 20% of oniscidean species from the central Aegean islands as endemic, a similar proportion to that of other Mediterranean archipelagos and indicative of geologically recent isolation from the mainland. Likewise, the observed percentage of endemism in the Transdanubian region of western Hungary, albeit considerably smaller than previously mentioned oceanic regions, is 16% (Hornung et al., 2008). Faunal influences from neighbouring regions show that Transdanubia is a diverse biogeographical crossroad for terrestrial isopods.

Peracarid fossils provide evidence that early ancestors of freshwater isopods had an incursion into continental ecosystems in the late Devonian, more than 360 mya (Robin et al., 2021). Further, fossils of phreatoicidean isopods show that this group was present in freshwater by the Triassic (~238 mya) and widespread on Gondwana by the Jurassic (G. D. F. Wilson, 2008b). This Gondwanan ancestry with subsequent vicariant events explains the modern biogeographical distributions of this group well. Continental drift is also the most probable explanation for the distribution of closely related freshwater microcerberids across Europe and North America (Wägele et al., 1995). Consequently, this group must be older than the Atlantic Ocean with at least a middle Cretaceous origin. Many freshwater isopods are stygobionts, living in caves, various groundwater ecosystems, or the interstitial. Aquatic hypogean environments were colonised multiple times by members of nearly all suborders (Wägele, 1990). Stygobiontic cirolanids, for example, are considered to be derived from ancestors with a widespread Tethyan distribution that were left stranded in newly developing subterranean habitats by marine transgressions and regressions (Holsinger et al., 1994). According to Wägele (1990), colonisation of subterranean aquatic biotopes occurred in two ways. He assumes that a few families entered the new environments via the coastal groundwater. Most families, however, are supposed to be derived from epigean freshwater ancestors. In the present day, many aquatic species are increasing their ranges, often with humans playing an important role as vectors in their dispersal (Kemp et al., 2020). The construction of canals, ship traffic and recreational fisheries greatly increased the connectivity of freshwater ecosystems and aided in the introduction of species to new regions.

2.6. Ecological and economic importance

Isopods are important components of healthy and functioning ecosystems. Since many isopods are scavengers or decomposers, they play an important role in nutrient recycling, both in aquatic and terrestrial ecosystems (Zimmer, 2002). They also act as bioturbators, resuspending fine soil particles, e.g., when numerous scavenging cirolanids emerge from the substratum in response to olfactory cues from carrion (Frutos & Sorbe, 2010). The giant isopod *Bathynomus giganteus* A. Milne-Edwards, 1879, too, generates considerable sediment disturbance while feeding on large carcasses (McClain et al., 2019). In the Baltic Sea, the predatory isopod *Saduria entomon* (Linnaeus, 1758) can alter the composition of the macrobenthic community by selectively preying on its preferred amphipod prey. However, when that is scarce, *S. entomon* picks the smallest size classes of a common bivalve to feed on, influencing the size distribution of the bivalve and consequently reducing intraspecific competition and decreasing population fluctuations (Ejdung & Elmgren, 2001). Isopods are themselves an important food source for an abundance of animals. Invertebrates that prey on aquatic or terrestrial isopods include insects, arachnids, centipedes, and hard corals (Brad et al., 2021; De Smedt & Henrard, 2022; Paris, 1963; Paula et al., 2021; Toft & Macías-Hernández, 2021). While predator-prey relationships with native predators usually do not threaten local isopod populations, in the case of an introduced crab in the Baltic Sea, predators can have a profound impact on naïve prey that has not yet learned an anti-predator response to the new threat (Yli-Renko et al., 2022).

Besides invertebrates, a wide variety of fish are known to include isopods in their diet (W. D. Burbanck, 1962; Kim et al., 2022; Reed et al., 2018). For example, parasitic gnathiids on coral reefs are the predominant prey items consumed by cleaner fishes (Artim et al., 2017; Grutter, 1997). Other fish species include isopods in their diet while they are still small-sized juveniles but not as fully grown adults (Fischer et al., 2022; Jacobson et al., 2019). Moreover, in California, non-native terrestrial isopods have emerged as a significant seasonal energy source in the diet of trout (Rundio & Lindley, 2021). Further vertebrates that prey on isopods include birds, reptiles, amphibians, as well as mammalian predators like shrews (W. D. Burbanck, 1962; Churchfield, 1982; García-Padrón, 2021; Lo Valvo & Pieri, 2021; Paris, 1963). Recently, it has been observed that a few isopod species can aid with the fertilisation of seaweeds and with fungal spore dispersal. Lavaut et al. (2022) have demonstrated that individuals of *Idotea balthica* (Pallas, 1772) living on red algae carry the alga's spermatia on their bodies. When the isopods moved from a male to a female alga, they significantly increased the fertilisation success of the alga. Similarly, in the terrestrial realm, *Oniscus asellus* Linnaeus, 1758 has been shown to act as a short-distance spore disperser for a truffle species (Thomas & Thomas, 2022). The fruitbodies of the truffle are a food source for terrestrial isopods, and ingested spores pass through the isopods' digestive tract and get excreted within faecal pellets up to 18 days post-feeding. In a nutshell, isopods play an essential role in food webs and are a crucial part of functioning ecosystems. However, isopods can also have adverse impacts on natural environments. When burrowing sphaeromatids occur in high densities, they have a significant bioerosive effect on the coastline, albeit localised. In Malaysia, *Sphaeroma triste* Heller, 1865 burrows into intertidal sandstone where, in some instances, it can remove between 20% to 50% of rock volume from the outcrop surface, therefore increasing the area exposed to erosion and significantly weakening the rock (Dodge-Wan & Nagarajan, 2020). In the same fashion, erosive effects of the invasive isopod *Sphaeroma quoianum* H. Milne-Edwards, 1840 alter salt marsh habitats in California. Their burrowing activities enhance sediment loss from banks and marsh edges, in some cases causing undercutting, which can lead to losses exceeding 100cm per year (Talley et al., 2001). With further degradation of the ecosystems, these bioerosive effects are only likely to increase in severity. Likewise, wood-boring isopods pose a threat to mangroves, destabilising the trees and consequently making the sediment prone to erosion. As a result of sphaeromatids burrowing into the prop roots of mangroves, the relative root growth rate is reduced by 55% (Ellison & Farnsworth, 1990). The isopod burrows provide easy access to the roots for decomposing bacteria and fungi, thus accelerating their fouling (Rehm & Humm, 1973). However, Ellison and Farnsworth (1990) also found that an epibiontic layer of certain sponge and ascidian species inhibits isopod colonisation, indirectly facilitating root growth and mitigating the destructive impact of isopods on mangrove fringes. Another instance of isopods exerting a detrimental impact on natural ecosystems involves an introduced bopyrid infecting mud shrimps in North American estuaries. The parasitic castrator *Orthione griffenis* Markham, 2004 caused population collapses and local extinctions of its previously abundant new host *Upogebia pugettensis* (Dana, 1852), a critical ecosystem engineer (Chapman et al., 2012). Consequently, the services provided to the ecosystem and the mud shrimp's dependent symbionts were greatly diminished.

Members of Isopoda are not only of ecological importance but also have economic significance. Though no isopod species are harvested commercially and reports of isopods as part of human diets are somewhat anecdotal (Poore & Bruce, 2012), isopods can profoundly impact commercial fisheries and aquaculture. As already pointed out earlier, there is a myriad of parasitic isopods and micropredators that either target fish or crustaceans. Reports of isopods attacking fish caught in traps and nets, resulting in the fish being unmarketable, are not unusual (Stepien & Brusca, 1985), and isopod infestations and attacks on cultured fish and prawns are well documented. In India, for example, bopyrid isopods are considered one of the prime threats to the emerging prawn industry. Freshwater prawns in an aquaculture facility were infested with a prevalence of overall 46.2% (Gopalakrishnan et al., 2017). Female prawns, however, had a much higher infestation rate than males and were rendered infertile by the parasite. In fish aquaculture isopods will often infest or attack juvenile stocks, reducing growth and causing deformations that will see the fish discarded (Čolak et al., 2018), or even causing up to 100% mortality within short time periods (Rajkumar et al., 2005), leading to severe economic losses (Ali et al., 2022; Sanil et al., 2009). Treatments of isopod infestation in cultured fish customarily include the use of chemicals. In this regard, deltamethrin, trichlorfon and diflubenzuron have been shown to be effective against isopods (Ali et al., 2022; Athanassopoulou et al., 2009). As an environmentally safe alternative to chemical treatment, Kavanat Beerahassan et al. (2021) suggested the use of a microbial consortium of exoskeleton-degrading bacterial strains to control outbreaks of parasitic isopods. Unlike the previously mentioned negative impacts of isopods on aquaculture, Svane and Barnett (2008) reported a positive effect of scavenging isopods at tuna farms. The scavengers were active at night and played an important role in removing waste feed from beneath the tuna farms that had sunken to the bottom. In ornamental aquaculture hatcheries, the use of the freshwater isopod *Asellus aquaticus* (Linnaeus, 1758) for biological prevention has been suggested. Since unfertilised fish eggs facilitate the spread of fungal and bacterial infections, they must be removed from the rearing tanks. Manual removal, however, is time-consuming and labour-intensive. Isopods held in the hatchery tanks feed on the microbial and fungal films that develop on dead eggs, therefore preventing the spread of infections, while at the same time posing no threat to newly hatched fish larvae (Kucska et al., 2022). Regarding another matter in aquaculture, isopod meal, which is rich in minerals and vitamins, has been proposed as an inexpensive and sustainable alternative food source for animals (Xu et al., 2021). Another group of isopods that collectively cause enormous damage and costs worldwide are the boring isopods – many Sphaeromatidae and Limnoriidae. The global damage to marine wooden infrastructure by woodborers (an umbrella term which also includes shipworms) in the early 2000s was estimated at US$ 1 billion per year (Rayes et al., 2015). Furthermore, Davidson (2012) reported widespread damage to polystyrene floats used in aquaculture facilities and the resulting microplastic pollution caused by boring isopods. He estimated that a colony of 100,000 individuals can potentially produce 490-630 million plastic particles during their burrowing activity. From a more favourable perspective, Davidson (2012) also demonstrated that floats encapsulated with a polyethylene cover, or simply a different type of float, namely extruded polystyrene, inhibit boring and, therefore, can prevent isopods from generating further microplastic pollution. Staying on the subject of pollution, isopods have long been contemplated as biomonitors for the accumulation of toxins and heavy metals in the environment. They ingest heavy metals through or with their food or by way of surface adsorption (O'Callaghan et al., 2019) and then store them in special organelles of the hepatopancreatic tissue (Zimmer, 2002), which gives them a high bioaccumulation capability. Van Der Spuy et al. (2023) showed that the marine parasite *Cinusa tetrodontis* Schioedte & Meinert, 1884 accumulated significantly higher concentrations of elements than its fish host, thereby presenting the potential to act as an early warning model organism. Apart from this, Ahadi et al. (2020) explored the potential use of terrestrial isopods in composting raw sewage sludge, which also contains many heavy metals. They demonstrated that the utilisation of isopods, especially in combination with earthworms, can significantly improve the physiochemical properties of sewage sludge, rendering it suitable for use as an agricultural fertiliser. Moreover, isopods can not only tolerate high heavy metal concentrations in the environment, but they are also highly resilient to radioactive contamination. Effects of chronic radiation exposure have been studied in isopods from freshwater lakes in the surroundings of Chernobyl. Radionuclide contamination neither impacted development (Fuller et al., 2017) nor reproductive output (Fuller et al., 2018), nor did it influence the genetic diversity of the studied isopod populations (Fuller et al., 2019). Given their resilience and their role as decomposers, isopods are generally appreciated for returning nutrients into the soil. However, if they occur in excessive numbers, they become a nuisance and are seen as pests. For example, the lack of pesticides and large amount of compost used in organic greenhouses, provide an ideal habitat for terrestrial isopods. When there is a population explosion, occasional feeding of isopods on greenhouse plants can cause serious damage (Messelink & Bloemhard, 2007). On the other hand, a favourable reduction of fungal pathogens on stored potatoes has been observed in the presence of isopods (Mészárosné Póss et al., 2022) contradicting their reputation as "storage pests". Finally, direct interactions between isopods and humans are rare. Only a few reports of isopod "attacks" on humans exist in the scientific literature. Stepien and Brusca (1985) mention that they had been bitten by cirolanids during nocturnal SCUBA dives, with the bites feeling much like horsefly bites. Similarly, Garzón-Ferreira (1990) described being attacked by another species of micropredatory isopod, which has weak host preferences, while snorkelling over seagrass meadows and patches of fire coral. He notes that after only a few minutes, a diver could have five or more individuals firmly attached to his skin and that the isopods began to feed immediately after attachment. In Australia, Tiemensma et al. (2017) documented a case of post-mortem scavenging of two cirolanid species on a human cadaver that had likely been submerged in the ocean for approximately 11.5 hours. In contrast to this destructive effect, Yue et al. (2019) consider isopods a promising source of novel medicinal compounds. They presented evidence that an extract from *Ligia (Megaligia) exotica* Roux, 1828 possesses anti-inflammatory and analgesic effects, hence supporting its use as a means for pain-relief in Chinese folk medicine.

3. Progress in the discovery of isopods – is the description rate slowing down?

3.1. Introduction

Species richness is a commonly used metric to measure biodiversity. Knowing how many different species there are in space and time is vital for all biodiversity-based research and sustainable conservation strategies. Scientists have long tried to answer the intriguing question of how many species exist on Earth. Estimates range from about 2 million species (Costello et al., 2012) to 10 million species (Grassle & Maciolek, 1992). Even numbers from "at least 1 billion to 6 billion" species have been estimated based on various assumptions like parasite-host ratios and a very high ratio of bacterial to animal species (Larsen et al., 2017). Many recent estimates of total species richness for different taxa are based on observed description rates, often from a global dataset which buffers local biases, and are of a more conservative nature (e.g., Bebber et al., 2007; Costello, 2016; Deng et al., 2016).

The first question to ask when it comes to estimating total species richness is how many species have already been described. At the beginning of this century this question was still difficult to answer. Compiling global datasets for various taxa would have been very time-consuming and tedious. The bulk of knowledge in the field of taxonomy was hidden away in large and expensive printed monographs or low-impact and regionally restricted print-only journals that could be hard to come by (Godfray, 2002). Godfray (2002) also stated that "taxonomy is made for the web" and needs to reinvent itself "as a twenty-first-century information science" where the global knowledge and achievements of the field are collected in one place and made easily accessible for everyone. Now, with the publication of continually updated databases like the Catalogue of Life (Bánki et al., 2021) and the World Register of Marine Species (S. T. Ahyong et al., 2023), which also account for some known synonymies, assessing the number of already described species is a lot easier, and many studies make use of these data (e.g., Arfianti et al., 2018; Costello et al., 2012; Mora et al., 2011; Pagès-Escolà et al., 2020; Pamungkas et al., 2019).

The rate of description of new species also depends on the number of taxonomists working towards a complete inventory of life on Earth. Some studies raised concerns that taxonomy was in crisis (Bacher, 2012; Gaston & May, 1992; Hopkins & Freckleton, 2002). While this may be true in some institutions and for some taxa, it does not apply to the global workforce. Other studies found that, in contrast to a proposed decline in the taxonomic workforce, the number of people describing new species has been increasing over recent decades (e.g., Appeltans et al., 2012; Arfianti et al., 2018; Costello, Wilson, et al., 2013; Eschmeyer et al., 2010; Songvorawit et al., 2021). However, the average number of species described per taxonomist showed a decrease (Costello et al., 2012), sometimes interpreted as a sign that it is getting harder to find new species from the shrinking pool of still undescribed species (Joppa et al., 2011b).

Isopods are a species-rich taxon of crustaceans found globally in terrestrial, marine and freshwater habitats. Based on expert opinion, Isopoda were said to be a promising taxon for tens of thousands of new species (Appeltans et al., 2012). Nevertheless, Poore and Bruce (2012) noted that the description rate of non-asellote marine isopods has slowed down since the 1990s. In a review by Williams and Boyko (2012) it was briefly mentioned that descriptions for parasitic isopods from the superfamilies Bopyroidea and Cryptoniscoidea (which were excluded from Poore and Bruce (2012)) showed two apparent peaks during the 1880–1930s and 1980–2000, while Costello (2016) found that the rate of description of parasitic isopods overall was declining since the 1990s. Previously, Costello et al. (2012) tried to predict the number of yet undescribed marine isopods based on past description rates. However, their statistical model yielded high uncertainties because the accumulation curve of species numbers still showed a steep increase and was not yet nearing an asymptote. Since these studies, many more species names have been added to WoRMS and more synonymies have been resolved. With this matured dataset of isopods available, this study examines the description rate for the whole order Isopoda, including terrestrial, marine and freshwater species, and subsets of parasitic and subterranean species. Moreover, an estimate of still undescribed isopod species is calculated by the non-homogeneous renewal process (NHRP) model after Wilson and Costello (2005). The NHRP is designed for this purpose and takes into account the variation between years to produce confidence limits around its predictions (S. P. Wilson & Costello, 2005). Additionally, indicators of taxonomic effort, such as the number of authors describing species, potentially biased by varying publication lifetimes of authors over time and changing trends in authorship practices, were analysed.

3.2. Methods

3.2.1. Data source and processing

Data including species names, authorities, the year of description and environment for the order Isopoda Latreille, 1816 were downloaded from the World Register of Marine Species (WoRMS) on 19th July 2018 (WoRMS, 2018) and updated on 20th February 2023 (WoRMS, 2023) after a delay due to the Covid-19 pandemic. All results, figures and tables in this chapter refer to the updated 2023 dataset. During the cleaning process of the update some substantial changes to the taxonomy of bopyroid and cryptoniscoid isopods, addressed in Boyko and Williams (2023), came to our attention and were incorporated into the update. Although WoRMS is predominantly a database for species that occur in marine habitats, it contains sub-registers like the World Marine, Freshwater and Terrestrial Isopod Crustaceans database. Therefore it was possible to extract data not only for marine isopods but also for freshwater and terrestrial species, allowing an analysis of the whole order Isopoda. To avoid overestimating the actual global number of isopod species, only species names listed in WoRMS as “accepted” and checked by a taxonomic editor have been included in the analysis. Moreover, only extant species and those ranked as “species” were analysed, excluding more than 30 fossil isopods and more than 500 subspecies, though their status was “accepted”. That left a species list with 10,333 entries for the 2018 dataset (Hartebrodt, 2019) and 10,687 accepted species in the updated list from 2023 (Hartebrodt, 2023b).

The data were checked for issues that may affect the analysis, and uncertainties were double-checked with WoRMS and corrected. The most common issues were misspellings and different spellings of authors’ surnames like “Magniez” and “Magneiz/Magnez/Magiez” or “Wägele” and “Waegele”. Those were corrected and only one spelling for each surname was used. In cases where different authors had the same surname, it was checked back with the original species descriptions to sort out individual authors. They were distinguished by adding their given names’ initials (e.g., E.H. Williams, J.D. Williams, and W.D. Williams). The number of taxonomists describing species over time is an indicator of taxonomic effort, which could be biased by changing authorship practices (Costello, Wilson, et al., 2013; Essl et al., 2013; Fisher et al., 2018; Joppa et al., 2011a). For the purpose of this paper, every author who published a scientific description of an isopod species is termed a “taxonomist” without any regard for the extent of his/her expertise in isopod taxonomy. In this analysis, only first authors have been considered to provide a minimum estimate of effort.

Isopods were classified as marine, freshwater or terrestrial species according to the environmental information available in WoRMS. Species inhabiting brackish environments were grouped with the marine species. In addition, subgroups of parasitic and subterranean isopods were classified from the literature. Only isopods that are obligate parasites were classified as “parasitic”. Therefore, species of Corallanidae and Aegidae, often termed as parasites, were not included since those are micropredators (Brusca, 1983a) rather than parasites by definition. In the subterranean category, stygobionts and troglobionts were included but not stygophile or troglophile isopod species because these usually have populations that live entirely aboveground.

3.2.2. Data analysis

The data were analysed in several ways to get an accurate picture of the description rate of isopod species over time. First, the cumulative number of species described per year was plotted to see whether there was a levelling out in recent years. Second, the annual number of species' descriptions was plotted to investigate the general trend of the description rate. Additionally, the non-homogeneous renewal process (NHRP) model of Wilson and Costello (2005) was used to make predictions about future discoveries. The model not only extrapolates the rate of description but also takes into account that description rates differ over time. It was used to estimate numbers on how many isopod species might be described by the years 2050 and 2100 with a 95% confidence interval. The equation used by the NHRP model is the following:

$$t = \frac{N}{1 + \exp\left(-\beta(t - \alpha)\right)}$$

Here **t** is the number of isopod species described by a particular year; **N** is the total number of species to be described; **β** stands for the overall rate of description, and **α** is the year of the maximum rate of description. A larger β implies a faster rate of description.

To estimate taxonomic effort, the number of first authors per year was plotted. Furthermore, the average number of species described per number of authors in a year was analysed over time. To determine the breakpoint from whereon the yearly average number of species described per author started to decline, a piecewise regression analysis was performed in R version 4.1.1 (R Core Team, 2021) using the "Segmented" package (Muggeo, 2008).

The publication lifetime of first authors was calculated as the number of years from an author's first description of an isopod species to their most recent. Decreasing lengths of publication lifetimes might suggest a decrease of taxonomists specialised in isopods. To examine whether there was a change in the span of authors' publication lifetime, linear regressions of publication lifetime against the year of an author's first species description were performed. Also, linear regressions on publication lifetime against the average number of species described by each author per year were performed to examine whether it has a significant effect on productivity. The regressions were done for all authors, once including and once excluding Vanhöffen, who published the descriptions of all 67 species he described in one extensive monograph resulting in a publication lifetime of only one year.

Authorship practices change over time and might bias the overall estimate of taxonomic effort. Over the years, there might be a trend toward multi-authored species descriptions, termed the “et al.” effect. During the analysis, the number of descriptions with multiple authors was counted, as well as the number of descriptions that had only a single author. Both were plotted per decade to compare them. The number of one-time authors, who described only a single isopod species, was also counted and was plotted as a proportion of all species descriptions per decade.

3.3. Results

3.3.1. Species diversity

Between 1758 and 2023 a total of 10,687 extant isopod species have been described by a cohort of 1,144 authors (755 first authors). Of the first authors analysed here, 282 were one-time authors who described only a single isopod species. The 21 most prolific authors, each describing more than a hundred species, together described about 43% of all accepted species (see Table A1). More than half of all named species are marine species — 6,151 in number. Isopods are the most species-rich crustaceans on land, with 3,840 terrestrial isopod species and 696 freshwater species. Approximately 14% of all species are obligate parasites, and 9% can be categorised as subterranean (i.e., cave-dwellers, groundwater species, inhabitants of interstitial spaces). A list of all subterranean species can be found in Appendix D, and an annotated list of parasitic isopod species is provided in Appendix E. The order Isopoda consists of 12 suborders comprising 141 families and 1,557 genera. The most species-rich genera, each containing over 100 species, are *Porcellio*, *Armadillidium, Cirolana*, *Gnathia*, *Venezillo*, *Proasellus* and *Trichoniscus*. The most species-rich isopod families are Sphaeromatidae, Armadillidae and Bopyridae (Table 3.1). At the other end of species richness, there are 15 monotypic families, which have only one genus containing a single species.

Table 3.1. A list of the 32 most species-rich families, each with more than 100 species. Families are ranked by the number of species. The percentage of species described within a family by certain time points is given.

Family	Number of genera	Number of species	First species described	Last species described	% of species described by			
					1850	1900	1950	2000
Sphaeromatidae	100	664	1787	2021	6	17	45	89
Armadillidae	82	647	1816	2023	1	14	60	94
Bopyridae	170	639	1798	2023	1	9	46	83
Cirolanidae	63	525	1804	2023	1	10	25	77
Trichoniscidae	87	524	1818	2023	1	4	46	89
Philosciidae	112	508	1763	2023	1	5	27	83
Cymothoidae	45	384	1758	2023	9	38	50	85
Munnopsidae	43	342	1861	2022	0	11	26	81
Asellidae	19	333	1758	2022	1	5	26	90
Porcellionidae	19	330	1804	2023	7	35	72	96
Anthuridae	26	309	1808	2022	1	3	12	89
Armadillidiidae	18	272	1798	2023	5	19	58	84
Eubelidae	50	257	1873	2018	0	14	53	94
Gnathiidae	12	237	1804	2023	1	8	32	72
Idoteidae	24	190	1766	2017	13	32	57	93
Agnaridae	14	189	1771	2022	3	9	44	77
Paramunnidae	45	185	1864	2022	0	4	21	46
Janiridae	22	175	1814	2022	2	11	38	94
Arcturidae	14	161	1806	2021	2	14	48	83
Aegidae	8	149	1758	2023	8	35	56	73
Desmosomatidae	20	145	1864	2020	0	6	19	75
Platyarthridae	8	136	1833	2021	1	9	44	83
Haploniscidae	8	125	1877	2017	0	1	7	74
Trachelipodidae	8	125	1833	2017	3	16	56	90
Styloniscidae	17	124	1853	2022	0	4	35	69
Munnidae	6	114	1839	2023	3	10	36	91
Ligiidae	6	113	1767	2022	7	20	54	78
Ischnomesidae	9	109	1866	2019	0	6	22	80
Scleropactidae	26	108	1854	2021	0	10	34	77
Serolidae	22	107	1775	2015	4	21	36	80
Antarcturidae	18	106	1881	2022	0	10	34	89
Leptanthuridae	14	105	1853	2021	0	9	21	93

The first 100 years of discovery after the publication of Linnaeus' *Systema Naturae* in 1758, in which the first seven still valid isopod species were described, yielded relatively few species. Until the end of the 18th century an average of only 6 species were described per decade. The following 50 years saw, on average, 43 species descriptions per decade, many of which were contributed by the three most prolific taxonomists of that time. Leach described 30 species between 1814 and 1818; J.F. Brandt contributed 37 species descriptions between 1831 and 1841; and H. Milne-Edwards added 34 new species in 1840, at which point the overall number of named isopod species had climbed to 194. For a detailed history of the discovery of marine isopods see Poore and Bruce (2012). From the 1850s to the end of the 19th century the average number of new species per decade climbed to 209. Descriptions of new isopod species started to accumulate faster, and after the 1880s the rate increased swiftly and steadily up to the 1970s, when the slope of the curve got even steeper (Fig. 3.1a). The terrestrial subgroup follows this overall pattern very closely (Fig. 3.1c), whereas for marine isopods the cumulative number of species seemed to plateau for short periods of time in the 1890s and the mid-20th century, before resuming a steep increase after the 1960s (Fig. 3.1b). A dip in descriptions during World War II and its aftermath is clearly visible in almost all groups (Fig. 3.2). Only freshwater isopods show a small peak in species descriptions during that time, largely due to Nicholls' work, who published 36 descriptions of freshwater isopods in 1943 and 1944 (Fig. 3.2b). Besides having far lower species numbers than marine isopods, discoveries of freshwater species stayed low until the 1880s (Fig. 3.1b). The discovery of subterranean species started later, and most were discovered after the 1950s (Fig 3.1d).

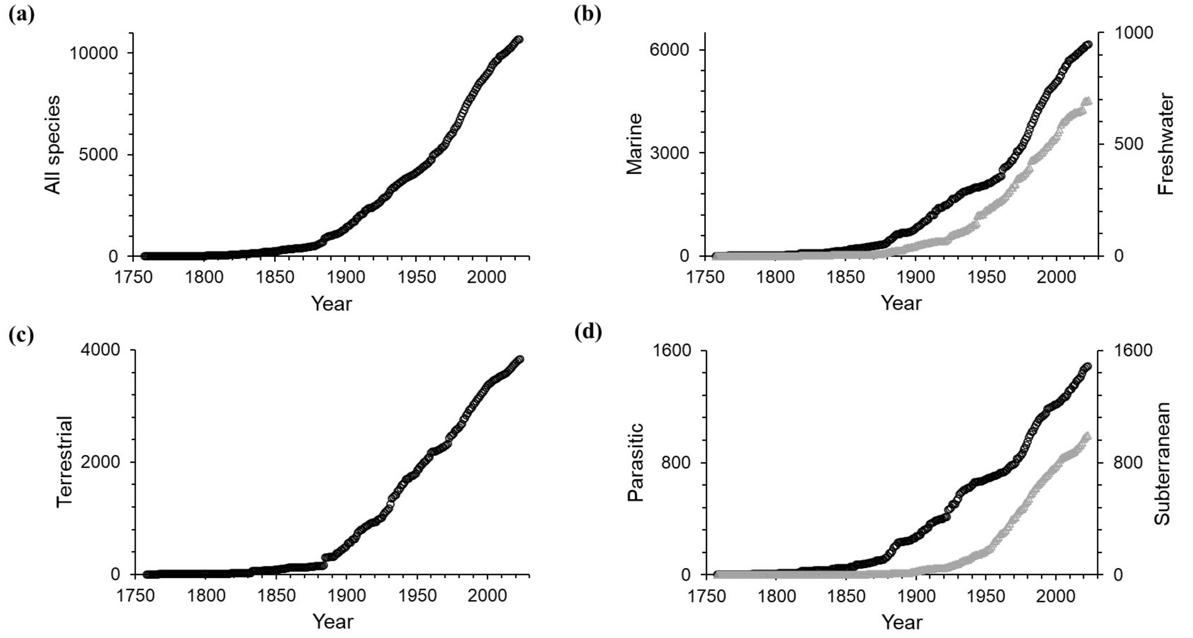

Figure 3.1. Cumulative numbers of isopod species described per year. **(a)** all isopods, **(b)** marine (black circles) and freshwater (grey triangles), **(c)** terrestrial and **(d)** parasitic (black circles) and subterranean (grey triangles). Note that the scales vary.

Isopods showed a peak of discovery in the late 20th century, with an all-time high of 200 species described in the year 1982 (Fig. 3.2a). Most subgroups peaked during the same period, except for freshwater isopods, which had their highest peak at the beginning of the 21st century and terrestrial species having their main peak earlier in the 1930s (Figs. 3.2b-d). In the past three decades the number of species described per year has decreased notably in overall species descriptions and specifically marine isopods. Yearly descriptions of freshwater isopods are generally low, although 2020 was a record year that saw 34 freshwater species described. This was more than 10-times the average of the previous 10 years. On average one third of yearly descriptions over the past 10 years were parasitic and subterranean species.

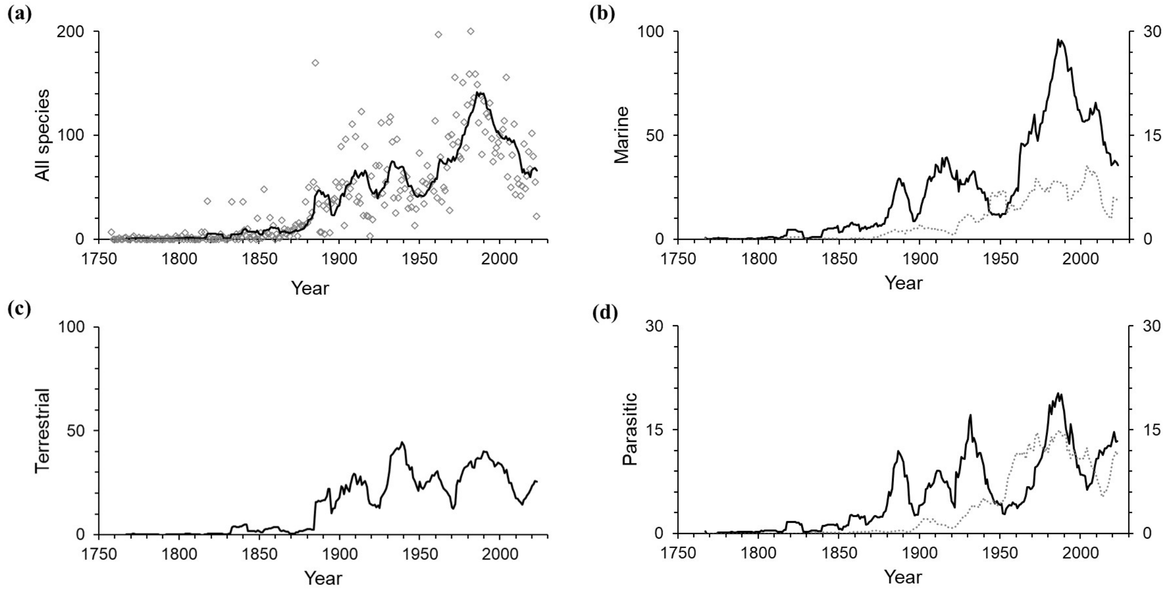

Figure 3.2. The number of isopod species described per year. **(a)** all isopods, **(b)** marine (solid line) and freshwater (dotted line), **(c)** terrestrial and **(d)** parasitic (solid line) and subterranean (dotted line). The lines are 10-year moving averages. Note that the scales vary.

3.3.2. Predictions of yet to be named species

The NHRP model predicted another 470 isopod species to be described by the year 2050 with a 95% confidence interval of 390 to 560 (Fig. 3.3a). Until 2100 a total of 660 (540–810) species were predicted to await scientific description, assuming the pace of description continues at its current rate. This would bring the cumulative number of isopod species up to 11,347 in 2100 (Fig. 3.3a). When split into subgroups, estimates from the model show that most of the future discoveries could be expected in marine and terrestrial environments, and only a small part will be from freshwaters (Fig. 3.3b).

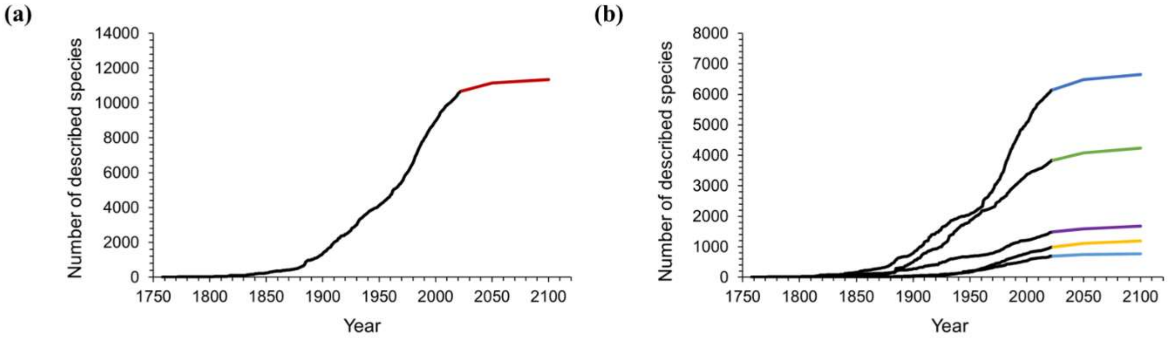

Figure 3.3. The observed and predicted cumulative number of isopod species described over time. **(a)** Observed (black line) and predicted (red line) cumulative number of all isopod species. **(b)** The observed (black lines) and predicted cumulative numbers of species within the subgroups (dark blue: marine; green: terrestrial; purple: parasitic; yellow: subterranean; and light blue: freshwater isopods).

3.3.3. Taxonomic effort

Since the first scientific description of an isopod species by Linnaeus, 755 first authors have described the species known today. Over time the number of first authors per year has increased. Since the 1950s there were more than three times as many authors involved in isopod taxonomy as during the first half of the 20th century (Fig. 3.4). This pattern can be seen in almost all subgroups (Fig. A1). However, the average number of species described per author has been declining over the last century (Fig. 3.4). Nevertheless, the overall trend sees many more taxonomists describing fewer species. A piecewise regression analysis found the breakpoint in the data series to be in 1916, whether zero values were excluded or not (Fig. 3.5). Since then, the average number of species described per authors active in the same year has declined.

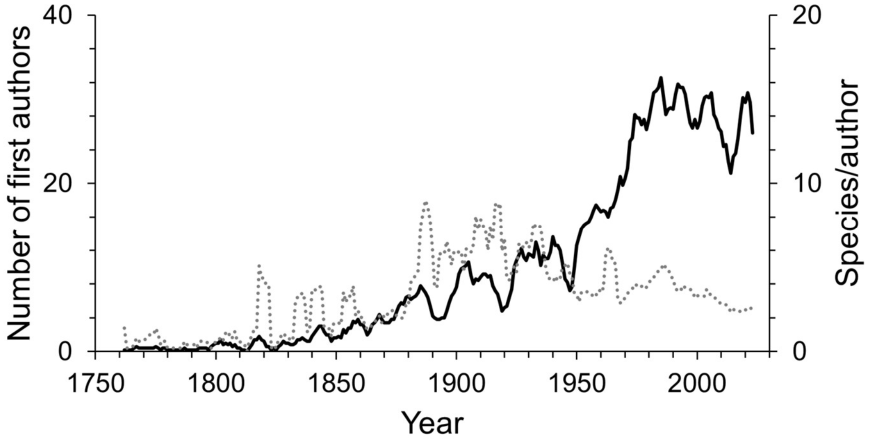

Figure 3.4. The number of first authors per year (solid line) and the average number of species described per author per year (dotted line). The lines are 5-year moving averages.

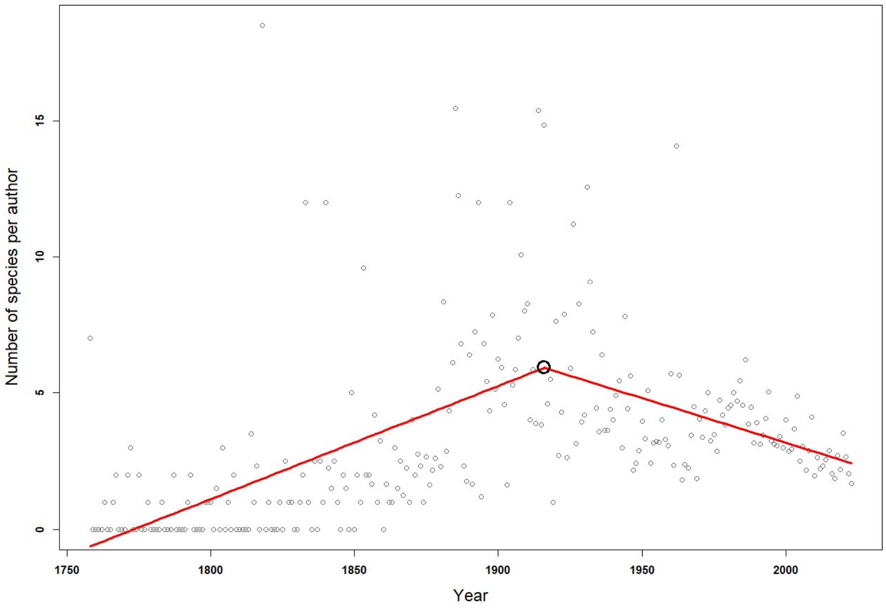

Figure 3.5. Breakpoint analysis for the average number of species described per number of authors in any given year. The red line is a fitted broken-line of the segmented model. The black circle indicates the breakpoint in 1916.

The average publication lifetime of an author was found to be 8.4 years, with 30% of authors ranking above the average. Although a linear regression shows a weak decreasing trend ($R^2 = 0.006$, $P < 0.05$) in publication lifetime over the years (Fig. A2a), this change was not significant ($R^2 = 0.00004$, $P = 0.88$) when data of authors who started publishing after 2010 were excluded (Fig. A2c) because these authors may still be publishing in the future. Again, a weak decreasing trend of publication lifetime ($R^2 = 0.01$, $P < 0.05$) could be detected when all one-time authors were excluded from the linear regression analysis (Fig. A2b), but this trend was again not significant ($R^2 = 0.0002$, $P = 0.77$) when data for authors who started publishing after 2010 were also excluded (Fig. A2d). Furthermore, there was no significant evidence ($P > 0.05$) for a change in productivity over time, whether Vanhöffen was included (Fig. A3a) or excluded (Fig. A3b).

Multi-authored descriptions became more abundant during the late 19th century but stayed relatively low until the late 1960s (Fig. 3.6). Since the beginning of the 21st century multi-authored descriptions outnumbered the number of species described by a sole author (Fig. 3.7), peaking at a proportion of about 70% of new species descriptions during the 2010s and slightly over 90% within the first three years of the current decade (Fig. A4b). In contrast, the number of descriptions published by one-time authors is negligible (Fig. 3.6). Their proportions were high in the early history of isopod discovery (Fig. A4a) when the overall number of descriptions was low. However, since the late 19th century, the contribution of one-time authors to isopod taxonomy has been small. During this time span, the highest proportion of one-time authors was found in the current decade with close to 7% (Fig. A4a). In the last “full” decade , the 2010s, the proportion of descriptions by one-time authors was about 5%.

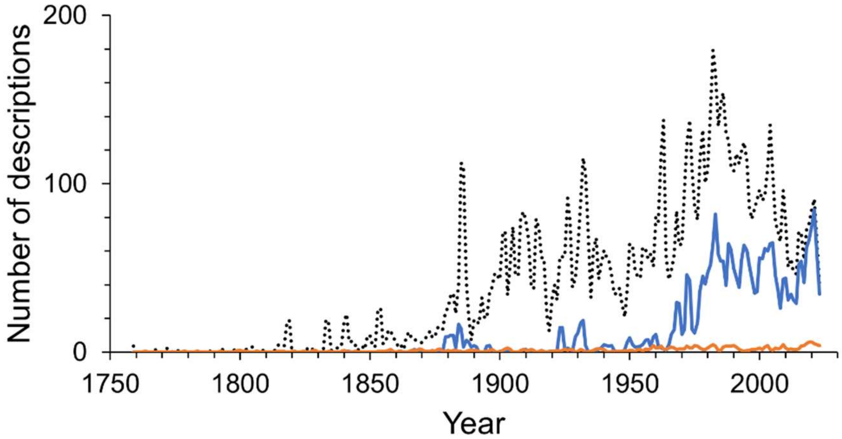

Figure 3.6. The annual number of descriptions for all species (dotted line). The solid blue line shows the multi-authored contributions per year, and the solid orange line shows the number of descriptions made by one-time authors. The lines are 2-year moving averages.

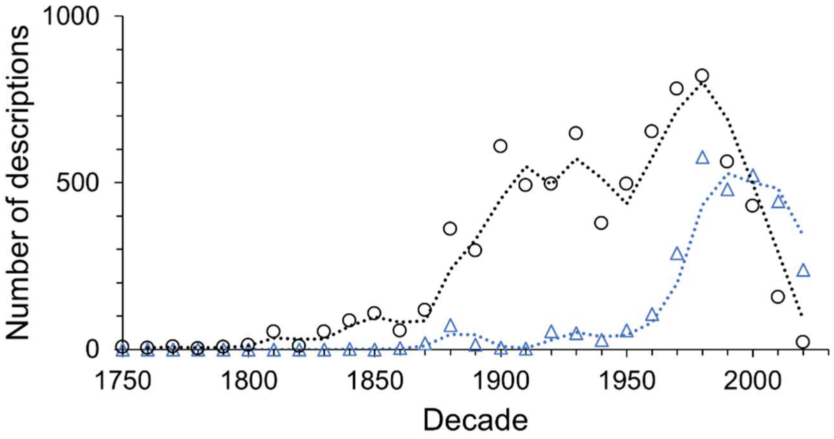

Figure 3.7. The number of descriptions published by sole (black circles) and multiple authors (blue triangles) in each decade.

3.4. Discussion

3.4.1. Named and unnamed species diversity

A decrease in the annual number of species described started more than three decades ago for all isopod species. Because this trend is not a short-term one, it cannot be explained by a time lag in data entry into the database. Estimates for future descriptions of species new to science from the non-homogeneous renewal process model predict approximately 660 additional species to be described until 2100. This suggests that 94% of isopod species that are predicted to be named by the end of this century already have been described. For other animal groups it has been estimated that about two thirds of all species are described, including stoneflies (DeWalt & Ower, 2019), scale insects (Deng et al., 2016), polychaete worms (Pamungkas et al., 2019), amphipods (Arfianti et al., 2018) and the world's marine species in general (Costello et al., 2012). Bryozoans have been labelled "one of the better-known taxa on Earth" due to the fact that about 80% of species predicted to be named by 2100 already had been described (Pagès-Escolà et al., 2020). Therefore, isopods represent a very well-known taxon. Of course, as more data will become available in the future these predictions may change. Bebber et al. (2007) showed that unless a taxon's species inventory is at least 90% complete extrapolations based on existing data may be associated with large margins of error.

Some more conspicuous taxa showed a decline in new species descriptions many decades ago, e.g., mammals globally (Fisher et al., 2018) and birds and flowering plants in the UK (Bebber et al., 2007). An asymptote in description rates was reached about one hundred years ago in well-studied regions, notably in Europe for mammals, birds, black corals, echiurans and euphasiid crustaceans (S. P. Wilson & Costello, 2005), as well as for fish, gastropods, sponges, cnidarians, echinoderms, bryozoans and tunicates in Britain and Ireland (Costello et al., 1996). Although an asymptote has not yet been shown for taxa globally, our data suggest that it may be emerging for isopods. Time will provide the confirmation needed. Similar analyses to those presented here for other taxa may show them to be reaching an asymptote as well.

This study did not take into account the number of already discovered but not yet formally described isopod species deposited in museum and research collections. Fontaine et al. (2012) noted an average shelf life of 21 years between the discovery and the taxonomic description of a new species. However, they also found that aquatic species have a shorter shelf life than terrestrial ones and that the shelf life of newly discovered invertebrate species is shorter than for plant or vertebrate species. For recently described isopod species, shelf life varied between 0 years (Monticelli Cardoso et al., 2022) and 54 years (Williams et al., 2020) with an overall tendency toward the lower range of the spectrum. For example, Malek-Hosseini et al. (2022) described a new groundwater species from Iran within three years of sampling, additionally using molecular data to corroborate its species status. In contrast, the material from which the first bopyrid isopod species from hydrothermal vents was described was collected 21 to 10 years before its taxonomic description (Kato et al., 2022). Naturally, field sampling continues to unearth new species. Depending on the sampling location, the proportions of reported unnamed isopod species in field samples may vary from none (in historically well-studied areas like Europe) to about 18% (López-Orozco et al., 2022 identified three new terrestrial species) and up to as much as 93% (Poore et al., 2015 found that only 9 o 127 marine species from western Australia were previously known to science). From the latter study, none of the sampled species were identifiable with any of the 359 isopod species collected on the continental slope of south-eastern Australia of which 90% were undescribed at the time of sampling (Poore et al., 1994), making Australia a rich source of new isopod species. Similarly, Brandt et al. (2007) found that only 13% of the discriminated 674 deep-sea isopod species from Southern Ocean samples were known to science. Thus, the Southern Ocean as well as the waters around Australia may account for a high proportion of the yet undescribed species globally. However, when these species will be described remains unknown. A list of 21 studies which reported undescribed species (Table A2) contains 1,225 possible new isopod species, of which most were sampled in the deep sea and in and around Australia. Given the average description rate of 75 descriptions/year from the past 20 years, it would take about 16 years to formally describe all these species. It has to be noted that those species were undescribed at the time of publication of the respective study. It has not been checked whether any of the reported species have been formally described since and might now already be part of our dataset of globally described isopod species. However, it is encouraging that a significant proportion of yet-to-be described species may already be collected and awaiting description.

Although scientists are continuously adding new names to the isopod inventory, not all of those names will prove to be valid. Several newly described species might be placed into synonymy over the years. Bouchet (2006) suggested that 10-20% of new species described each year will turn out to be synonyms. Likewise, Appeltans et al. (2012) note that it takes time to discover synonyms and estimated that for every five newly named species, at least two had already been described. Most synonymies will likely be identified and resolved during comprehensive revisions of isopod genera or families (e.g., Stransky et al., 2020; Taiti & Monticelli Cardoso, 2020). Examination of museum specimens may reveal synonyms (Hughes et al., 2020), as well as lead to the recognition of species new to science (Garcia, 2020). Thus, taxonomic revisions can decrease the number of accepted species, as well as discover new species.

3.4.2. Cryptic diversity

Another issue that adds to uncertainty about the number of existing isopod species is cryptic diversity whereby species can only be distinguished using molecular methods. However, isopod and other crustacean taxonomists stated they could always find morphological differences on close examination and thus true cryptic diversity in isopods is negligible (Appeltans et al., 2012, supplemental information). Recent years have seen an increase in species delimitation studies using molecular data as well as integrative taxonomic approaches (Pante et al., 2015), with some of them discovering putative new species. Species under scrutiny in such cryptic diversity studies tend to be geographically widespread species either in the deep sea (Raupach et al., 2007) or coastal habitats (Hurtado et al., 2016) or recognised species complexes already thought to harbour hidden diversity (Schnurr et al., 2018). Held (2003), for instance, tested the single-widespread-species-hypothesis of a morphologically variable Antarctic serolid isopod and identified two strongly distinct genetic clades uncovering an overlooked species. Likewise, a molecular analysis by Schnurr et al. (2018) disentangled two widely distributed munnopsid species complexes in Icelandic waters. Their data suggested that the *Eurycope producta* species complex consists of eight separate species, and the *Eurycope inermis* complex harbours four distinct species. Some of the discovered genetic clades could be linked to other already described species, leaving a total of seven species new to science. Even more putative new species have been uncovered during a genetic study of *Haloniscus* species from groundwater, springs, caves and salt lakes in Australia (Guzik et al., 2019). Each of the 26 new species was found to be restricted to a small geographical range. However, almost none of the previously unknown species detected by genetic sampling were truly cryptic species. Morphological characters could be found in just about every case, separating the new species from similar ones. Circling back to the problem of collected but unnamed species, few of the newly delimited species from molecular studies were formally described following their detection (Pante et al., 2015; Schlick-Steiner et al., 2007). Most studies note that additional taxonomic work is required to fully support a species hypothesis with a combination of DNA data and morphological characters (e.g., Guzik et al., 2019; Jennings et al., 2020). While molecular methods can be helpful in indicating specimens which may represent new species, and have been used since the 1980s for isopods and other taxa, there is no indication that they significantly increase description rates overall (Appeltans et al., 2012).

3.4.3. Taxonomic effort

The number of taxonomists describing new species of isopods has increased markedly over time, as it has for all taxa globally. Over the past fifty years, more authors have described isopod species than ever before (Fig. 3.4). Only for authors describing freshwater isopods has there been a steep decline within the past two decades (Fig. A1b), and this substantial decline is also evident in species numbers. Although it seems that freshwaters may not yield many more new species, it has been suggested that non-saline environments harbour high cryptic diversity (G. D. F. Wilson, 2008a). Indeed, a meta-analysis of cryptic diversity studies found that more posited cryptic species have been discovered in freshwater than in terrestrial or marine environments (Poulin & Pérez-Ponce de León, 2017). However, whether this genetic diversity translates into high species diversity is uncertain. Another interpretation of the decline in new freshwater species could be less taxonomic interest, but there seems no reason to assume why this may be the case.

Increasing numbers of people describing new species have been found in all similar studies (e.g., Appeltans et al., 2012; Arfianti et al., 2018; Costello, Vanhoorne, et al., 2015; Joppa et al., 2011a; Pagès-Escolà et al., 2020; Pamungkas et al., 2019; Tancoigne & Dubois, 2013), at least partly contradicting a not uncommon view that the field of taxonomy is in crisis (Bacher, 2012; Godfray, 2002; Hopkins & Freckleton, 2002). There is no doubt that taxonomy will benefit from more funding and renewed prestige (Agnarsson & Kuntner, 2007; Christenhusz & Byng, 2016; Higgs, 2016), but a lack of people describing new species is not evident from the data.

The field of taxonomy is not in decline but changing. It modernised itself from a primarily morphological discipline towards a multi-disciplinary field including genetics and phylogeny. Integration of these different skill sets could explain the now higher number of multi-authored descriptions. To avoid this trend of increasing proportions of multi-authored descriptions from affecting the trend in numbers of active taxonomists over time, only the first author of a species description was considered in our analysis. Therefore, the given numbers of authors contributing to isopod taxonomy are an underestimate of the taxonomic force. Also, the proportion of authors who described only a single isopod species has not increased for more than a century. Nor have taxonomists' publication lifetimes significantly decreased over this time. This further indicates that the increased number of taxonomic authors is an increase in effort, as concluded by others on other taxa (Appeltans et al., 2012; Essl et al., 2013; Joppa et al., 2011b), and not reduced by having proportionally more part-time taxonomists or more people who stop publishing descriptions after only a few years.

The present analysis did not consider the level of expertise of every author because this could not be determined from the available data. Some are well-established taxonomists who have spent a lifetime building up their extensive knowledge of a taxon and can therefore be considered true experts. Others are at the start of their career and still working towards expert status. Again, others contribute an essential amount of their work in other research fields, nevertheless adding valuable information with every published species description. Some people do not think it appropriate to call everyone who describes a species a taxonomist (Wheeler, 2014) and most likely, not everyone who does describe a species now and then would characterise themselves as such. However, regardless of which labels one puts on the authors of species descriptions, the fact remains that all of them contribute to the scientific inventory of the planet's biodiversity and draft testable hypotheses. Our data show that the percentage of people who publish only a single species description is tiny and has not increased for over a century. For more information on the perceived and detectable loss of expertise and the state of taxonomy in different countries, see Lovejoy et al. (2010), Boxshall & Self (2011), Coleman (2015), and the Australian Academy of Science (Taxonomy Decadal Plan Working Group, 2018). These assessments of taxonomy in the UK, Canada and Australia and New Zealand all considered people who described new species as a sub-set of all those working in taxonomy.

Although there have never been so many taxonomic authors than in recent decades, the average annual number of isopod species described per taxonomist has declined strongly over the last century. Such a decline in species per taxonomist has also been found for the closely related Amphipoda (Arfianti et al., 2018) and for other taxa, such as scale insects (Deng et al., 2016), flowering plants (Joppa et al., 2011a), as well as spiders, amphibians, birds and mammals (Joppa et al., 2011b), marine and terrestrial parasites (Costello, 2016), fossil and extant marine bryozoans (Pagès-Escolà et al., 2020) and overall marine and non-marine species (Costello, Wilson, et al., 2013). The reduction in the description rate of isopod species observed here, despite peak numbers of taxonomists, suggests that most species have already been named, as concluded for other taxa (Arfianti et al., 2018; Joppa et al., 2011b; Pamungkas et al., 2019). Contradicting this interpretation, Sangster & Luksenburg (2015) proposed that the lower number of species described per taxonomist is rather a consequence of the improved quality of species descriptions than a slowdown of progress in species discovery. They found that the number of pages of taxonomic descriptions has increased compared to the 1930s. So has the number of specimens on which the description of a new species is based, the number of characters to differentiate it from its most closely related species and the number of illustrations in a publication. With this increased effort put into the scientific description of a species, it may take more time from the initial discovery of a species until the publication of its formal description. However, other studies point to greater efficiencies in taxonomy due to greater access to field samples and literature, and improved museum collections, laboratory methods, publication efficiency, and communication between people (Costello et al., 2014; Eschmeyer et al., 2010). We found a similar productivity of taxonomists over their isopod-description careers, indicating that modern efficiencies and co-authorships may indeed balance out the richer species descriptions.

At the upper end of productivity, 21 taxonomists (only 3% of the taxonomic workforce over time) have described approximately 43% of all known isopod species. The three most prolific authors described almost exclusively terrestrial isopod species, which are more easily accessible and can be sampled without the deployment of advanced sampling equipment by comparison with marine isopods. Accordingly, our model estimates suggest that a considerable proportion of future discoveries might be made in the less accessible marine environment. Also, because large and geographically widespread species tend to be named first (Costello, Lane, et al., 2015; Higgs & Attrill, 2015), many of the yet-undiscovered isopod species are likely to be small and/or geographically restricted species (Liu et al., 2022; Scheffers et al., 2012). There is speculation on whether most of the yet-undescribed species will be found in collections (Coleman, 2015; Scheffers et al., 2012) or will be newly discovered during fieldwork (Grieneisen et al., 2014). However, both named and unnamed species, especially freshwater and endemic species, are at risk of extinction due to human impacts (Costello, 2015; Liu et al., 2022). Because many new species tend to be discovered in biodiversity rich-spots, which already face many threats like extensive habitat loss, they will be more vulnerable (Manes et al., 2021; Scheffers et al., 2012) and are at risk of going extinct before they are even discovered (Costello, May, et al., 2013). It is therefore important that taxonomists continue to describe new species. Only named, and as such well delimited species, can be included in threat reports and conservation plans.

4. Isopod taxonomic diversity is bimodal with latitude

4.1. Introduction

Biodiversity is not uniformly distributed across Earth. One of the most striking and historically well-explored patterns of biodiversity is the latitudinal diversity gradient (LDG) in species richness. Until relatively recently, the longstanding view was that the LDG is categorically unimodal, i.e. bell-shaped with a diversity peak at or near the equator and decreasing species richness toward the poles (e.g., Stehli et al., 1969). While this pattern can indeed be observed in some terrestrial organisms (Clarke & Crame, 1997; Dantas & Fonseca, 2023) and a few marine groups (e.g., Boltovskoy & Correa, 2017), in recent years evidence has accumulated that the LDG is indeed bimodal for most taxa with a dip in the equatorial region (Cerezer et al., 2022; Chaudhary et al., 2016). Although some argued that the observed bimodality is an artefact of sampling bias due to insufficient sampling of equatorial regions (Fernandez & Marques, 2017; Menegotto & Rangel, 2018), several studies that accounted for sampling bias in their data concluded it does not affect the overall shape of the LDG (Boltovskoy & Correa, 2017; Chaudhary et al., 2017; Rivadeneira & Poore, 2020). The precise shape of the gradient is taxon-specific (Chaudhary et al., 2017), in many cases showing interhemispheric asymmetry (Crame, 2000; Hernáez et al., 2021; Kussakin, 1973) or exhibiting an inverse latitudinal pattern with diversity peaks outside the tropics (Cerezer et al., 2022; Gray & Rabeling, 2023; Krug et al., 2007).

Modern latitudinal diversity gradients have likely been maintained for millions of years. The fossil record of 50,000 marine species indicates that there has been reduced species richness at the equator during warm interglacial periods (Chaudhary et al., 2016). Studying both marine and terrestrial taxa, Crame (2023) suggested that the gradient may be a fossil feature that formed in the Early Cenozoic when evolutionary rates were higher in the tropics before extra-tropical regions expanded in the Late Cenozoic providing more opportunities for speciation in higher latitudes. Culver and Buzas (2000) assumed a similar temporal placement (~36 mya) for the origin of the LDG in their study on benthic deep-sea foraminifera. However, they argued for a seasonally fluctuating food supply in higher latitudes as a cause in the wake of global climatic cooling, an argument Crame also used in an earlier study (Crame, 2020). Focusing on planktonic foraminifera, in contrast, resulted in the hypothesis that a modern-style LDG emerged only 15 mya when enhanced thermal niche partitioning at low latitudes led to higher speciation rates (Fenton et al., 2023).

Although the generality of the LDG is widely accepted, there is no consensus on the drivers behind this pattern (Currie et al., 1999; Hillebrand, 2004a). Even though most studies only consider an individual process or category of variables, the reality is far more complex, and there is likely no single explanatory mechanism that shaped and maintained the LDG (Condamine et al., 2012; Cruz-Motta et al., 2020; Gaston, 2000). Causes are taxon-specific, like the shape itself (Cerezer et al., 2022), and it is essential to keep in mind that, although the gradient in species richness is commonly analysed at a global scale, local and regional processes strongly influence diversity, too (Cruz-Motta et al., 2020; Dantas & Fonseca, 2023). The numerous hypotheses brought forward to explain the causality of the LDG can roughly be grouped into evolutionary and ecological processes. Some researchers propose that the modern latitudinal diversity gradient has been formed due to varying speciation and extinction rates (Allen & Gillooly, 2006; Crame, 2023; Krug et al., 2007). Others suggest that environmental variables like temperature, salinity, and precipitation (Boltovskoy & Correa, 2017; Brayard et al., 2005; Dantas & Fonseca, 2023; Hernáez et al., 2021) have shaped the gradient in species richness or else the seasonality of primary productivity (Crame, 2020; S. J. Culver & Buzas, 2000; Knauber et al., 2023). Boag et al. (2021) demonstrated a strong connection between temperature and marine diversity through time, with diversity peaks at moderate temperatures of 15-25°C (also see Costello et al., 2023). They assumed that temperature indirectly drives the gradient by affecting the aerobic scope of ectotherms (Boag et al., 2021). Similarly, Culp et al. (2019) found support within the freshwater realm for the hypothesis that physiological tolerance is an important driver of species richness. Biotic interactions like the intensity of predation and its impact on the prey community have also been hypothesised as drivers of species richness across latitudes (Ashton et al., 2022; Freestone et al., 2021).

While early broad-scale studies on biogeographic patterns in isopod species richness often did not find a clear latitudinal gradient within the group or broader region they examined (Kussakin, 1973; G. D. F. Wilson, 1998), current research points to a well-formed bimodal latitudinal diversity gradient both in the ocean and on land (Rivadeneira & Poore, 2020; Saeedi et al., 2022; Sfenthourakis & Hornung, 2018). The most comprehensive analysis of the LDG of marine isopods so far on spatial and taxonomic scales, as well as the amount of data, was performed by Rivadeneira and Poore (2020). They found an asymmetric bimodal LDG for marine Isopoda with diversity peaks in temperate areas that was variable across depth.

However, a similar comprehensive global study for terrestrial and freshwater isopods is still lacking. Here, a global dataset of occurrence records for the whole order Isopoda is compiled from the OBIS (https://obis.org) and GBIF (https://www.gbif.org) databases to analyse latitudinal patterns in species richness across all environments. The resulting dataset contains almost double the number of marine species included in the study of Rivadeneira and Poore (2020) and is also analysed according to different depth categories, in addition to the overall marine gradient. In this study, it is analysed whether the LDG of isopods is bimodal in terrestrial and freshwater environments, too. Possible asymmetry and the location of diversity peaks are also examined.

4.2. Methods

4.2.1. Data source and cleaning process

Global occurrence records of isopods were downloaded from the Ocean Biodiversity Information System (OBIS, 2022) and the Global Biodiversity Information Facility (GBIF, 2022) on 10th June 2022. The datasets from the two databases were merged and cleaned in R version 4.1.1 (R Core Team, 2021). First, duplicates, records without coordinates, and those not identified down to species level were removed. Then, to minimise the number of records with data-entry errors, the dataset was further cleaned using the "CoordinateCleaner" package (Zizka et al., 2019), removing records with equal latitude and longitude coordinates, those that were geo-referenced to capitals or country centroids, and records in close vicinity (100 m) of biodiversity institutions. All species names were verified using the taxon matching tool from the World Register of Marine Species (Ahyong et al., 2022) to resolve synonyms and misspellings. Further, all records that were found to be fossil were removed during the cleaning process, and information on whether a species is parasitic or subterranean was added to the dataset. Finally, the data were mapped with QGIS version 3.28.2, and all marine records mapped on land and non-marine records mapped in the ocean were removed or, where possible, resolved according to their provided locality information. Remaining records with a coordinate uncertainty of more than 100 km were removed if they had no specific locality information, and therefore, their locations could not be verified. The resulting dataset, which was used for the analysis, contains 388,881 occurrence records of 5,935 marine and non-marine isopod species (Hartebrodt, 2023a). A list of all included datasets can be found in Appendix B (Table B1).

4.2.2. Data analysis

First, the latitudinal diversity gradient of isopods was examined using two components of species richness, namely alpha and gamma diversity. For alpha diversity, the number of species in each 5° latitude-longitude cell was counted, and subsequently, mean alpha diversity per 5° latitudinal band was plotted. Gamma diversity was counted as the number of distinct species in each 5° latitudinal band. This was done for the complete dataset and subsets according to habitat (marine, freshwater, terrestrial) and ecology (parasitic or subterranean isopods). Additionally, the marine subset was further divided into different depth categories because diversity patterns vary across depth (Clarke & Crame, 1997; Rivadeneira & Poore, 2020; Saeedi et al., 2022). Approximately 52% of marine occurrence records included bathymetric information. These were grouped into three depth categories: shallow (0 to 200 m), intermediate (>200 to 500 m), and deep (>500 m). Both alpha and gamma diversity are, to a varying extent, biased by sampling effort. Therefore, Hill numbers with order $q = 1$ (Shannon diversity) were calculated to account for differing sampling effort in each latitudinal band using the "iNEXT()" function of the R package "iNEXT" (Hsieh et al., 2022). Estimates were rounded to whole numbers so that they were counts. For an in-depth assessment of how hill diversities improve estimates of species diversity, see Roswell et al. (2021). They also recommend using the Hill-Shannon diversity metric when the research goal is to characterise gradients in biodiversity because it emphasises neither rare nor common species.

4.3. Results

After all cleaning steps were performed, the resulting dataset used in this analysis contained 388,881 occurrence records of 5,935 isopod species. Compared with the global species list compiled from WoRMS data in Chapter 3, this corresponds to a coverage of approximately 56% of all described isopod species. About 72% of genera and 91% of families are represented in the dataset. Of all analysed species, 4,499 are marine (coverage of ~73%), 1,048 species are terrestrial (~27%), and 388 species live in freshwater (~56%). Within the two ecological subsets, 887 (~60%) parasitic and 407 (~41%) subterranean isopod species are recorded.

There is a strong sampling bias towards the northern hemisphere (see Table 4.1 and graph b in Figs. 4.1 – 4.9). While 351,600 occurrences are recorded for the northern hemisphere, the dataset contains only 37,281 occurrences for the southern hemisphere. However, there is no such marked difference in species numbers. In the northern hemisphere, 3,257 isopod species are recorded, and only a few less (3,183 species) in the southern hemisphere. A pattern of higher species richness in the northern hemisphere is consistent in all subsets except the marine one. Although considerably more records are documented in the northern hemisphere, the southern hemisphere is richer in marine isopod species (2,745) than the northern (2,203 species). The southern hemisphere is substantially under-sampled for freshwater isopods and species from subterranean habitats (see Table 4.1).

Table 4.1. Number of occurrence records and species per hemisphere for the complete dataset and the various subsets.

	Southern hemisphere		**Northern hemisphere**	
	# Records	# Species	# Records	# Species
All isopods	37,281	3,183	351,600	3,257
Marine	31,221	2,745	129,186	2,203
Terrestrial	5,181	331	182,513	768
Freshwater	879	107	39,901	287
Parasitic	2,646	440	12,201	579
Subterranean	137	46	8,647	362

Both alpha and gamma diversity of all observed isopod species show a well-formed bimodal latitudinal gradient with a trough in equatorial regions (Fig. 4.1a, c). Figure 4.1b depicts the number of occurrence records per 5° latitudinal band with an extreme peak at 55°N. Nevertheless, total species richness displays two fairly symmetrical peaks of similar height (Fig. 4.1c). In both hemispheres, the peaks are situated outside the tropics at 35°S and 45°N, respectively. Still, alpha and gamma diversity are positively correlated with the number of occurrence records in each 5° latitudinal band (Spearman's rho = 0.80 for mean alpha diversity; Spearman's rho = 0.61 for gamma diversity). To account for the sampling bias in the data, Hill numbers were used as a metric for estimated species richness. Regardless, the resulting graph of the latitudinal gradient shows a bimodal pattern with a dip around the equator (Fig. 4.1d). In this visualisation, the diversity peaks move inside the tropics, and the expected diversity within the temperate northern hemisphere is considerably lower than the observed diversity in the dataset.

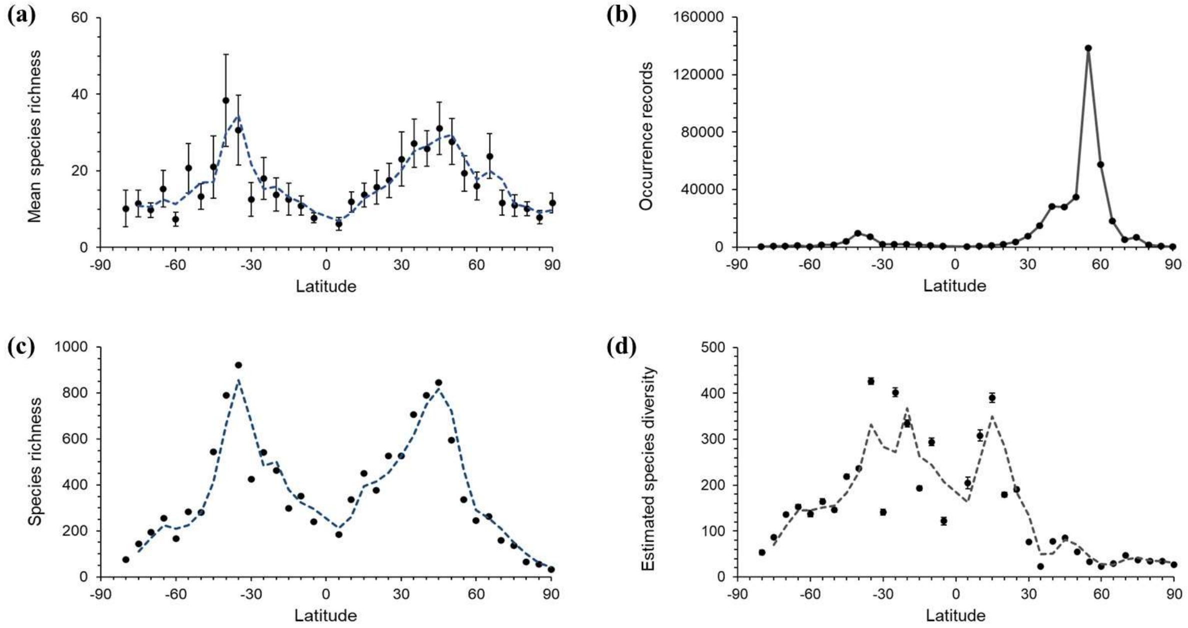

Figure 4.1. Latitudinal patterns of species richness and sampling effort of the order Isopoda. **(a)** Alpha diversity, **(b)** number of occurrence records (as a proxy of sampling effort), **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

The latitudinal gradients of alpha and gamma diversity of marine isopods are also bimodal with peaks outside the tropics (Fig. 4.2a, c). In contrast to the overall LDG of all isopod species, the marine gradient shows a marked asymmetry with a smaller peak in the northern hemisphere (Fig. 4.2c). The peak is situated at 35°N with 539 observed species within the corresponding 5° latitudinal band. Species richness in the southern hemisphere oceans peaks at 35°S with 800 recorded species. Again, most occurrence records are from the northern hemisphere, with a high peak at 55°N (Fig. 4.2b). The southern hemisphere peak of marine occurrences is larger (in relation to the northern hemisphere peak) than the one from the complete dataset (see Fig. 4.1b) but is still four times smaller than the northern peak. The bimodality of the gradient is retained with the estimated species diversity, again with a relatively lower estimated diversity in the temperate northern hemisphere (Fig. 4.2d). However, for estimated richness, tropical diversity is expected to be higher than it is observed in the analysed dataset.

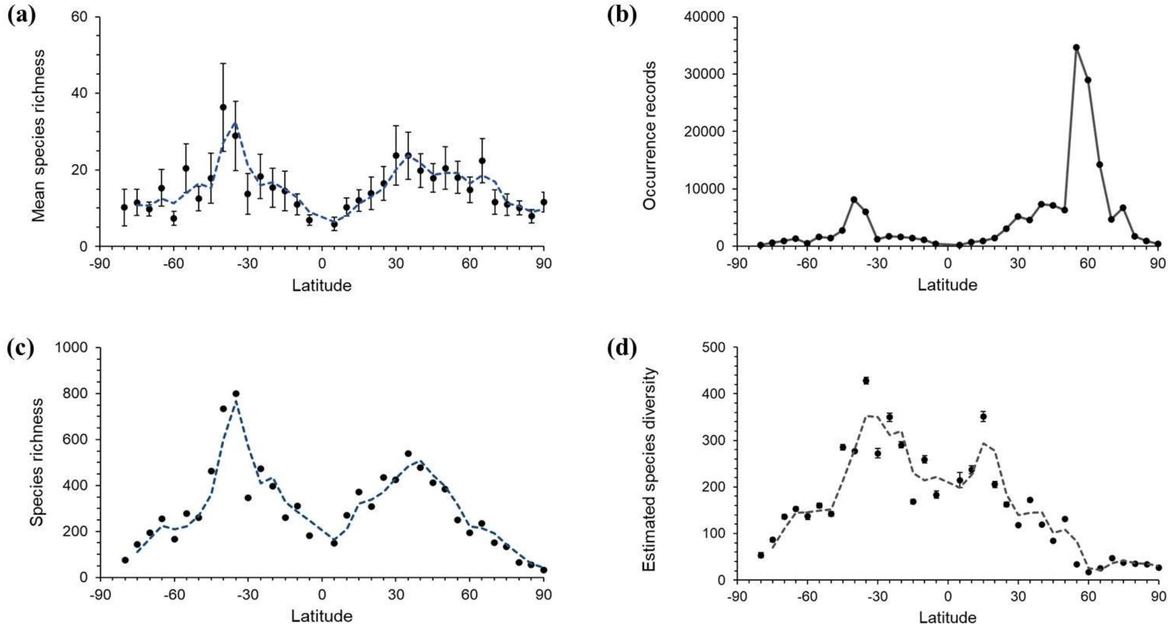

Figure 4.2. Latitudinal patterns of species richness and sampling effort of marine isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

There are considerable differences between the diversity patterns of shallow-water isopods and species occurring below 200m. Most of the marine species were recorded in the shallow depth category (2,107 species; see Fig. 5.2 in Chapter 5). The LDG resulting from this subset of the marine data is similar to the overall marine gradient with a more prominent diversity peak in the southern hemisphere outside the tropics and a relatively higher estimated diversity within tropical regions (Fig. 4.3). Species diversity below 200m shows additional peaks at high latitudes (Figs. 4.4 & 4.5). The dataset of the intermediate depth category contains 739 species, of which only 150 species were reported exclusively between 200m and 500m. The observed species richness within this depth category is highest in northern hemisphere high latitudes but also shows considerable peaks in the southern hemisphere mid- and high latitudes (Fig. 4.4c). Estimated species diversity is, however, highest in the southern hemisphere (Fig. 4.4d). The deep category dataset with occurrences below 500m contains 1,331 species. Most of these species are recorded from mid- and high latitudes in both hemispheres, with the highest diversity peak in the southern hemisphere at 40°S (Fig. 4.5c). The latitudinal gradient of the estimated deep-sea species diversity exhibits a very similar pattern to the observed diversity, with a more marked difference between the peak heights in the northern and southern hemispheres (Fig. 4.5d).

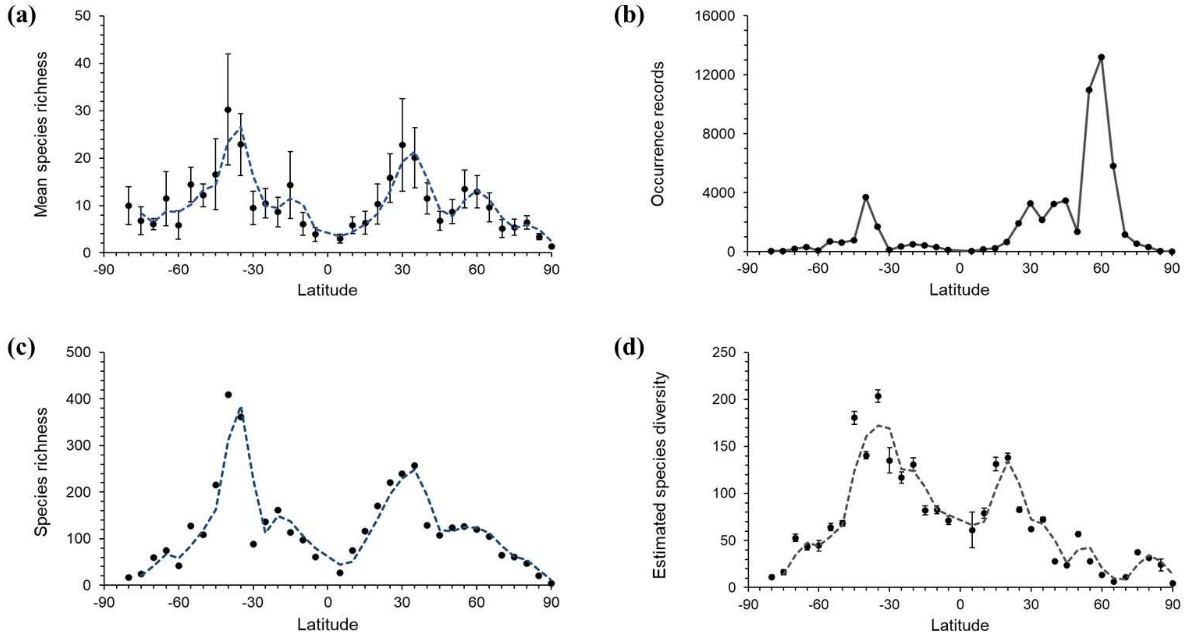

Figure 4.3. Latitudinal patterns of species richness and sampling effort of shallow-water marine isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

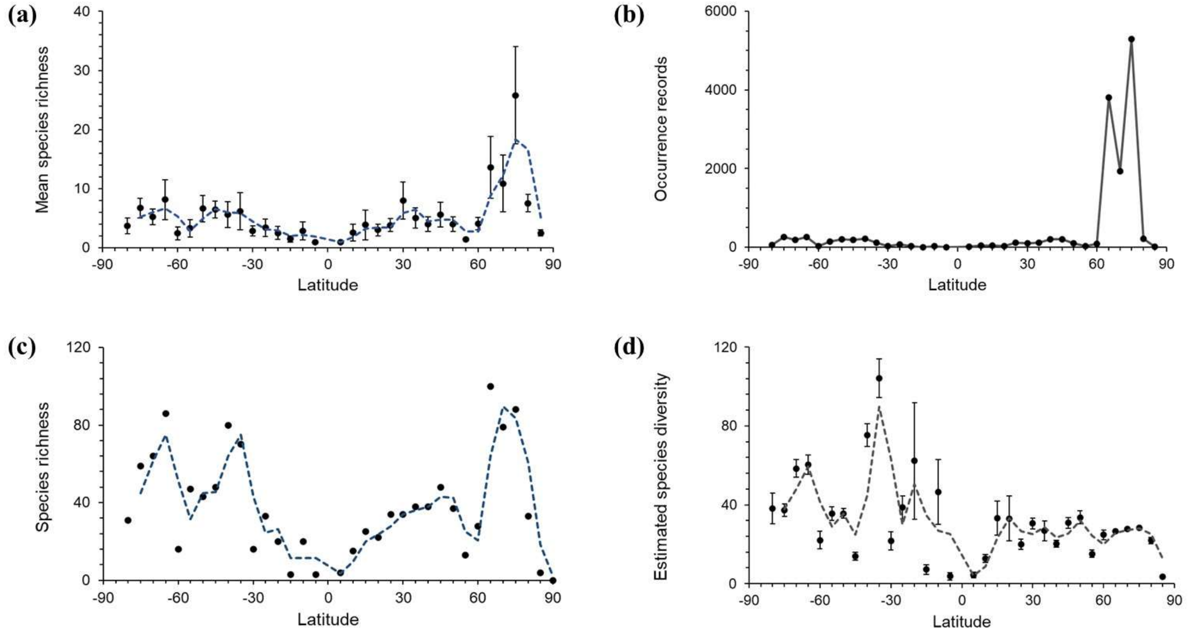

Figure 4.4. Latitudinal patterns of species richness and sampling effort of marine isopods in the intermediate depth category. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

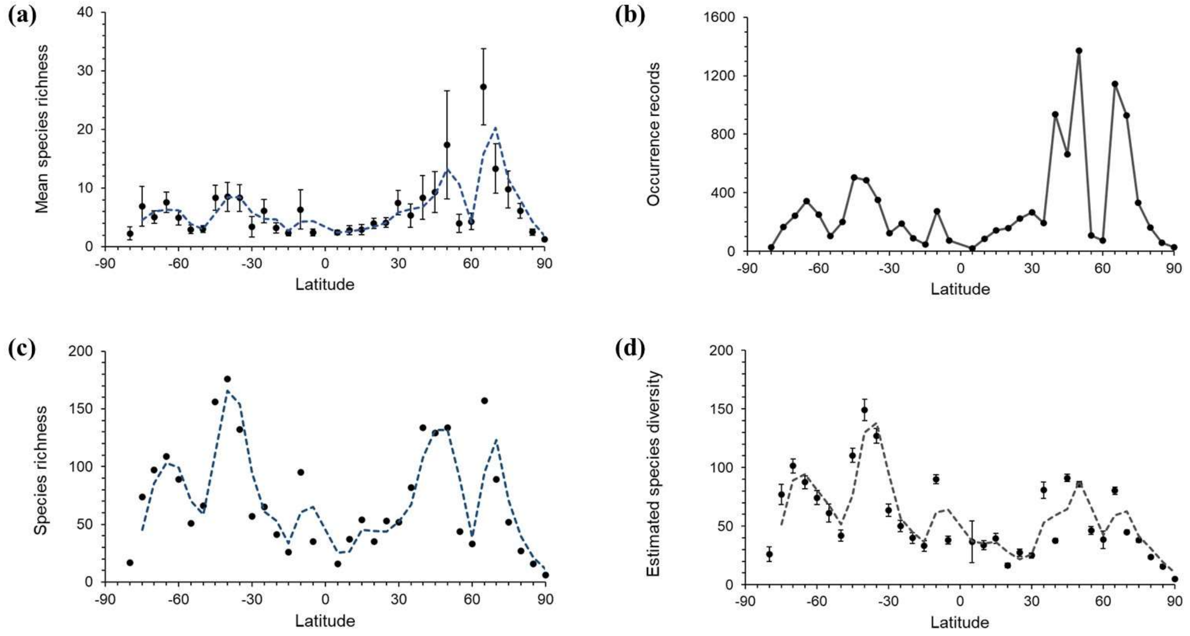

Figure 4.5. Latitudinal patterns of species richness and sampling effort of deep-sea isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

Like the overall marine gradient, the LDG of terrestrial isopods is asymmetrically bimodal with peaks outside the tropics (Fig. 4.6a, c). However, unlike marine isopods, terrestrial isopods have a larger peak in the northern hemisphere at 45°N. The majority of occurrence records are documented from the northern hemisphere as well, again with its peak at 55°N (Fig. 4.6b). After accounting for sampling bias, the gradient of estimated species diversity shows three peaks (Fig. 4.6d). The lower southern hemisphere peak and the trough in equatorial regions are similar to the observed species richness, though the peak moved into the tropics. However, the high northern hemisphere peak is diminished, and the now highest peak within the northern hemisphere tropics is added. This might suggest a strong effect of sampling bias on the observed latitudinal gradient of gamma diversity and under-sampled tropical regions. That is not surprising since the analysed dataset contains less than a third of all described terrestrial species and is, therefore, highly incomplete.

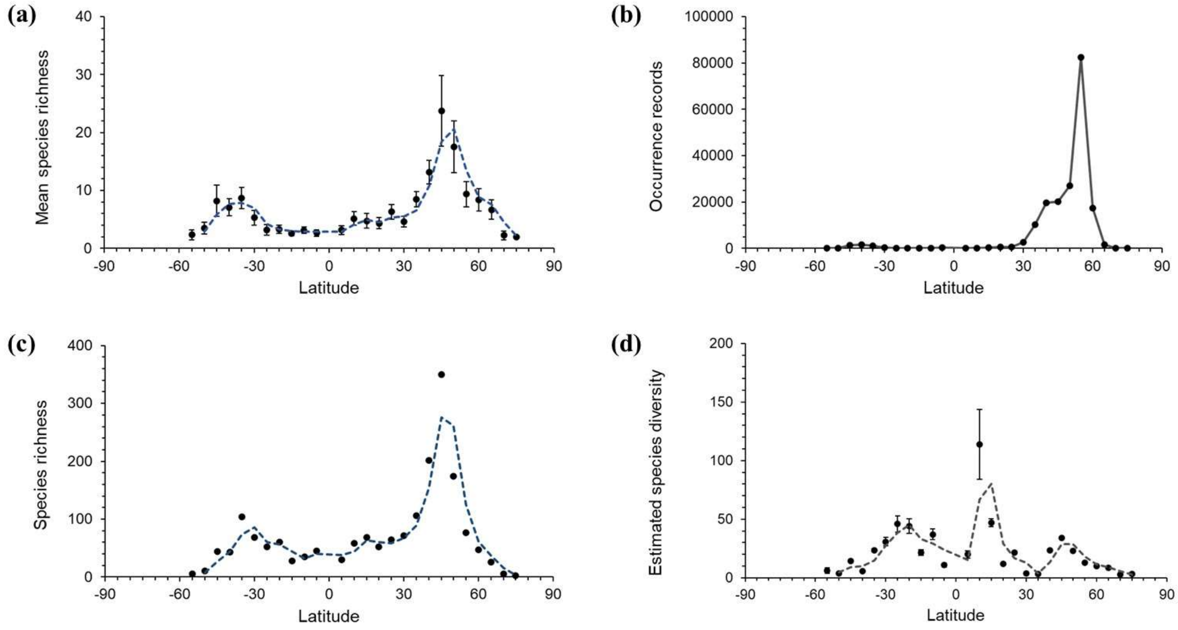

Figure 4.6. Latitudinal patterns of species richness and sampling effort of terrestrial isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

Freshwater isopods also exhibit an asymmetrically bimodal diversity gradient (Fig. 4.7a, c). Again, both peaks are situated outside the tropics, with the more prominent peak in the northern hemisphere at 40°N. Like within the other subsets, most occurrences are recorded in the northern hemisphere (Fig. 4.7b). According to the Hill diversity estimates, more freshwater species should be expected in tropical regions (Fig. 4.7d). Nevertheless, the highest diversity peaks are still situated outside the tropics.

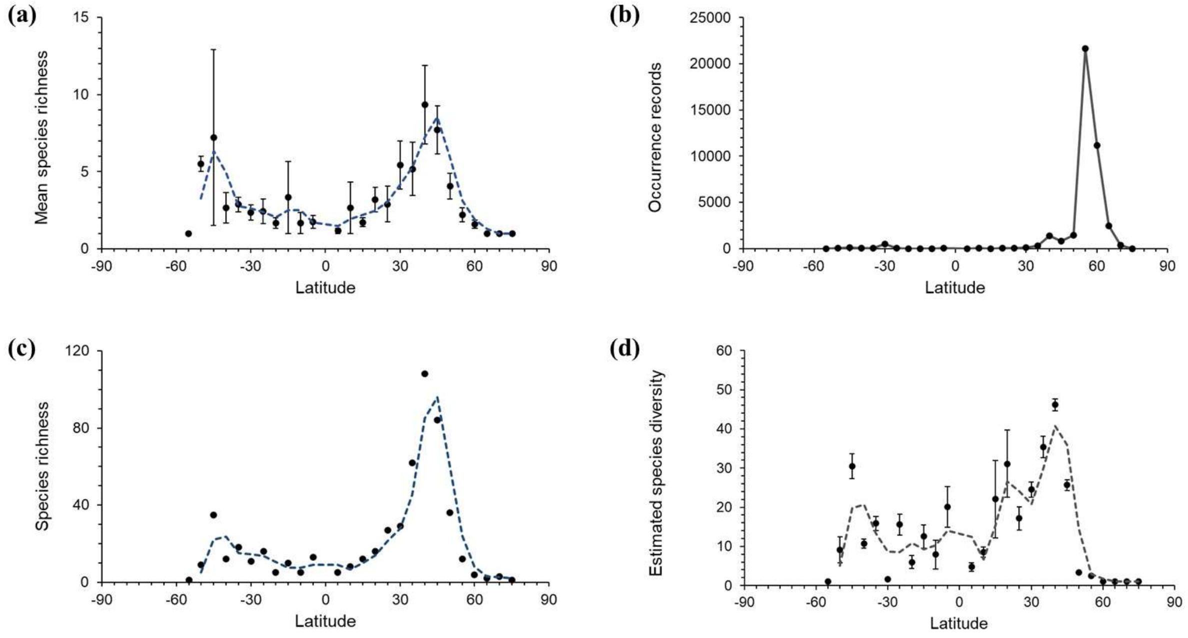

Figure 4.7. Latitudinal patterns of species richness and sampling effort of freshwater isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

The parasitic subset contains all obligate parasite species present in the complete dataset. All parasitic isopods are aquatic, with the majority being marine. Nevertheless, unlike the entire marine group, parasitic isopods show a higher diversity peak in the northern hemisphere (Fig. 4.8a, c). The LDG of parasites is asymmetrically bimodal with peaks at 30°N and 25°S and a dip near the equator. Tropical diversity is, nonetheless, high. In addition to the high temperate northern hemisphere peak in occurrences, sampling effort is also spread out over lower latitudes in both hemispheres (Fig. 4.8b). Estimated species diversity is consistent with showing low numbers of parasite species at high latitudes and most of the diversity within the tropics (Fig. 4.8d). Species diversity in equatorial regions is expected to be slightly higher than observed in the analysed dataset.

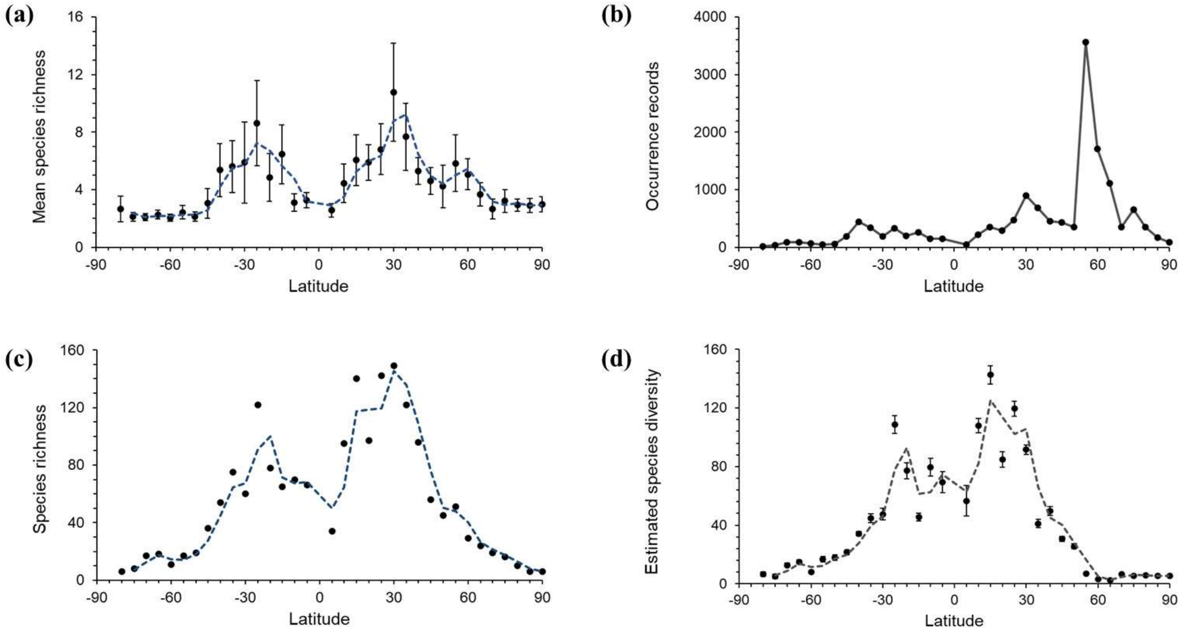

Figure 4.8. Latitudinal patterns of species richness and sampling effort of parasitic isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

Species that were included in the subterranean subset are cave and groundwater species and species that inhabit interstitial spaces. Of the 407 recorded species, 181 live in freshwater, 128 are terrestrial, and 53 live in the marine environment. Almost all records are from the northern hemisphere mid-latitudes (Fig. 4.9b). The bimodal LDG of subterranean species is highly asymmetrical (Fig. 4.9a, c). While the small peak in the southern hemisphere is situated at the edge of the tropics at 25°S, the more than four times larger northern hemisphere diversity peak is located outside the tropics at 45°N. Hill diversity depicts a similar gradient with slightly higher diversity in tropical regions, especially in the northern hemisphere (Fig. 4.9d).

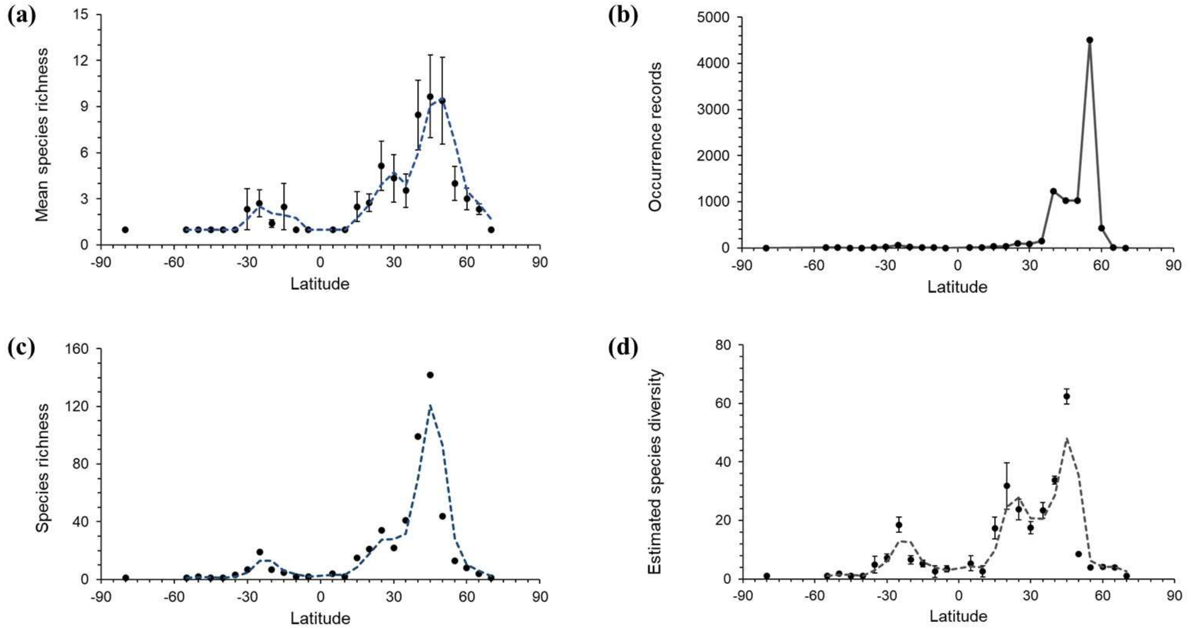

Figure 4.9. Latitudinal patterns of species richness and sampling effort of subterranean isopods. **(a)** Alpha diversity, **(b)** number of occurrence records, **(c)** gamma diversity, and **(d)** estimated species diversity. The dashed lines are two-point moving averages.

4.4. Discussion

The presented results of latitudinal diversity patterns of isopods are consistent with the reported bimodality of latitudinal gradients in most marine taxa (Chaudhary et al., 2016, 2017), as well as many examples from terrestrial and freshwater taxa (e.g., Cerezer et al., 2022; Gray & Rabeling, 2023; Orr et al., 2021). The overall LDG of the whole order Isopoda is symmetrically bimodal with similar species richness in both hemispheres (Fig. 4.1). However, for most of the subsets, the northern hemisphere shows a higher diversity peak than the southern hemisphere (see Figs. 4.6 – 4.9), as was found in most groups that were analysed by Chaudhary et al. (2016). It could be argued that this pattern arises from uneven sampling, as the majority of occurrence records are from northern hemisphere temperate latitudes. Nevertheless, even when accounted for sampling bias, estimated richness is still higher in the northern hemisphere in these cases. Even more compelling is the example of marine isopods, where higher species richness is observed in the southern hemisphere despite four times more records being available in the northern hemisphere (Table 4.1 & Fig. 4.2). Several studies have demonstrated that sampling bias has only a marginal effect on the overall shape of the LDG (Boltovskoy & Correa, 2017; Chaudhary et al., 2017; Rivadeneira & Poore, 2020), as well as that the omittance of large numbers of unidentified species does not significantly affect large-scale diversity patterns (Pos et al., 2014; Rivadeneira et al., 2011).

Chaudhary et al. (2016) attributed the higher northern hemisphere richness of marine species to the greater availability of coastal shelf area since the majority of species are distributed in shallow waters. Interestingly, in the current analysis, marine isopods are an exception to the widely observed pattern of higher northern hemisphere richness. They show greater species richness in the southern hemisphere, both for observed gamma diversity and estimated diversity (see Fig. 4.2). In part, this might be attributed to the fact that isopods are also very diverse in the deep sea (Brandt et al., 2016; Kussakin, 1973), of which the southern hemisphere provides an extensive area, as well as to the higher diversity of Antarctic waters compared to the Arctic Ocean (Clarke & Crame, 1997). Thermal isolation of the Southern Ocean by a deep-water current system in the Cenozoic may have been crucial to the radiation of Antarctic isopod species and led to the immense extant biodiversity observed in the region (Brandt et al., 1999). When examined within different depth categories, deep-sea isopods and shallow-water species exhibit a higher diversity peak in the southern hemisphere (Figs. 4.3 & 4.5).

The southern hemisphere peak in marine diversity coincides with the location of some very species-rich biogeographic regions like Australia, New Zealand, and South Africa (see Chapter 5). Australia is a well-known diversity hotspot for marine isopods, which harbours a high percentage of yet undescribed species (Poore et al., 1994, 2015). Also, Kaiser et al. (2020) found asellote diversity in New Zealand waters to be average to high compared to other regions in both hemispheres. Levels of endemism for marine isopods are higher than for most other taxa. For example, Kensley (2001) recorded 226 endemic species (68%) for the South African region. Besides these mid-latitude regions, the Southern Ocean is another diversity hotspot for isopods, inhabited by a distinct fauna with many species new to science (Brandt, Brix, et al., 2007). Marine isopods are not the only group with higher southern hemisphere diversity. Similar richness patterns were observed in, e.g., amphipods (Arfianti & Costello, 2020; but see the contrasting results of Rivadeneira & Poore, 2020), polychaetes (Pamungkas et al., 2021), hard corals and fish (Chaudhary et al., 2016), and benthic deep-sea foraminifera (S. J. Culver & Buzas, 2000).

Latitudinal diversity gradients of marine crustaceans have been found to vary according to the lifestyle of different groups, i.e. whether a taxon possesses a planktonic larval phase. Taxa with pelagic larvae exhibit steeper LDGs and higher species richness towards the tropics than taxa without planktonic larvae (Rivadeneira & Poore, 2020). Similarly, examining diversity patterns in the northwestern Pacific Ocean, Knauber et al. (2023) observed a diversity peak for pelagic crustacean species at 30°N, while benthic species richness peaked at 45°N. These findings agree with the results of the current analysis that show an inverse LDG with peaks outside the tropics for marine isopods, which are benthic brooders. The LDG of marine isopods presented herein is almost identical to the one presented by Rivadeneira and Poore (2020), although their results are based on a smaller dataset, and occurrences had been binned in 10° latitudinal bands instead of 5° bands like in the current analysis. This shows again that the unavoidable biases within the analysed datasets (i.e., number of occurrences, taxonomic completeness) do not significantly affect the overall shape of a large-scale diversity gradient. The location of peaks within marine diversity gradients has been found to vary during cold and hot periods in Earth's history. Boag et al. (2021) determined that diversity peaks are located within areas with moderate temperatures of 15-25°C and assumed that the effects of ocean temperature on the aerobic scope of marine organisms are a primary driver behind the gradient. This would explain why there is lower species richness in equatorial regions. It is simply too hot for many species. With further climate warming, the dip in the LDG around the equator will likely become more pronounced (Chaudhary et al., 2021). Another factor that is possibly involved in the formation of a diversity trough around the equator is the increased interaction strength of predation in tropical latitudes, both in the ocean and on land (Ashton et al., 2022; Freestone et al., 2021; Roslin et al., 2017).

All parasitic isopods are aquatic, i.e. either marine or freshwater species. However, although both marine and freshwater groups have their diversity peaks outside the tropics at intermediate latitudes, the subset of parasitic isopods exhibits its highest diversity at low to intermediate latitudes (see Fig. 4.8), with a higher proportion of tropical species than their "parent" groups. As the geographical distribution of parasites presumably reflects their host species' distribution (Markham, 1986), this high tropical diversity is not surprising. The majority of hosts are either decapod crustaceans or bony and cartilaginous fish, both of which are very species-rich at low latitudes (Lin et al., 2021; Rivadeneira & Poore, 2020).

Since depth adds an important third dimension to the ocean, Clarke and Crame (1997) recommended considering patterns of diversity separately for shallow waters and the deep sea. Indeed, the gradients for shallow-water isopods and deep-sea species show different patterns (also observed by Rivadeneira & Poore, 2020). While the diversity of shallow-water species peaks at mid-latitudes, deep-sea diversity exhibits additional peaks at higher latitudes within polar seas (see Figs. 4.3 & 4.5). The structure of the deep sea floor is not as homogeneous as often believed (e.g., Riehl et al., 2020), and species richness and composition can be highly variable between regions (Malyutina & Brandt, 2020). While Rex et al. (1993) could not observe significant latitudinal differences in deep-sea isopod diversity in the southern Atlantic Ocean due to the small number of samples over a restricted latitudinal range, they noted high species diversity in the Argentine Basin at temperate latitudes. Further, in the northwest Pacific, the highest deep-sea richness was found at intermediate latitudes of 42°-44°N, which was best explained by topography and temperature (Saeedi et al., 2020). Also, intensive sampling in the Southern Ocean revealed higher species richness in the deep sea than along the Antarctic shelf (Brandt et al., 2016). It is assumed that both ecological and evolutionary processes shape the diversity patterns of the deep sea benthos (Rex et al., 1997).

Terrestrial isopods exhibit their highest diversity in the northern hemisphere temperate latitudes where a lot of landmass area is available, but also most of the occurrences were recorded (Fig. 4.6). The peak in number of occurrences and species richness coincides with the location of well-studied regions in central Europe and the northern United States. Nevertheless, despite scarce records, the gradient also shows a smaller peak in the southern hemisphere. When sampling bias is accounted for, higher diversity than currently observed is expected in the tropics in both hemispheres. Earlier examinations of oniscidean global-scale distribution data hinted at a bimodal LDG that peaked at mid-latitudes, in which some of the species-rich families have their main distribution range (Sfenthourakis & Hornung, 2018). For terrestrial isopods, precipitation and mean annual temperature are important environmental variables structuring their distribution (Csonka et al., 2018; Kuznetsova & Gongalsky, 2012). Also, at finer spatial scales, environmental heterogeneity strongly influences patterns in species richness (Gentile et al., 2022). For instance, Hornung et al. (2008) observed a relatively uniform richness pattern with high compositional turnover in the Transdanubian region of western Hungary. However, species richness significantly decreased from natural, wet habitats to disturbed, dry habitats (Hornung et al., 2008).

The herein-analysed terrestrial dataset also includes subterranean species. D. C. Culver et al. (2006) found temperate areas in Europe and North America to be biodiversity hotspots for obligate cave-dwelling invertebrates. These areas are characterised by high surface productivity and cave density. A high richness of cave-dwelling species in the northern hemisphere mid-latitudes fits the observed patterns of isopods well. The subterranean subset contains not only terrestrial species but also groundwater-associated species (which are part of the freshwater dataset). The resulting LDG of subterranean isopods (Fig. 4.9) is very similar in shape to the observed terrestrial and freshwater gradients (Figs. 4.6 & 4.7) with a high diversity peak in temperate latitudes of the northern hemisphere. However, after accounting for sampling bias in the data, the gradients of estimated species diversity differ considerably from each other (see graph d in Figs. 4.6, 4.7 & 4.9). Data for non-marine isopods are scarce in the southern hemisphere, especially for freshwater and subterranean species (see Table 4.1). The scientific literature has documented many more occurrences than have been made available in the biodiversity databases used in the current analysis. Those and future occurrence records need to be added to global databases to improve data access for large-scale studies and the valuable results that can be gained. With more data available, the LDGs will be refined. It will be interesting to see whether species richness in the tropics for terrestrial and freshwater isopods is indeed higher than currently known.

Latitudinal diversity gradients of freshwater species have been observed to be less steep than terrestrial or marine gradients (Hillebrand, 2004b, 2004a). Examining diversity gradients of earthworms in North America, Ikeda et al. (2020) found that freshwater diversity peaked at mid-latitudes, while terrestrial diversity peaked at lower latitudes. These patterns correspond with the estimated species diversity gradients of isopods, where terrestrial diversity peaks moved into the tropics, while freshwater diversity still peaked outside the tropics. Ikeda et al. (2020) also observed that many freshwater species had limited geographic ranges compared to wider-ranging terrestrial ones. However, north of 40° latitude, species had expanded their ranges into habitats that became available after glaciation periods (Ikeda et al., 2020). Physiological tolerance limits of species are an important driver of species richness. In Arctic freshwater macrofauna, diversity declined with increasing latitude as species were lost instead of being replaced by other species (Culp et al., 2019). The highest peak in observed and estimated freshwater diversity of isopods coincides with latitudes in which the most freshwater habitat is available, according to a global study on gradients within the freshwater biome, which revealed that river and lake densities are highest in northern hemisphere boreal latitudes (Dodds et al., 2019).

This study has shown that the LDG of isopods is bimodal, with diversity peaks outside the tropics. The gradient is markedly asymmetrical, viewed separately for the different subgroups analysed here. However, the gradient of the whole order is fairly symmetrical between hemispheres, as the higher northern hemisphere diversity of the terrestrial and freshwater subgroups is levelled by the high species richness of marine isopods in the southern hemisphere. The gradients depicted here also clearly show the differences between deep-sea and shallow-water richness patterns. While the diversity of shallow-water species is highest at intermediate latitudes and declines towards the poles, the species richness of deep-sea isopods reveals additional peaks in the polar seas, where deep-sea diversity exceeds shallow-water diversity. Further, even though sampling bias does not have a marked effect on the overall shape of the diversity gradient, estimated species richness predicts a higher diversity within low latitudes than is currently observed. Nevertheless, the bimodality of the gradient with a dip in species richness in equatorial regions is upheld. However, this reveals that more sampling for aquatic and terrestrial isopods is needed in tropical regions. Those regions might hold a good part of the diversity of isopods that has been overlooked so far.

There is likely no single explanatory mechanism or process underlying the pattern of the latitudinal diversity gradient of species richness (Condamine et al., 2012; Cruz-Motta et al., 2020; Gaston, 2000), especially since causes are presumed to be highly taxon-specific (Cerezer et al., 2022; Chaudhary et al., 2017). However, temperature may be assumed to be an important driver that is closely connected to various other mechanisms, which might play a role in forming and maintaining gradients in species richness (Boag et al., 2021; Chaudhary et al., 2021; Costello et al., 2023). Gaining a better understanding of the drivers behind the gradients will require improved methods that integrate fossil data and molecular phylogenies (Jablonski et al., 2017). When determining areas for conserving biodiversity, longitudinal variance in species richness must also be considered, as it differs within latitudinal bands (Currie et al., 1999). Several studies have also shown that the shape of the LDG varies between ocean basins or New World, Old World, and Australasian landmasses (e.g., Boltovskoy & Correa, 2017; Orr et al., 2021; Rivadeneira & Poore, 2020).

5. Global bioregionalisation and endemicity of isopods

5.1. Introduction

In addition to the well-documented latitudinal variations in species richness, global biodiversity exhibits other geographical patterns that have captured the attention of scientists since the time of Darwin and Wallace. Earth's biodiversity is not uniformly spread across all continents and oceans. Rather, some regions stand out as biodiversity hotspots, teeming with a remarkable profusion of species, while diversity in other areas is comparatively scarce. Unique species assemblages cluster in specific geographic areas and can vary significantly from assemblages in neighbouring regions. Biogeographic regions, or bioregions, do not conform to human societies' political or socio-economical borders and boundaries. Instead, they are delineated based on environmental factors and species distributions. Bioregionalisation builds a framework that can act as a basis for informed decision-making in conservation planning, e.g., by highlighting areas of immense species richness and high endemicity (Dinerstein et al., 2017; Lourie & Vincent, 2004). Historically, such efforts have been focused on the terrestrial realm (e.g., Wallace, 1876). Attempts to divide the world's oceans into distinct biogeographic regions started to emerge much later, at first with little evidence of distinct boundaries (Briggs, 1974; Ekman, 1953). Today, detailed maps of biogeographic regions (often termed ecoregions, nested within biomes and realms) have been produced for the terrestrial environment (Olson et al., 2001), the freshwater realm (Abell et al., 2008), and the coastal and shelf areas of the world's oceans (Spalding et al., 2007) as well as for pelagic surface waters (Spalding et al., 2012). However, within the world's oceans, it has been recognised that a third dimension – depth – also plays an important role. Therefore, separate biogeographic frameworks for the mesopelagic zone (Sutton et al., 2017) and the deep sea (Watling et al., 2013) have been produced.

Many of the above-mentioned extensive biogeographic studies primarily focus on environmental criteria and topographic features, sometimes relegating species distributions to a secondary role. However, Costello et al. (2017) took a different approach in creating a comprehensive global map of coastal and offshore oceanic realms by analysing occurrence records of 65,000 marine species. Notably, a substantial portion of their identified realms closely corresponded to higher-level regions previously outlined by Spalding et al. (2007, 2012) and Watling et al. (2013). It is essential to acknowledge that each taxon has its own evolutionary history and dispersal capabilities, resulting in distinct distribution patterns and diversity hotspots. Accordingly, several studies have concentrated on the biogeographic distribution of specific taxa, often comparing their results to the proposed bioregions of previously mentioned general biogeographic frameworks (Arfianti & Costello, 2020; Bribiesca-Contreras et al., 2019; Pamungkas et al., 2021; Victorero et al., 2023; Watling & Lapointe, 2022). While regional assessments of species richness of various taxonomic and ecological groups within the order Isopoda are abundant in the scientific literature, no comprehensive global analysis of the distribution of isopod species has yet been performed for aquatic or terrestrial isopods.

Kensley (2001), for instance, conducted a biogeographic study of Indian Ocean isopods based on a compiled list of approximately 1,000 species, most of which were shallow-water records. Given that isopods are predominantly benthic brooders with limited dispersal abilities due to the absence of a planktonic larval phase, he noted that wide-ranging species are scarce, and local endemism is expected to be high. Similarly, in a study examining the distribution ranges of deep-sea peracarids, including isopods, Brandt et al. (2012) concluded that only very few, if any, peracarid species are truly widespread. They suggested that many species assumed to be wide-ranging may comprise cryptic species complexes, as has been demonstrated in molecular studies of several isopod species (Held, 2003; Hurtado et al., 2016; Raupach et al., 2007). However, long-distance dispersal can be achieved by rafting (Gutow et al., 2006; Leese et al., 2010) or, in the case of parasitic species, by attachment to highly mobile hosts (Hadfield & Smit, 2020; Nicholson et al., 2020). Kensley (2001) classified approximately 84% of the studied isopod species as endemic to the Indian Ocean as a whole, with varying levels of endemism within sub-regions, ranging from 46% to 79%. In the species-rich Southern Ocean, 87% of collected isopod species are assumed to be endemic to the region (Brandt, Brix, et al., 2007; Brandt, De Broyer, et al., 2007). Similarly, Guzik et al. (2019) observed strong regional endemicity for groundwater-associated *Haloniscus* species in Australia. Within the terrestrial realm, rates of endemism are reported, for example, for Oniscidea in Greece at 69% (Sfenthourakis & Giokas, 1998) or for North America at 66.1% (Jass & Klausmeier, 2000). However, at finer spatial scales, endemism rates unsurprisingly tend to be lower. For instance, Sfenthourakis (1996) documented 20% of oniscidean species from the central Aegean islands as endemic, a similar proportion to other Mediterranean archipelagos and indicative of geologically recent isolation from the mainland. Likewise, the observed percentage of endemism in the Transdanubian region of western Hungary is 16% (Hornung et al., 2008).

Faunal influences from neighbouring regions highlight Transdanubia's role as a diverse biogeographical crossroads for terrestrial isopods. It will be interesting to see how marine biogeographic patterns and endemicity differ from their terrestrial counterparts in the global assessment of isopod occurrences herein.

An ongoing debate is whether dispersal or vicariance is the most crucial mechanism shaping modern biogeographic patterns. However, considering both mechanisms can best explain large-scale distribution patterns (Brusca, 1983b, 1984). Plate tectonics resulting in continental drift and the opening of new oceans play an essential role in driving deep biological separation of biota (Ficetola et al., 2017; Wägele et al., 1995; G. D. F. Wilson, 2008b). In addition, environmental variables such as temperature, salinity, and precipitation have been identified as equally important factors underlying modern biogeographic patterns (Belanger et al., 2012; Victorero et al., 2023). Apart from this, Wallerstein and Brusca (1982) proposed that biotic interactions such as predator-prey dynamics also shape species' distribution ranges, predominantly on local to regional scales. Topography is another determinant of extant biogeographical boundaries (Ficetola et al., 2017). Marine ridges and trenches, for example, may present a dispersal barrier, especially for non-swimming isopods (Bober et al., 2018; Johannsen et al., 2020). However, several studies have also shown that distributions of many isopod species cross certain well-known biogeographic barriers (e.g., Pearman et al., 2020; Schnurr et al., 2014).

Although there are many regional studies on the biogeography of various isopod taxa and ecological groups (e.g., Brusca, 1987; Castelló et al., 2020; Kensley, 2001; Sfenthourakis & Giokas, 1998; Wägele, 1990), no comprehensive global-scale analysis has yet been carried out. Copious occurrence records are available in the Ocean Biogeographic Information System (OBIS, https://obis.org) and the Global Biodiversity Information Facility (GBIF, https://www.gbif.org). This study makes use of those data and examines global-scale bioregionalisation within Isopoda. Based on differences in species composition, biogeographic regions, or bioregions, are delineated for the marine, freshwater, and terrestrial realms, and each region's endemicity is determined.

5.2. Methods

5.2.1. Data source

The dataset of global occurrence records used here to analyse biogeographic regions is the same dataset compiled for Chapter 4. Occurrence records were obtained from OBIS and GBIF on 10th June 2022. For information on processing and cleaning of the data, see section 4.2.1. in Chapter 4. The cleaned dataset contains 388,881 occurrence records of 5,935 isopod species (Hartebrodt, 2023a).

5.2.2. Data analysis

"Infomap Bioregions" (https://www.mapequation.org/bioregions/) is an interactive web application that applies network theory to identify taxon-specific biogeographic regions (bioregions) from species distribution data (Edler et al., 2017). First, the input data are binned into geographical grid cells, which creates a bipartite network between species and grid cells. Then, the Infomap algorithm clusters occupied grid cells with sufficient data points into bioregions (Edler et al., 2017, and references therein). The similarity of grid cells and clustering into bioregions is based on their species composition. Therefore, a bioregion – as for the purpose of this study – is defined as a geographic area that harbours a distinct community of isopod species, which differs from that in other regions. Here, for these basic bioregionalisations, which are a first step towards a more detailed understanding of global isopod biogeography, environmental variables are not taken into account for the structuring of bioregions. The application "Infomap Bioregions" also identifies the most common and the most indicative species with the highest relative abundance in each derived bioregion. In the output data, indicative species are listed with a score, which is defined as the ratio between the frequency of the species in the bioregion and its frequency in all regions (Edler et al., 2017). For example, an indicative score of 2 means that a species is twice as frequent in a specific bioregion than in the entire dataset. This information helps to identify endemic species within bioregions. Spatial resolution was set to 4° latitudinal-longitudinal grid cells to achieve the closest possible similarity to the 5° spatial resolution used in the analysis of the same dataset in the previous chapter (Chapter 4 – Latitudinal diversity gradients), and the clustering algorithm trials were set to 5 allowing several runs to find the best solution. Before analysis, the dataset of isopod occurrence records was split into three separate datasets according to habitat information. The resulting terrestrial dataset contains 187,694 occurrences, the freshwater dataset has 40,777 occurrence records, and the marine dataset consists of 160,410. The different datasets were then uploaded separately to "Infomap Bioregions" to produce distinct maps of global bioregions. The maximum and minimum cell capacity were set to 100 and 50 for terrestrial isopods, respectively. Following initial analyses, the cluster cost was set to 1.0 as a higher cluster cost of 1.5 would have clustered all of North America and Europe into a single bioregion, likely owing to the inclusion of cosmopolitan and circumglobal isopod species like *Armadillium vulgare* (Latreille, 1804), *Porcellio scaber* Latreille, 1804, and *Oniscus asellus* Linnaeus, 1758. Since the freshwater dataset provided mostly low numbers of occurrence records per grid cell, maximum cell capacity was chosen at 50, minimum at 10, and a cluster cost of 1.5. Settings for marine isopod occurrences were 100 maximum cell capacity, 50 minimum cell capacity, and 1.5 cluster cost. Since depth adds to the complexity of the marine environment, another dataset was prepared with all marine occurrence records that had information on the depth of occurrence. This yielded a dataset containing 83,313 records, which was then further divided into three different depth categories: shallow (0 to 200 m), intermediate (>200 to 500 m), and deep (>500 m). Each depth dataset was also uploaded to and analysed with "Infomap Bioregions" with the following settings: 100 maximum cell capacity, 50 minimum cell capacity, and 1.5 cluster cost. The percentage of unique species for each resulting bioregion was also calculated to explore endemicity patterns.

5.3. Results

5.3.1. The marine realm

The marine dataset contains occurrence records for 4,499 species in 799 genera and 87 families. That covers approximately 73% of all marine species listed in the global list of isopod species (Hartebrodt, 2023b) compiled from WoRMS data in Chapter 3. The coverage for marine isopod families is close to 96%, and almost 88% of all marine genera are included here. The clustering algorithm of "Infomap Bioregions" yielded 33 distinct bioregions (Fig. 5.1) when the complete dataset of marine isopods was analysed. Some of these are extensive and represent realistic biogeographic realms as they have been delineated in previous biogeographic classifications, like Bioregion 3, which covers the entire Southern Ocean and a few adjacent areas. Bioregion 6 stretches along the entire west coast of North America in the North Pacific Ocean. However, the spatially most extensive bioregion is Bioregion 1, which encompasses the entire Arctic Ocean but also includes the Baltic Sea, the North Sea, the Mediterranean Sea, the Black Sea, and several spots in the Atlantic Ocean. Other bioregions are based on only a single 4° grid cell, many of which are relatively isolated archipelagos like Hawaii (Bioregion 21), the Galapagos Islands (Bioregion 30), or New Caledonia (Bioregion 17). Other single-cell bioregions likely represent more extensive but under-sampled areas with unique species composition. For example, the Persian Gulf (Bioregion 28) is one such region. The Gulf of Aden and the Arabian Sea (Bioregion 24) are another example, as well as the Laccadive Sea and adjacent areas at the southern tip of India (Bioregion 18). Interestingly, the Strait of Gibraltar (Bioregion 31), the connection between the Atlantic Ocean and the Mediterranean Sea (of which both neighbouring cells are grouped in Bioregion 1), was classified as a distinct bioregion by the clustering algorithm. However, this might be an artefact of insufficient data, as the Strait of Gibraltar grid cell harbours precisely the minimum amount of occurrence records (50) to be included in the analysis and the top most indicative species, which were exclusively recorded in this region, are only represented by one or two occurrence records in the whole dataset (see Table 5.1). The most common species in Bioregion 31, with 32 occurrences, is *Natatolana borealis* (Liljeborg, 1851), of which most of its other occurrence records fall into Bioregion 1. The most species-rich bioregion is Bioregion 2 (656 recorded species; see Table 5.1), which encompasses most of the northwest Atlantic, the Gulf of Mexico, the Caribbean Sea and areas in the southwest Atlantic along the Brazilian coast. It is followed by Bioregion 4 (southern Australia), Bioregion 1 (Arctic Ocean and adjacent seas), Bioregion 3 (Southern Ocean and adjacent areas), and Bioregion 7 (northern Australasian region + one African grid cell). The Red Sea, which could have been expected to form its own bioregion due to its relative isolation, did not have a single grid cell with sufficient data to be included in the analysis. Endemicity ranged from 27% in Bioregion 15 (coasts of Uruguay and the Buenos Aires Province, Argentina) to 87% in Bioregion 22 (off the southwestern African coast). The Southern Ocean and adjacent areas (Bioregion 3) also had one of the highest endemism rates, with 80% (see Table 5.1). Like the Southern Ocean, Australia has previously been reported as a region of high endemism for marine isopods. In this analysis, the seas around Australia are divided into a southern bioregion (Bioregion 4), in which 71% of species are endemic, and a northern region (Bioregion 7) with 64% endemism. The most widespread marine species in the analysis was *Idotea metallica* Bosc, 1801, an obligate rafter with enhanced dispersal ability (Gutow et al., 2006), which has been recorded in 14 of the 33 bioregions (see Table 5.2). Many other widespread species are wood-boring or burrowing isopods that may disperse passively within dislodged plant material. Others are parasites or temporarily attached micropredators of highly mobile fish hosts.

Table 5.1. Bioregions of marine Isopoda. For each region, the most common species, i.e. the species with the most occurrence records within the region, is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	95,116	505	110	64	*Saduria entomon*	*Rocinela danmoniesis, Jaera (Jaera)ischiosetosa, Microcharon harrisi, Dendrotion setosum, Eurydice dollfusi*
2	18,956	656	42	67	*Edotia triloba*	*Edotia acuta, Chiridotea arenicola, Harrieta faxoni, Erichsonella isabelensis, Sphaeroma papillae*
3	5,070	502	40	80	*Glyptonotus antarcticus*	*Neastacilla marionensis, Munna neglecta, Pleurosignum magnum, Paranthura possessia, Edotia tangaroa*
4	10,303	570	15	71	*Bullowanthura pambula*	*Bullowanthura pambula, Natatolana corpulenta, Amakusanthura olearia, Serolina acaste, Ulakanthura lara*
5	4,209	373	12	75	*Leptanthura laevigata*	*Metacirolana arnaudi, Cirolana sulcata, Austroarcturus africanus, Austroarcturus quadriconus, Cirolana rugicauda*
6	6,859	250	20	62	*Pentidotea wosnesenskii*	*Idarcturus allelomorphus, Colidotea rostrata, Cortezura penascoensis, Synidotea media, Ianiropsis epilittoralis*
7	3,463	458	12	64	*Onychatrium forceps*	*Apanthura restio, Onychatrium forceps, Bathynomus immanis, Joeropsis goobita, Cirolana capricornica*
8	1,585	192	8	62	*Isocladus armatus*	*Natatolana aotearoa, Limnoria reniculus, Scutuloidea maculata, Bullowanthura crebrui, Dynamenopsis varicolor*

Table 5.1. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
9	1,685	151	8	68	*Cirolana harfordi*	*Cyathura higoensis, Gnorimosphaeroma naktongense, Synidotea hikigawaensis, Leptosphaeroma gottschei, Koridotea hoonsooi*
10	637	120	3	33	*Chelator insignis*	*Bathybadistes hoplitis, Prochelator litus, Mirabilicoxa acuminata, Eugerda pannosa, Momedossa profunda*
11	242	84	3	86	*Microcope ovata*	*Eugerdella hadalis, Stylomesus malyutinae, Mastigoniscus latus, Chaetarcturus pacificus, Dendromunna okhotensis*
12	364	96	3	51	*Expanathura macronesia*	*Apanthuroides calculosa, Apanthuroides aldabrae, Amakusanthura cosmoledo, Joeropsis dimorpha, Anthomuda quadrilineata*
13	318	72	3	78	*Haploniscus bruuni*	*Eurycope manifesta, Ilyarachna pervica, Mastigoniscus concavus, Chaetarcturus praecipius, Cyproniscus octospinosus*
14	178	71	2	55	*Athelges takanoshimensis*	*Aporobopyrus retrorsa, Minicopenaeon intermedium, Renocila kohnoi, Renocila yamazatoi, Pleurocryptella laevis*
15	197	44	2	27	*Uromunna peterseni*	*Uromunna peterseni, Leptoserolis sheppardae, Munnogonium quequensis, Cassidias argentinea, Macrochiridothea lilianae*
16	137	52	2	58	*Accalathura phuketensis*	*Eophrixus brevicauda, Pendanthura siamensis, Gnathia serrula, Cerceis bicarinata, Schizobopyrina kossmanni*

Table 5.1. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
17	119	50	1	62	*Chaetarcturus crosnieri*	*Leptanthura segonzaci, Pseudione clevai, Pendanthura anophthalma, Eragia profunda, Sedorolis simplex*
18	75	52	1	42	*Mothocya renardi*	*Pleurocope dasyura, Cymothoa parupenei, Nerocila loveni, Rhiothra callipia, Cymodoce bicarinata*
19	63	39	1	46	*Dolicholana enigma*	*Bopyrione longicapitata, Eophrixus shojii, Bopyrinella albida, Bopyrione toloensis, Apophrixus constrictus*
20	115	34	1	65	*Neonaesa rugosa*	*Hansenium hanseni, Bourbonanthura vaitapensis, Munna temae, Gnathostenetroides polynesica, Liocoryphe algreti*
21	256	38	1	45	*Neonaesa rugosa*	*Mesanthura hieroglyphica, Paranthura bellicauda, Creniola breviceps, Joeropsis hawaiiensis, Colidotea edmondsoni*
22	76	31	1	87	*Acanthocope galatheae*	*Nannoniscus antennaspinis, Regabellator abyssi, Pseudomesus pitombo, Nannoniscus meteori, Eurycope tumidicarpus*
23	100	34	1	44	*Ianiropsis longipes*	*Munna varians, Iais elongata, Vermectias caudiculata, Ianiropsis longipes, Neojaera hirsuta*
24	70	23	1	43	*Cassidias africana*	*Oxinasphaera furcata, Baharilana lira, Metacirolana chemola, Cirolana somalia, Elaphognathia gladia*

Table 5.1. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
25	50	24	1	58	*Cymothoa pulchrum*	*Zeuxokoma luetzeni, Mastigoniscus microcephalus, Gnathia camuripenis, Cterissa sakaii, Munnopsis megacephalus*
26	74	29	1	41	*Natatolana rossi*	*Sporonana litoralis, Spiculonana petraea, Paramunna snaresi, Cymodoce allegra, Limnoria loricata*
27	51	23	1	70	*Apanthura stocki*	*Pendanthura tinggiensis, Tinggianthura alba, Mesanthura asiatica, Pendanthura tiomanensis, Asymmetrione sallyae*
28	50	20	1	65	*Cymodoce fuscina*	*Cymodoce fuscina, Heterodina mccaini, Gnathia luxata, Eurydice paxilli, Caenanthura enigmatica*
29	54	20	1	65	*Macrostylis bipunctatus*	*Stylomesus spinulosus, Ischnomesus paucispinis, Ischnomesus magnificus, Ischnomesus bidens, Haploniscus minutus*
30	52	15	1	60	*Nerocila californica*	*Aegiochus longicornis, Rocinela wetzeri, Aegiochus francoisae, Antarcturus multispinis, Aega acuminata*
31	50	13	1	46	*Natatolana borealis*	*Astacilla paucisaetosa, Astacilla cingulata, Astacilla poorei, Astacilla bonnierii, Stenosoma raquelae*
32	66	10	1	50	*Haploniscus silus*	*Haploniscus silus, Chauliodoniscus tasmanaeus, Hydroniscus lobocephalus, Bathybadistes andrewsi, Haploniscus saphos*

Table 5.1. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
33	61	10	1	60	*Brucerolis brandtae*	*Haploniscus miccus, Prochelator tupuhi, Notopais zealandica, Epikopais mystax, Rocinela pakari*

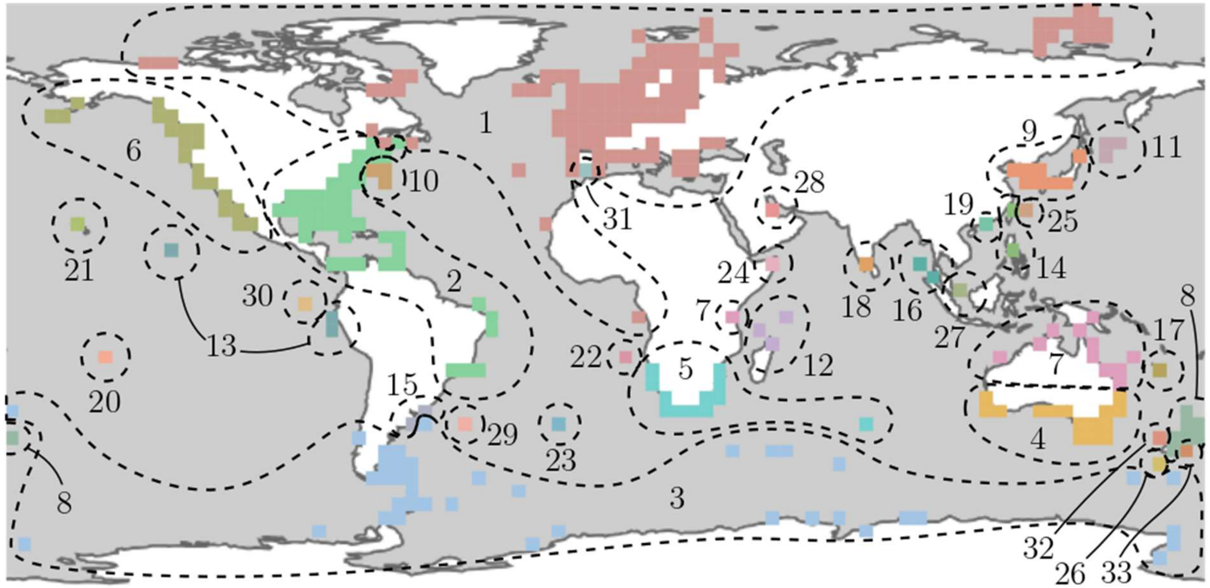

Figure 5.1. Bioregions of marine Isopoda. The coloured blocks are all those grid cells that had sufficient data points to be included in the analysis. Blocks of the same colour were clustered together into a bioregion by the Infomap algorithm. The dashed lines are only meant to serve enhanced clarity and do not represent actual bioregion boundaries. For more information on each bioregion, see Table 5.1.

Table 5.2. List of marine isopod species that were recorded in more than five bioregions.

Species	Number of bioregions occupied	List of occupied bioregions
Idotea metallica	14	1, 2, 3, 4, 5, 6, 7, 8, 9, 10, 12, 15, 23, 32
Limnoria tripunctata	9	1, 2, 4, 5, 6, 7, 9, 15, 21
Paracerceis sculpta	8	2, 4, 6, 7, 9, 15, 19, 21
Sphaeroma serratum	8	1, 2, 4, 5, 12, 15, 16, 18
Iais pubescens	8	2, 3, 4, 5, 7, 8, 23, 26
Sphaeroma walkeri	8	2, 4, 5, 6, 7, 19, 21, 27
Elthusa raynaudii	7	4, 5, 7, 8, 9, 14, 23
Sphaeroma terebrans	7	2, 4, 5, 7, 12, 14, 16
Paradella dianae	7	2, 4, 6, 7, 10, 19, 21
Cirolana harfordi	7	2, 4, 6, 8, 9, 19, 30
Nerocila orbignyi	7	1, 2, 4, 5, 7, 8, 24
Lanocira gardineri	7	5, 7, 12, 14, 19, 24, 28
Limnoria quadripunctata	6	1, 2, 4, 5, 6, 8
Cirolana parva	6	2, 5, 6, 7, 18, 21
Limnoria indica	6	1, 4, 7, 12, 16, 18
Paralimnoria andrewsi	6	2, 7, 9, 12, 21, 25

In the marine realm, depth adds another dimension and more complexity to the environment. The marine dataset was divided into three depth categories to examine how bioregionalisation within isopods changes with depth. Of the 4,177 species for which depth information was available, ~6% are eurybathic and were sampled in all three depth categories (Fig. 5.2). A further 55 species are shared between the deep and shallow datasets but are absent from the intermediate one. The deep and intermediate datasets share 144 species not documented in shallow waters. 207 species are shared between the shallow and intermediate datasets but were not sampled deeper than 500 m.

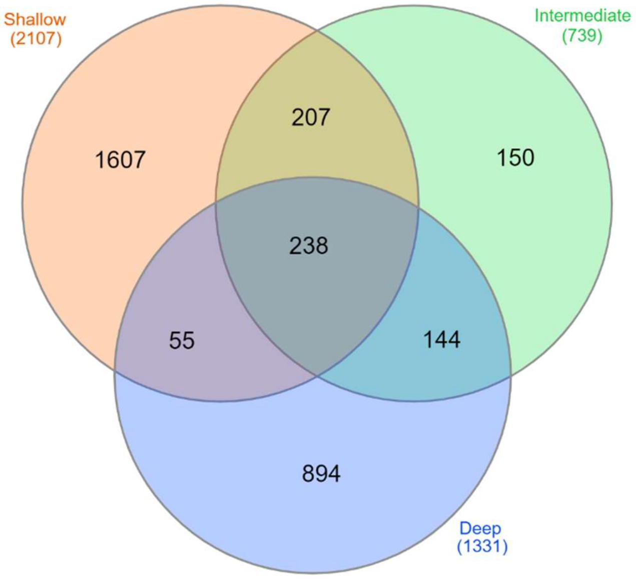

Figure 5.2. Venn diagram of marine isopod species shared between the shallow (0 to 200 m), intermediate (>200 to 500 m) and deep (>500 m) datasets.

Most species were recorded from shallow waters, and ~71% of marine occurrence records fall in this depth category. For these, 14 bioregions could be identified (Fig. 5.3a). The most extensive bioregion still encompasses the Arctic Ocean, parts of the North Atlantic, the Baltic Sea, the North Sea, the Mediterranean Sea, and the Black Sea (here Bioregion 2). The Southern Ocean and adjacent areas (Bioregion 5) are still recognised as one extensive bioregion, too, as is the west coast of North america in the North Pacific Ocean (Bioregion 4). Most of the northwest Atlantic, the Gulf of Mexico, and the Caribbean Sea continue to group together (Bioregion 1). However, a grid cell along the Brazilian coast (Bioregion 14) is now recognised as a bioregion with a distinct species composition. The coastal waters around Australia are still divided into south (Bioregion 3) and north. However, for shallow-water species only, distinct species assemblages cluster in north-eastern (Bioregion 8) and north-western Australia (Bioregion 11). Endemicity in shallow-water regions ranges from 53% in Bioregion 12 (Hawaii) to 88% in Bioregions 5 (Southern Ocean) and 3 (southern Australia) (see Table C1).

The intermediate dataset contains approximately 17% of marine occurrence records and only 739 species, of which most are shared with the other two depth categories (Fig. 5.2). Only eight bioregions can be identified based on these data (Fig. 5.3b). Again, the most extensive bioregion is the Arctic Ocean, with adjacent areas in the northern North Atlantic. There are insufficient occurrence records for intermediate depths in the North, Baltic, Mediterranean, and Black Seas to test whether they would still group with more northern ocean regions. Both bioregions on the west and east coast of North America are maintained. However, there is insufficient occurrence data from the Gulf of Mexico and the Caribbean Sea to see whether they persistently cluster with the rest of the northwest Atlantic. Southern Australian and New Zealand bioregions are sustained. The Southern Ocean clusters into three distinct bioregions with data from the intermediate dataset. Herein, species assemblages from the Weddell Sea margins differ from communities at the edge of the Ross Sea and assemblages from an adjacent area in the southern Indian Ocean. Endemism rates range from 50% in the northwest Atlantic to 100% in the Australian bioregion (see Table C2).

Approximately 12% of marine occurrence records are part of the deep dataset. Nevertheless, the deep-sea data show high species richness, with ~32% of species occurring deeper than 500 m and ~21% exclusively in this depth category (Fig. 5.2). From these data, 16 bioregions can be identified (Fig. 5.3c). Based on deep-sea isopod species, north-eastern Atlantic regions that group with the extensive Arctic Ocean bioregion in the shallow water and the complete marine datasets now form a distinct bioregion. The deep dataset also contains sufficient occurrence records within grid cells in the Gulf of Mexico and along the eastern North American coast to compare the regions, and the clustering algorithm of “Infomap Bioregions” groups them into two distinct biogeographic regions. Within the Southern Ocean, only two grid cells at the edge of the Weddell Sea contain enough data to be included in the analysis. Deep-sea isopods sampled around New Zealand cluster into three distinct bioregions, whereas New Zealand was a single bioregion for isopods at shallow and intermediate depths. The percentage of endemic deep-sea isopods ranges from 20% in the low-diversity bioregion off the North American west coast to 100% in the deep sea surrounding New Caledonia and a grid cell in the southern Indian Ocean. Furthermore, Australia and the Southern Ocean continue to stick out as high-endemism areas, with 90% and 87% endemicity, respectively (see Table C3).

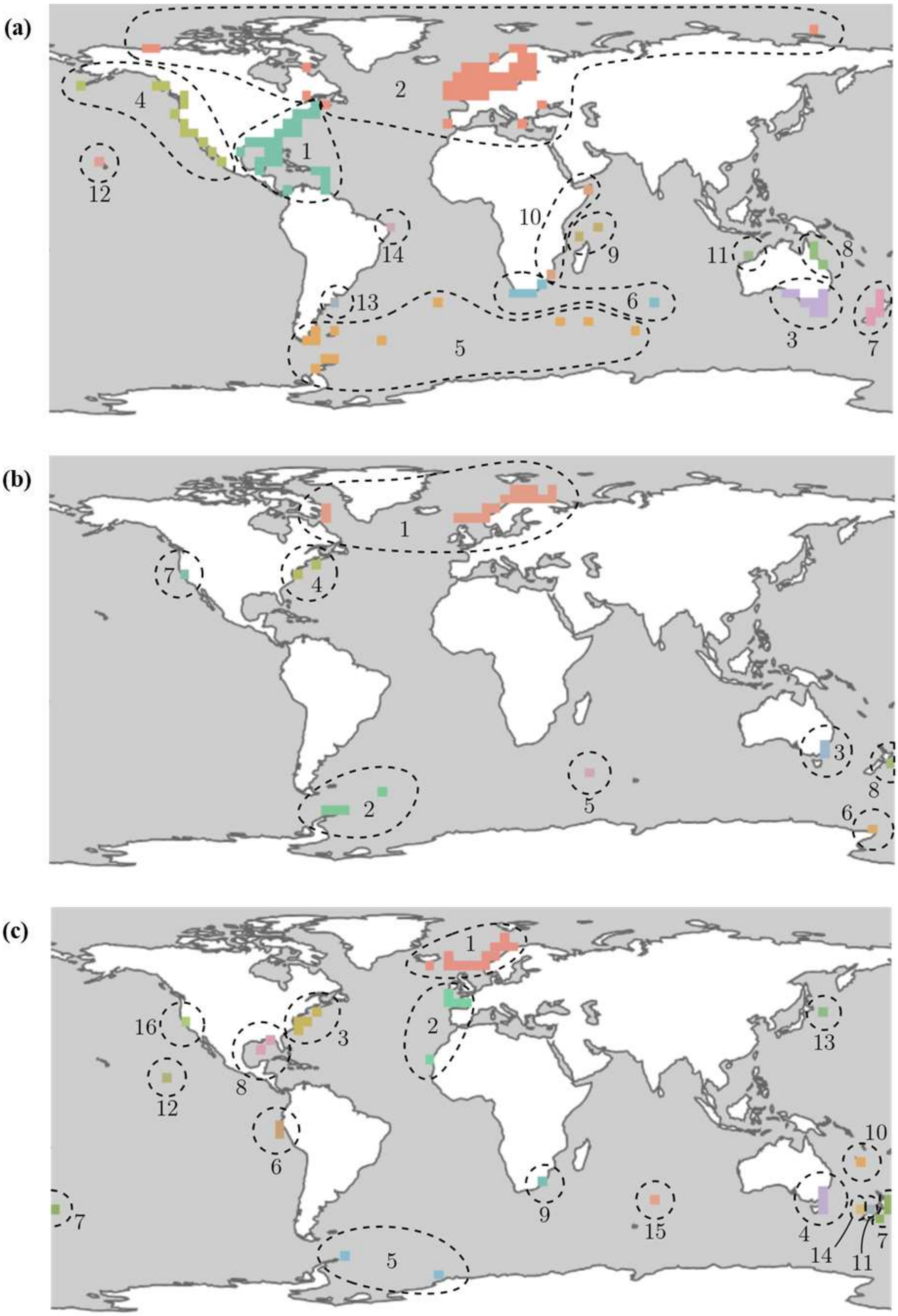

Figure 5.3. Bioregionalisation within the different marine depth categories. **(a)** Shallow: 0 to 200 m, **(b)** intermediate: >200 to 500 m, and **(c)** deep: >500 m. The coloured blocks are all those grid cells that had sufficient data points to be included in the analysis. Blocks of the same colour were clustered together into a bioregion by the Infomap algorithm. The dashed lines are only meant to serve enhanced clarity and do not represent actual bioregion boundaries. For more information on each bioregion, see Table C1, C2 and C3, respectively.

5.3.2. The terrestrial realm

The cleaned dataset contains occurrence records for only about a third of all terrestrial isopod species (~27%). However, 79% of families and approximately 43% of all terrestrial genera are included. For the terrestrial realm, 28 bioregions were recognised (Fig. 5.4). The most extensive bioregion is Bioregion 1, which includes the majority of occurrences for cosmopolitan or circumglobal species like *Armadillium vulgare* (Latreille, 1804), *Porcellio scaber* Latreille, 1804, and *Oniscus asellus* Linnaeus, 1758. Bioregion 1 encompasses most of North America and mostly the northern parts of Europe and stray areas in South America and Korea. *A. vulgare* is the most common species in 15 of the 28 bioregions (see Table 5.3). Some bioregions in the current analysis can be characterised as well-defined e.g. Hawaii (Bioregion 24), the Azores archipelago (Bioregion 12), Sicily (Bioregion 23), Lord Howe Island (Bioregion 17), New Zealand (Bioregion 15), or Australia (Bioregion 7), which has only sufficient data for the south-eastern part, including Tasmania. Grid cells of other bioregions are sometimes spread over extensive, entirely unconnected areas like the stray cells of Bioregion 1 and 6. The terrestrial dataset has the lowest coverage regarding species numbers and consequently seems more strongly affected by sampling bias than the marine dataset. Most occurrence records are concentrated in Europe, especially in north-western Europe and a few areas in the United States. Species richness is also highest in Europe, with the highest species number recorded in Bioregion 2 (mostly south-eastern France), followed by Bioregion 1 (mainly Europe and North America), Bioregion 5 (north-eastern Iberian Peninsula), and Bioregion 3 (parts of eastern Europe). Figure 5.4 shows vast areas with insufficient occurrence records. On the African continent, only the area around Cape Town (Bioregion 21) had enough data to be included in the analysis, and Madagascar is entirely blank. Similarly, only a few grid cells were sufficiently sampled in South America. Most of Asia and all of Indonesia are completely blank on the map in Figure 5.4. In Australia, only grid cells in the southeastern part of the continent had enough data to be considered in this analysis. Endemism rates of terrestrial bioregions ranged from 12% in Bioregion 12 (the Azores archipelago) to 95% in Bioregion 22 (Socotra Island). High endemism on an island seems intrinsically logical in organisms with low dispersal abilities. However, no neighbouring grid cells have been analysed here to examine how distinct the isopod fauna of Socotra Island is to the Arabic mainland or the fauna of Somalia. Another island that forms its own bioregion is Lord Howe Island off eastern Australia (Bioregion 17). 79% of the 29 species recorded for the region in this dataset are endemic to the area (see Table 5.3). In this case, neighbouring grid cells of the nearest mainland had sufficient data and clustered into a separate bioregion (Bioregion 7), which shows equally high endemicity (76%). Despite these high rates of endemism, terrestrial isopods tended to be more widespread than marine species. More than 100 terrestrial species were recorded in three or more bioregions. The two most widespread species, *Armadillidium vulgare* (Latreille, 1804) and *Porcellionides pruinosus* (Brandt, 1833), were both recorded in 25 of the 28 delineated bioregions (see Table 5.4). Several regional subspecies are described of the latter species.

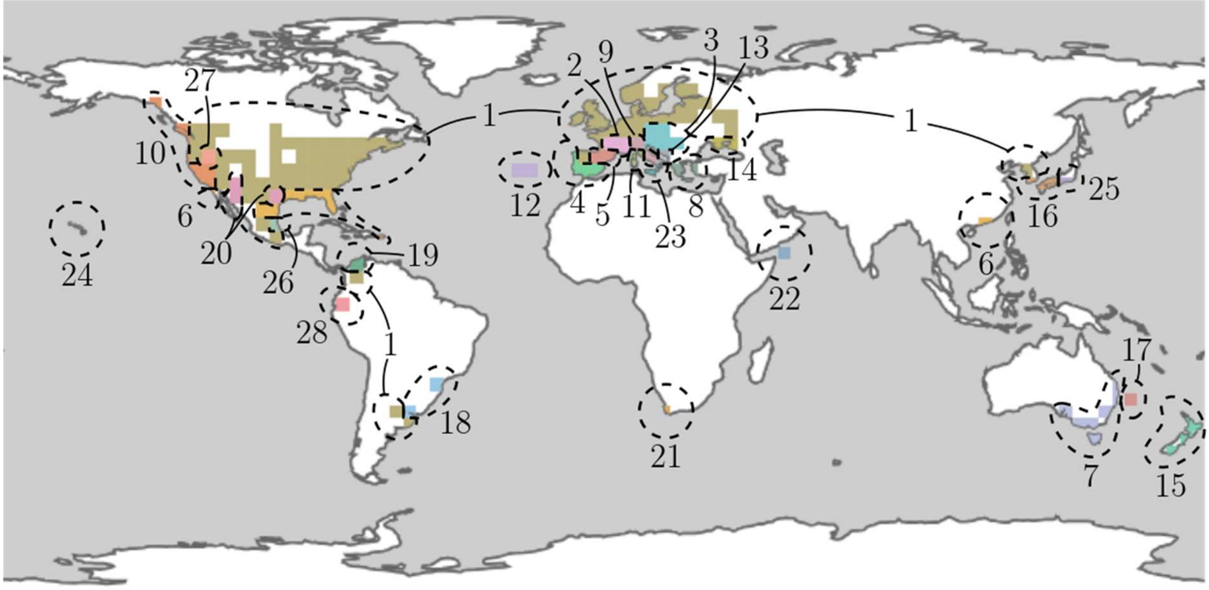

Figure 5.4. Bioregions of terrestrial Isopoda. The coloured blocks are all those grid cells that had sufficient data points to be included in the analysis. Blocks of the same colour were clustered together into a bioregion by the Infomap algorithm. The dashed lines are only meant to serve enhanced clarity and do not represent actual bioregion boundaries. For more information on each bioregion, see Table 5.3.

Table 5.3. Bioregions of terrestrial Isopoda. For each region, the most common species is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	141,288	127	89	15	*Oniscus asellus*	*Ligidium elrodii, Metatrichoniscoides celticus, Porcellio novus, Trichoniscus demivirgo, Miktoniscus racovitzai*
2	7,642	149	3	28	*Philoscia muscorum*	*Oritoniscus virei, Caeroplastes porphyrivagus, Armadillidium quinquepustulatum, Buddelundiella zimmeri, Trichoniscus voltai*
3	3,671	102	5	30	*Armadillidium vulgare*	*Hyloniscus adonis, Haplophthalmus austriacus, Armadillidium carniolense, Tachysoniscus austriacus, Trichoniscus nivatus*
4	1,230	61	4	34	*Armadillidium vulgare*	*Trichoniscoides machadoi, Proporcellio mirabilis, Eluma tuberculata, Porcellionides rufocinctus, Oniscus lusitanus*
5	1,969	116	2	29	*Armadillidium vulgare*	*Oritoniscus trajani, Porcellio duboscqui, Oritoniscus intermedius, Oritoniscus bonneti, Trichoniscoides modestus*
6	2,411	37	10	32	*Armadillidium vulgare*	*Trichorhina donaldsoni, Venezillo culebrae, Portoricoscia richmondi, Ligidium floridanum, Synuropus granulatus*
7	1,986	51	8	76	*Armadillidium vulgare*	*Actaecia thomsoni, Cubaris hickmani, Styloniscus hirsutus, Styloniscus maculosus, Styloniscus nichollsi*
8	611	61	3	54	*Armadillidium vulgare*	*Armadillidium insulanum, Orthometopon turcicum, Orthometopon phaleronense, Ligidium werneri, Armadillidium aegaeum*

Table 5.3. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
9	643	93	2	28	*Armadillidium vulgare*	*Armadillidium furcatum, Armadillidium tirolense, Armadillidium ponalense, Armadillidium marmorivagum, Lepidoniscus pruinosus*
10	9,365	35	8	26	*Armadillidium vulgare*	*Ligidium lapetum, Ligidium latum, Venezillo microphthalmus, Alloniscus mirabilis, Armadilloniscus lindahli*
11	1,635	95	1	36	*Porcellio orarum*	*Tiroloscia corsica, Cylisticus vandeli, Tiroloscia macchiae, Oritoniscus ocellatus, Platyarthrus corsicus*
12	6,014	33	2	12	*Eluma caelata*	*Chaetophiloscia guernei, Porcellio laevissimus, Armadillidium amicorum, Miktoniscus chavesi, Cordioniscus stebbingi*
13	265	62	1	34	*Armadillidium klugii*	*Alpioniscus magnus, Armadillidium scaberrimum, Armadillidium stagnoense, Echinarmadillidium fruxgalii, Armadillidium saxivagum*
14	115	22	2	23	*Armadillo officinalis*	*Trachelipus kervillei, Schizidium reinoehli, Cylisticus rotabilis, Tauronethes lebedinskyi, Trachelipus lutshnikii*
15	1,081	17	4	53	*Porcellio scaber*	*Spherillo rufomarginatus, Cubaris ambitiosa, Tylos neozelanicus, Cubaris tarangensis, Styloniscus commensalis*
16	360	30	3	60	*Ligia (Megaligia) exotica*	*Ligidium koreanum, Lucasioides nishimurai, Lucasioides ashiuensis, Mongoloniscus maculatus, Venezillo longispinus*

Table 5.3. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
17	186	29	1	79	*Cubaris crenata*	*Cubaris crenata, Australiodillo anomalus, Stigmops howensis, Cubaris hirsuta, Australiodillo armus*
18	305	28	2	46	*Armadillidium vulgare*	*Alboscia jotajota, Benthana schmalfussi, Benthana carijos, Calycuoniscus compar, Neotroponiscus daguerrii*
19	119	24	1	63	*Tylos niveus*	*Trichorhina bermudezae, Littorophiloscia amphindica, Agnara madagascariensis, Armadilloniscus caraibicus, Ischioscia unicartagenae*
20	2,078	18	3	17	*Armadillidium vulgare*	*Brackenridgia cavernarum, Brackenridgia sphinxensis, Brackenridgia reddelli, Venezillo arizonicus, Platyarthrus aiasensis*
21	118	27	1	63	*Armadillidium vulgare*	*Gerufa hirticornis, Marioniscus spatulifrons, Venezillo furcatus, Venezillo rufescens, Venezillo pumilus*
22	80	22	1	95	*Socotroniscus sacciformis*	*Socotroniscus sacciformis, Ligia dioscorides, Uluguroscia pohli, Pseudoagnara wraniki, Uluguroscia obscura*
23	68	26	1	31	*Armadillo officinalis*	*Armadillidium decorum, Porcellio albicornis, Armadillidium calabricum, Armadillidium siculorum, Platyarthrus briani*
24	141	19	1	42	*Porcellio laevis*	*Ligia rolliensis, Burmoniscus mauritiensis, Ligia mauinuiensis, Ligia pele, Australophiloscia societatis*

Table 5.3. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
25	130	18	1	33	*Armadillidium vulgare*	*Mongoloniscus vannamei, Venezillo hasegawai, Leptophiloscia kiiensis, Burmoniscus dasystylus, Littorophiloscia nipponensis*
26	61	9	1	22	*Armadillidium vulgare*	*Mexiconiscus laevis, Cylindroniscus vallesensis, Brackenridgia bridgesi, Cubaris murina, Porcellio laevis*
27	93	7	1	29	*Armadillidium vulgare*	*Oregoniscus nearcticus, Amerigoniscus malheurensis, Porcellionides floria, Armadillidium vulgare, Porcellio scaber*
28	223	6	1	33	*Armadillidium vulgare*	*Scleropactes concinnus, Circoniscus ornatus, Porcellio laevis, Cubaris murina, Armadillidium vulgare*

Table 5.4. List of terrestrial isopod species that were recorded in more than five bioregions.

Species	Number of bioregions occupied	List of occupied bioregions
Armadillidium vulgare	25	1, 2, 3, 4, 5, 6, 7, 8, 9, 10, 11, 12, 13, 14, 15, 16, 18, 20, 21, 23, 24, 25, 26, 27, 28
Porcellionides pruinosus	25	1, 2, 3, 4, 5, 6, 7, 8, 9, 10, 11, 12, 13, 14, 15, 16, 17, 18, 19, 20, 21, 23, 24, 25, 26
Porcellio laevis	23	1, 2, 3, 4, 5, 6, 7, 8, 9, 10, 11, 12, 13, 14, 16, 17, 18, 20, 21, 23, 24, 26, 28
Porcellio scaber	22	1, 2, 3, 4, 5, 7, 8, 9, 10, 11, 12, 13, 15, 16, 18, 20, 21, 24, 25, 26, 27, 28
Armadillidium nasatum	15	1, 2, 3, 5, 6, 9, 10, 11, 13, 14, 16, 18, 20, 25, 27
Porcellio dilatatus	14	1, 2, 3, 4, 5, 6, 7, 10, 11, 12, 18, 20, 23, 25
Agabiformius lentus	14	1, 2, 3, 4, 5, 6, 8, 9, 11, 12, 18, 20, 23, 24
Ligia (Megaligia) exotica	12	1, 6, 7, 10, 12, 16, 18, 20, 21, 24, 25, 26
Porcellionides sexfasciatus	12	1, 2, 4, 5, 7, 8, 9, 11, 12, 15, 18, 21
Oniscus asellus	11	1, 2, 3, 4, 5, 8, 9, 10, 12, 20, 27
Haplophthalmus danicus	11	1, 2, 3, 4, 5, 10, 11, 12, 15, 16, 25
Armadilloniscus ellipticus	11	1, 2, 3, 6, 8, 11, 12, 13, 14, 16, 25
Ligia italica	11	2, 3, 4, 5, 8, 9, 11, 12, 13, 14, 23
Halophiloscia couchii	10	1, 2, 3, 4, 5, 8, 11, 12, 13, 14
Platyarthrus schoblii	10	1, 2, 3, 4, 5, 8, 11, 12, 14, 22
Armadillo officinalis	10	1, 2, 3, 4, 5, 8, 11, 13, 14, 23
Porcellio lamellatus	9	1, 2, 3, 4, 5, 11, 12, 13, 14
Philoscia muscorum	9	1, 2, 3, 5, 8, 9, 10, 11, 13
Platyarthrus hoffmannseggii	9	1, 2, 3, 4, 5, 9, 11, 13, 14
Armadillidium granulatum	9	1, 2, 4, 5, 8, 9, 11, 13, 23
Cylisticus convexus	8	1, 2, 3, 5, 6, 9, 10, 18
Trichoniscus pusillus	8	1, 2, 3, 4, 5, 9 ,10, 12
Chaetophiloscia elongata	8	1, 2, 3, 5, 8, 9, 11, 13
Trichoniscus pygmaeus	8	1, 2, 3, 4, 5, 9, 11, 12
Chaetophiloscia cellaria	8	1, 2, 3, 5, 8, 9, 11, 14
Armadillidium arcangelii	8	1, 2, 4, 5, 8, 9, 11, 13
Cubaris murina	7	1, 6, 18, 20, 24, 26, 28

Table 5.4. Continued

Species	Number of bioregions occupied	List of occupied bioregions
Ligia oceanica	7	1, 2, 3, 4, 5, 10, 21
Chaetophiloscia sicula	7	1, 2, 3, 5, 9, 11, 13
Androniscus dentiger	7	1, 2, 3, 4, 5, 9, 12
Eluma caelata	7	1, 2, 4, 7, 10, 12, 15
Armadillidium depressum	7	1, 2, 5, 9, 11, 13, 23
Philoscia affinis	7	1, 2, 3, 5, 9, 11, 13
Trichoniscus provisorius	7	1, 2, 3, 5, 9, 11, 12
Leptotrichus panzerii	7	2, 4, 6, 9, 11, 12, 23
Armadillidium assimile	7	2, 4, 5, 9, 11, 12, 23
Porcellio obsoletus	7	3, 8, 9, 11, 13, 14, 23
Trachelipus rathkii	6	1, 2, 3, 6, 9, 13
Hyloniscus riparius	6	1, 2, 3, 9, 10, 14
Tylos europaeus	6	1, 2, 4, 5, 11, 12
Acaeroplastes melanurus	6	1, 2, 4, 5, 11, 12
Stenophiloscia glarearum	6	1, 2, 8, 11, 13, 23
Stenoniscus pleonalis	6	1, 2, 3, 8, 9, 11
Platyarthrus aiasensis	6	1, 2, 5, 10, 11, 20
Porcellionides myrmecophilus	6	2, 8, 9, 11, 13, 23

5.3.3. Freshwater biomes

A bit more than half (~56%) of the world's freshwater isopods are included in the analysed dataset. Coverage for families and genera is higher, with approximately 82% and 70%, respectively. The clustering algorithm yielded 23 bioregions based on the species compositions of freshwater isopods (Fig. 5.5). It recognised two extensive bioregions in North America (Bioregion 1) and Europe (Bioregion 2), respectively. All other bioregions are of a small spatial extent, many of which consist only of a single grid cell. North and Central America, southern Europe, and Australia all have relatively sufficient data coverage and are divided into several distinct bioregions, often in close proximity to each other (see Fig. 5.5). Similar to the terrestrial dataset, the freshwater one shows significant data gaps of occurrence records for freshwater Isopoda on the African continent, Madagascar, Indonesia, and almost all of Asia and South America. The central to western United States also lack data, as do all regions north of the US. The most species-rich bioregion with 91 recorded freshwater isopods is Bioregion 1 in North America, which is mainly defined by *Caecidotea* species (see Table 5.5). Regarding species richness, it is followed by Bioregion 4 (Tasmania), Bioregion 3 (north-eastern Iberian Peninsula), and Bioregion 2 in Europe. Although Bioregion 21 in central southern Australia contains 455 records (much more than Bioregions 3 and 4), only a single species is recorded there, *Phreatomerus latipes* (Chilton, 1922). Endemicity ranges from 62% in Bioregion 2 to 100% in Bioregions 11 (New Zealand), 15 (western Australia), 17 (northern tip of Sumatra), 21 (central southern Australia), 22 (in eastern Australia), and 23 (Yucatan Peninsula). Freshwater species had the most restricted biogeographic distributions, with only 19 species recorded in two or more bioregions (Table 5.6). The most widespread species in the analysis was *Proasellus coxalis* (Dollfus, 1892) (6 occupied bioregions), followed by *Asellus (Asellus) aquaticus* (Linnaeus, 1758) (5 bioregions). Of both, several regional subspecies are recognised.

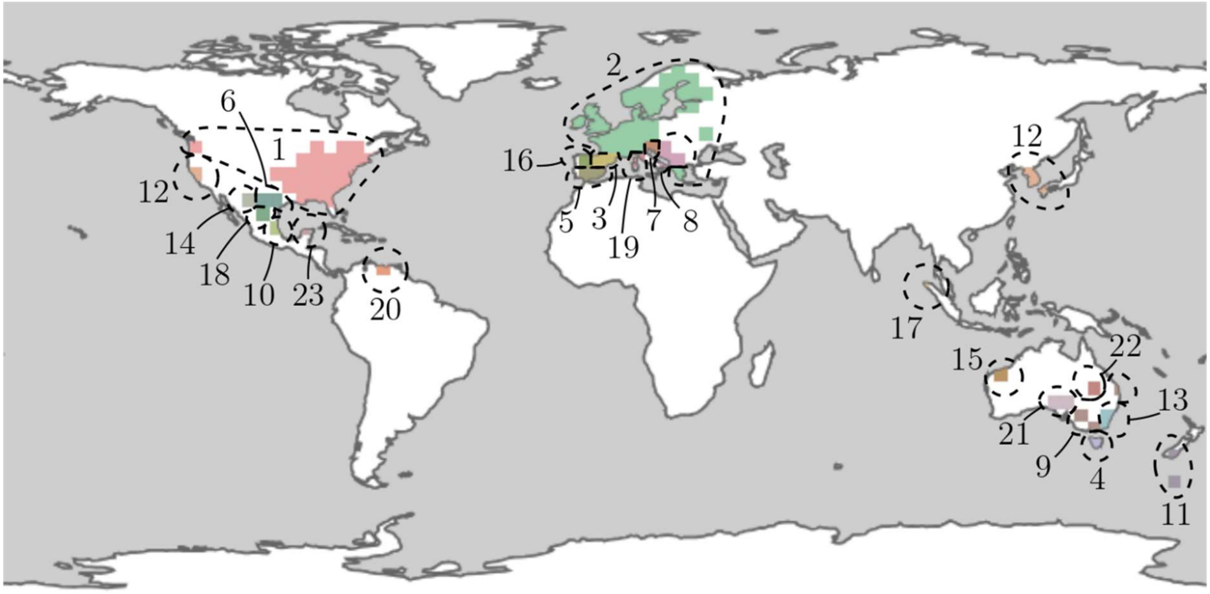

Figure 5.5. Bioregions of freshwater Isopoda. The coloured blocks are all those grid cells that had sufficient data points to be included in the analysis. Blocks of the same colour were clustered together into a bioregion by the Infomap algorithm. The dashed lines are only meant to serve enhanced clarity and do not represent actual bioregion boundaries. For more information on each bioregion, see Table 5.5.

Table 5.5. Bioregions of freshwater Isopoda. For each region, the most common species is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	1,930	91	25	93	*Caecidotea communis*	*Caecidotea kenki, Lirceus fontinalis, Caecidotea recurvata, Caecidotea richardsonae, Caecidotea bicrenata*
2	36,983	26	39	62	*Asellus (Asellus) aquaticus*	*Proasellus hermallensis, Proasellus valdensis, Proasellus synaselloides, Gallasellus heilyi, Proasellus franciscoloi*
3	150	29	2	72	*Stenasellus virei*	*Proasellus lescherae, Stenasellus buili, Proasellus cantabricus, Proasellus aquaecalidae, Proasellus ebrensis*
4	112	30	1	93	*Onchotelson brevicaudatus*	*Onchotelson brevicaudatus, Mesacanthotelson setosus, Mesacanthotelson tasmaniae, Colubotelson chiltoni, Uramphisopus pearsoni*
5	26	15	2	80	*Proasellus coxalis*	*Proasellus escolai, Proasellus beticus, Proasellus granadensis, Proasellus comasi, Proasellus lagari*
6	58	13	2	77	*Cirolanides texensis*	*Caecidotea bilineata, Lirceolus pilus, Lirceolus bisetus, Lirceolus cocytus, Cirolanides texensis*
7	81	15	1	67	*Asellus (Asellus) aquaticus*	*Proasellus intermedius, Proasellus istrianus, Proasellus deminutus, Proasellus parvulus, Proasellus slovenicus*
8	70	10	3	80	*Proasellus anophtalmus*	*Proasellus remyi, Proasellus gjorgjevici, Monolistra (Monolistra) monstruosa, Proasellus anophtalmus, Proasellus karamani*

Table 5.5. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
9	40	8	3	75	*Tachaea caridophaga*	*Phreatoicoides gracilis, Crenoicus shephardi, Colubotelson searlei, Heterias pusilla, Tachaea caridophaga*
10	29	11	1	91	*Speocirolana pelaezi*	*Mexilana saluposi, Speocirolana pubens, Caecidotea chicoensis, Speocirolana prima, Speocirolana xilitla*
11	37	9	2	100	*Austridotea lacustris*	*Austridotea lacustris, Notamphisopus benhami, Notamphisopus littoralis, Austridotea benhami, Notamphisopus dunedinensis*
12	119	7	5	71	*Asellus (Asellus) hilgendorfii*	*Caecianiropsis psammophila, Calasellus californicus, Caecidotea tomalensis, Asellus (Asellus) hilgendorfii, Gnorimosphaeroma insulare*
13	42	6	2	83	*Crenoicus buntiae*	*Crenoicus buntiae, Metaphreatoicus lacustris, Metaphreatoicus australis, Crenoicus harrisoni, Ptyosphaera alata*
14	14	6	1	83	*Thermosphaeroma macrura*	*Thermosphaeroma macrura, Thermosphaeroma smithi, Lirceolus nidulus, Thermosphaeroma mendozai, Thermosphaeroma milleri*
15	27	6	1	100	*Pygolabis humphreysi*	*Kagalana tonde, Pygolabis humphreysi, Pygolabis paraburdoo, Pygolabis eberhardi, Pygolabis weeliwolli*
16	17	7	1	71	*Synasellus bragaianus*	*Proasellus stocki, Proasellus oviedensis, Bragasellus escolai, Bragasellus oscari, Synasellus bragaianus*

Table 5.5. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
17	16	6	1	100	*Probopyrus abhoyai*	*Probopyrus brachysoma, Probopyrus bengalensis, Probopyrus alcocki, Probopyrus gangeticus, Probopyrus abhoyai*
18	17	6	1	83	*Sphaerolana interstitialis*	*Mexistenasellus nulemex, Speocirolana thermydronis, Sphaerolana affinis, Sphaerolana interstitialis, Sphaerolana karenae*
19	13	5	1	80	*Proasellus beroni*	*Proasellus acutianus, Proasellus faesulanus, Proasellus beroni, Proasellus ruffoi, Proasellus coxalis*
20	10	4	1	75	*Calabozoa pellucida*	*Calabozoa pellucida, Afrocerberus letabai, Protocerberus schminkei, Probopyrus floridensis*
21	455	1	2	100	*Phreatomerus latipes*	*Phreatomerus latipes*
22	16	2	1	100	*Ponderella bundoona*	*Ponderella ecomanufactia, Ponderella bundoona*
23	21	2	1	100	*Creaseriella anops*	*Yucatalana robustispina, Creaseriella anops*

Table 5.6. List of freshwater isopod species that were recorded in more than one bioregion.

Species	Number of bioregions occupied	List of occupied bioregions
Proasellus coxalis	6	2, 3, 5, 7, 8, 19
Asellus (Asellus) aquaticus	5	1, 2, 3, 7, 8
Stenasellus virei	4	2, 3, 5, 16
Ptyosphaera alata	3	4, 9, 13
Caecidotea communis	3	1, 2, 12
Proasellus meridianus	3	2, 3, 16
Proasellus cavaticus	3	2, 3, 7
Heterias pusilla	2	4, 9
Probopyrus bithynis	2	1, 10
Thermosphaeroma subequalum	2	6, 14
Mexistenasellus coahuila	2	6, 18
Caecidotea intermedia	2	1, 6
Probopyrus floridensis	2	1, 20
Caecidotea racovitzai	2	1, 12
Proasellus margalefi	2	3, 5
Proasellus walteri	2	2, 3
Proasellus boui	2	2, 3
Proasellus strouhali	2	2, 7
Proasellus slavus	2	2, 7

5.4. Discussion

5.4.1. The marine realm

The clustering algorithm discriminated 33 distinct marine bioregions for isopods, a few of which are spatially extensive, but most are quite limited in extent. A similar analysis of the distribution of 65,000 marine animal and plant species (including both benthic and pelagic species) was performed by Costello et al. (2017). Their multi-taxa approach revealed 30 marine biogeographic realms with distinct species compositions. Given the benthic lifestyle of isopods without a pelagic larval phase and their subsequent low dispersal ability, it is no surprise that the current analysis found many small, much more "localised" bioregions that are nested within the broader realms delineated by Costello et al. (2017). Nevertheless, there are some similarities between the two bioregionalisations. Both found an extensive Antarctic bioregion, a bioregion that spans the southern tip of the African continent and which is distinct from the seas around Madagascar, a bioregion along the western North American coastline, and one that stretches from the Gulf of Mexico into the Caribbean Sea and along the eastern North American coastline. Both approaches also divided the waters around Australia into a northern and a southern bioregion. However, the most extensive bioregion proposed by the current analysis (Bioregion 1, see Fig. 5.1) encompasses several of Costello et al.'s (2017) high and mid-latitude northern hemisphere realms. Bioregion 1 spans the entire Arctic Ocean, which they divided into three separate regions. It also includes the Baltic Sea, the North Sea, the northern North Atlantic Ocean, the Mediterranean Sea, and the Black Sea, all of which Costello et al. (2017) delineated as distinct biogeographic regions. When the clustering algorithm of "Infomap Bioregions" is set to put weight on abundance in order to highlight patterns of abundant isopod species, the structuring of European seas resembles that of Costello et al. (2017) a bit more closely. In that case, the Arctic Ocean is distinct from adjacent seas. The North Sea and the Mediterranean Sea still cluster together, but are now distinct from both the Baltic and the Black Sea, which form two separate bioregions. For most of the other delineated marine bioregions putting weight on abundance does not lead to significant changes. However, Bioregion 2 is divided into two separate regions. A northern region that encompasses the eastern North American coastline, the Gulf of Mexico, and the Caribbean Sea, and a southern region in which the grid cells along the South American coast cluster together. Lowering the cluster cost of the algorithm to e.g., 1.0 (to get a higher number of clusters) does not significantly change the extent of most delineated bioregions, but rather highlights sampling bias within the dataset by singling out grid cells in which more rare species had been sampled than in adjacent grid cells. Therefore, it does not improve the bioregionalisation or closer depict reality.

There are certainly regional variations in species compositions within some of the more extensive bioregions for isopods (e.g., Castelló et al., 2020; Zimina et al., 2019). However, for the algorithm to pick up localised sub-regions, one would have to perform a regional analysis of a subset of the data at a finer scale (e.g., 1° grid cell size or smaller). In such a case, Bioregion 6, for example, which stretches along the entire North American Westcoast, would be subdivided into four sub-regions. However, such differences were not detected in the current global analysis, which was performed at a coarse resolution of 4° grid cells to balance out spatial differences in data density. Additionally, in the case of the immensely extensive Bioregion 1, common, wide-ranging species, may have led to some extent of homogenisation within species compositions of its various grid cells. Within marine isopods, there are several wide-ranging species, especially ones associated with rafting in or on detached vegetation as a means of dispersal. This includes wood-boring species and herbivorous isopods that live on macroalgae. One such species, *Idotea metallica* Bosc, 1801, is even a cosmopolitan species adapted to a rafting lifestyle (Brusca, 1984; Gutow et al., 2006) and was recorded in 14 of the 33 proposed bioregions. Another idoteid, *Idotea balthica* (Pallas, 1772), is a common intertidal grazer on both sides of the Atlantic Ocean in Europe and North America with still ongoing trans-Atlantic colonisation besides having historically isolated populations (Wares, 2001). Borges et al. (2014) reported the distribution of a wood-boring limnoriid species from Arctic waters into temperate regions and other limnoriids ranging from the temperate Atlantic into the Mediterranean. In fact, many Mediterranean species have an Atlantic origin with distribution ranges spanning both seas (Bakalem et al., 2020; Cartes & Figueroa, 2020). On the other hand, many Arctic regions undergo an “Atlantification” with temperate species shifting or expanding their distribution ranges northwards with a warming climate (Borges et al., 2014; Zimina et al., 2019). Also, for some isopod species, potential biogeographic barriers like the Greenland-Scotland Ridge do not restrict the faunal exchange between the Arctic and North Atlantic Oceans (Schnurr et al., 2014). All these examples taken together can explain the enormous extent of Bioregion 1 and how all these seas might be connected.

Another analysis of biogeographic patterns of shallow-water benthic organisms found that modern bioregions are very similar to ones from the late Cenozoic reconstructed from fossil data and, therefore, have been relatively stable for the past 10 million years (Kocsis et al., 2018). Based on their data, Kocsis et al. (2018) proposed an extensive Arctic bioregion similar to the one proposed for isopods, which also stretched southwards into temperate latitudes. However, they also delineated a separate European bioregion that included the Baltic, North, Black, and Mediterranean Seas, as well as the northeastern Atlantic Ocean. Similar to what the analysis of marine isopod occurrences proposes, they delineated an Antarctic bioregion that encompasses the coastlines of southern South America. In accordance with this, Brandt et al. (2016) mentioned that the isopod composition of the Southern Ocean shows most biogeographic links to the fauna of the South Atlantic. According to Kocsis et al. (2018), deep ocean basins and the joint structure of landmass distribution primarily defined the boundaries of benthic coastal bioregions. However, the best secondary predictor of modern bioregion distributions was seawater temperature. After testing the robustness of their biogeographical partitioning, they concluded that meaningful bioregions can be outlined even in relatively poor sampling conditions and without environmental information (Kocsis et al., 2018).

Recently, many analyses of global marine biogeographical patterns within specific taxa have been performed, for example, for coastal cephalopods (Rosa et al., 2019), benthic amphipods (Arfianti & Costello, 2020), mangrove crabs (Sharifian et al., 2020), polychaete worms (Pamungkas et al., 2021), and brittle stars (Victorero et al., 2023). Even though there are apparent taxon-specific differences in the extent and location of bioregion boundaries, there are overlapping similarities. Where appropriate for the taxon, all the abovementioned studies found circumglobal Arctic and Antarctic bioregions, as in the current analysis of marine isopods. Other similarities include distinct New Zealand and Australian biota (Arfianti & Costello, 2020; Pamungkas et al., 2021; Victorero et al., 2023), the division of Australia into a northern and a southern bioregion (Rosa et al., 2019; Sharifian et al., 2020; Victorero et al., 2023), and a bioregion encompassing the tropical to temperate western Atlantic Ocean (Arfianti & Costello, 2020; Rosa et al., 2019; Sharifian et al., 2020; Victorero et al., 2023). While some of the single-taxa studies grouped all or most of the European seas into a combined bioregion with parts of the northern North Atlantic Ocean, all of them delineated that bioregion as distinct from an Arctic bioregion, contrary to what was found for isopods.

While some of the single-cell bioregions delineated herein likely represent biogeographic provinces (i.e. remote archipelagos, etc.) with distinct species communities that will still be identified as such in future analyses when more data will be available, other grid cells, which show up as a separate bioregion in this analysis, might integrate into more extensive bioregions once more occurrence records become available. Some grid cell clusters likely represent specific sampling events rather than actual distinct species communities. In the case of marine Bioregion 10, for example, most of the occurrence records are part of two deep sea sampling series from the Smithsonian Institution's National Museum of Natural History and Senckenberg's Census of Abyssal Marine Life. Therefore, this cell cluster includes mostly deep sea species, which is why the current analysis classifies it as different from adjacent grid cells, whose species communities are complemented by shallow-water species. The same seems to be the case for Bioregion 29, which includes mostly occurrence records of deep sea species that were collected during two sampling series from the Senckenberg Institution.

Many studies that have examined species composition over a broad depth range reported significant changes in species assemblages along the depth gradient (Brandt, De Broyer, et al., 2007; Brandt et al., 2016; Schnurr et al., 2014; Zimina et al., 2019). Depth adds complexity to the marine environment, and species richness tends to decrease with depth. However, several studies of isopods reported a peak in species richness in the deep sea, especially for asellote isopods (e.g., Brandt et al., 2016; Saeedi et al., 2022; G. D. F. Wilson, 1998). In the current analysis, the marine dataset was, in addition to being analysed as a whole, divided into three depth categories to examine if and how depth affects bioregionalisation. Many isopod species are eurybathic, which is reflected in the analysed dataset as there is a lot of overlap between depth categories (see Fig. 5.2). Only very few species were recorded exclusively at intermediate depths of 200 - 500 m. Generally, only a small proportion of isopod species were recorded at intermediate depths, and only a few grid cells had sufficient records to be included in the analysis. The resulting bioregions are consistent with what was found for the complete dataset, with an Arctic bioregion, distinct species assemblages for the Atlantic and Pacific coasts of North America, and distinct Australian and New Zealand bioregions. However, the Antarctic bioregion is split into three species clusters (see Fig. 5.3b). They are all far apart; two consist only of a single grid cell. It is likely that these clusters highlight local environmental conditions that influence species composition or that simply the scarcity of available records within the diversity databases or the employed sampling methods (e.g., epibenthic sled vs. box corer) are responsible for the observed pattern. Only further sampling can show whether these clusters extend to a broader range and would merit the division of the Antarctic bioregion into subregions. In the shallow-water dataset, which is richer in data points, the same grid cells group into a single Antarctic bioregion. Most studies that examined species composition within the Southern Ocean were carried out in the Weddell Sea and adjacent Atlantic sector, and none compared their findings to other Southern Ocean regions (Brandt, Brix, et al., 2007; Brandt, De Broyer, et al., 2007; Brandt et al., 2016; Di Franco et al., 2020). Nevertheless, published literature usually treats the Southern Ocean as a single, relatively isolated region. Its biogeographic isolation is promoted by the Antarctic Circumpolar Current (Barker et al., 2007; Crame, 1999), which is assumed to aid in the passive dispersal (via rafting) of species across biogeographic barriers within the Antarctic bioregion (Leese et al., 2010).

Most occurrence records are from coastal, shallow waters and produce a similar bioregionalisation overall to the one resulting from the complete dataset. The deep-sea dataset, however, splits two of the extensive northern hemisphere bioregions into separate clusters and shows a heterogeneous species composition in the seas surrounding New Zealand (see Fig. 5.3c). This contradicts the long-held belief that the deep sea is a very homogeneous environment with only few but widespread species. Isopods are among the taxa that show high species richness in the deep sea, with Asellota as the dominant suborder (Hessler et al., 1979; Kussakin, 1973). Hessler and Sanders (1967) observed that their diversity is much higher than previously assumed. Recently, widely available abyssal rock patches were revealed, significantly increasing the knowledge about habitat heterogeneity in the deep sea (Riehl et al., 2020). In a study examining the distribution ranges of deep-sea peracarids, including isopods, Brandt et al. (2012) concluded that only very few, if any, peracarid species are truly widespread. They suggested that many species assumed to be wide-ranging may comprise cryptic species complexes, as has been demonstrated in molecular studies of several isopod species (Held, 2003; Hurtado et al., 2016; Raupach et al., 2007).

5.4.2. The terrestrial realm

Within terrestrial isopods, much more wide-ranging species (both in number and range size) were found than in aquatic isopods. Several habitat generalists are cosmopolitan or circumglobal, having spread over all continents or at least the northern hemisphere, often through anthropogenic dispersal vectors. Cosmopolitan species like *Armadillidium vulgare* (Latreille, 1804) and *Porcellionides pruinosus* (Brandt, 1833) have been reported to have high tolerance limits to, e.g., habitat disturbance and desiccation. They thrive not only in pristine and moist natural environments but also in heavily degraded, dry or urban environments (Csonka et al., 2018; Hornung et al., 2008; Vilisics et al., 2007). In many regions, high numbers of non-native terrestrial isopod species are reported (e.g., Hornung et al., 2008). Therefore, it is not surprising that one very extensive bioregion (Bioregion 1, Fig. 5.4) was detected by the analysis covering big parts of North America and Europe and a few stray grid cells, which contain highly populated areas. Operating at low spatial resolution for the global-scale analysis, the clustering algorithm probably did not pick up differences in species assemblages at finer scales that the broad distributions of cosmopolitan species might have overshadowed.

The most detailed biogeographic framework of the terrestrial realm in the scientific literature to date was done by Olson et al. (2001). They produced a global map of 867 distinct ecoregions nested within 14 biomes and eight biogeographic realms. Olson et al.'s (2001) bioregionalisation is far too detailed to be comparable with the herein-produced global bioregionalisation of terrestrial isopods. Besides, the characterisation of ecoregions in Olsen et al. (2001) relied mainly on landforms and vegetation type, which seem of low importance when looking at global patterns of terrestrial isopod diversity. Only a few island ecoregions were delineated similarly by both approaches. These are the Azores archipelago (Bioregion 12, see Fig. 5.4), Socotra Island (Bioregion 22), and Lord Howe Island (Bioregion 17). The islands of Corsica and Sardinia (Bioregion 11) and Sicily (Bioregion 23) were placed into the same ecoregion by Olson et al. (2001); however, they harbour distinct isopod communities. The other isopod bioregions encompass all two or more ecoregions, i.e. different forest or grassland types.

Ficetola et al. (2017) examined the global drivers of terrestrial bioregionalisation. They concluded that the interplay of multiple drivers has shaped the distribution of biogeographic boundaries. Tectonic movements have led to deeply divergent biogeographical realms, while sharp changes in climate and dispersal barriers like mountain ranges determine biogeographical boundaries within those realms. The distribution of isopod species on local scales reflects a species' tolerance limits and the availability of suitable hiding places, i.e. environmental heterogeneity (Csonka et al., 2018; Sfenthourakis & Hornung, 2018). For example, desiccation resistance in terrestrial isopods is associated with morphological traits like cuticle thickness. A thicker cuticle minimises water loss and enables an individual to survive in drier conditions than individuals from species with thinner cuticles can withstand (Csonka et al., 2018). For example, Csonka et al. (2018) found that the globally occurring habitat generalist *Armadillidium vulgare* has a relatively thick cuticle that offers effective protection. Temperature is another factor that is an explanatory variable for isopod distributions on land. Within the area of the former USSR, species diversity decreased northwards, with the northernmost occurrence records in the southern taiga (Kuznetsova & Gongalsky, 2012). That study concluded that the mean annual temperature was the limiting factor. No isopods were found north of the isocline of 120 days a year with a temperature of more than 10°C (Kuznetsova & Gongalsky, 2012).

Occurrence records for only approximately 27% of globally described terrestrial isopod species have been available for this analysis. This means that the underlying dataset for the produced bioregionalisation is highly incomplete. The clearest structuring of bioregions was produced for southern Europe (see Fig. 5.4). That is also where the highest species richness per grid cell was found. Another reliable result is the distinct bioregions of Australia and New Zealand. The somewhat chaotic structuring of North and Central America with several disjunct bioregions will improve when more data become available. Looking at Figure 5.4, there are clearly still enormous geographical gaps in our knowledge of terrestrial isopod diversity and distribution. Except for the Cape Town region, Africa is entirely blank, so are Madagascar, the Arabian Peninsula, Indonesia, and most of Asia. Most regions in South and Central America lack sufficient occurrence records, as does most of the Australian continent. Many of the blank areas on the map have many species described and occurrences recorded in the scientific literature. However, those data were not uploaded to the biodiversity databases, which built the basis of the current analysis. Integration of these "missing" data will highly improve large-scale analyses like the one presented here. Also, integrating phylogenetic data where possible will provide valuable insights into historical regional relationships (Holt et al., 2013).

5.4.3. Freshwater biomes

Early ancestors of extant isopods had an incursion into freshwater environments in the late Devonian, more than 360 mya (Robin et al., 2021). Fossils of phreatoicidean isopods provide evidence that this group of freshwater Isopoda was widespread on Gondwana by the Jurassic period, and vicariant events during the fragmentation of the supercontinent can explain the suborder's modern distribution patterns (G. D. F. Wilson, 2008b; G. D. F. Wilson & Edgecombe, 2003). Similarly, continental drift is the most probable explanation for the geographic distribution of freshwater microcerberids on both sides of the Atlantic Ocean, placing their origin in the Cretaceous period prior to the formation of the Atlantic (Wägele et al., 1995). Many freshwater isopods are stygobionts, living in caves, various groundwater ecosystems, or the interstitial. Aquatic hypogean environments were colonised multiple times by members of nearly all suborders (Wägele, 1990). Stygobiontic cirolanids, for example, are considered to be derived from ancestors with a widespread Tethyan distribution that were left stranded in newly developing subterranean habitats by marine transgressions and regressions (Holsinger et al., 1994). Considering the evidence gathered in the studies mentioned above, vicariance is the most crucial factor that has structured global diversity patterns of freshwater Isopoda, with marine dispersal and subsequent incursion of freshwater habitats ruled unlikely in most cases.

This analysis delineated 23 bioregions based on occurrence records of freshwater Isopoda on a global scale. A comprehensive study by Abell et al. (2008) produced a map of 426 freshwater ecoregions derived mainly from freshwater fish data and restricted to surface waters. Data on freshwater isopods are scarce in most parts of the world, which makes this group poorly suited to a global analysis at present. Nevertheless, several of the bioregions found here are similar to ecoregions proposed by Abell et al. (2008). For example, the division of the Iberian Peninsula shows great similarities. The current analysis divided it into three bioregions (see Fig. 5.5). Bioregion 5 covers the south of the peninsula and is equivalent to the Southern Iberia ecoregion (number 413 in Abell et al., 2008) of the freshwater ecoregions of the world (FEOWs). This study lacks the fine-scale resolution of the FEOWs, so Bioregions 16 and 3 are comparable to the Western Iberia ecoregion (no. 412) and the Eastern Iberia ecoregion (no. 414), respectively; however, both also incorporate a part of the Cantabric Coast – Languedoc ecoregion (no. 403) that stretches along the Bay of Biscay coast and covers the south of France. Bioregion 19 in Europe is equivalent to the Italian Peninsula & Islands ecoregion (no. 416), although it lacks records for most of the Italian mainland and Sicily. Abell et al. (2008) proposed a rather extensive ecoregion for central and western Europe (no. 404), which was also found in the current analysis. Here, however, adjacent areas in northern Europe, as well as Greece and a grid cell in Ukraine, are also integrated into Bioregion 2 (Fig. 5.5). The most common species recorded for this bioregion was *Asellus (Asellus) aquaticus* (Linnaeus, 1758), which is the most widespread freshwater isopod in Europe. Several regional subspecies have been described, including one with many cave-adapted populations in karst areas. Other asellids (*Proasellus* spp.) were recently found to increase their distribution ranges from central Europe into northern European countries, probably aided by ship traffic and recreational fishermen (Kemp et al., 2020). Other bioregions (7 & 8) delineated in this study within Europe correspond well to the Upper Danube (no. 417) and Lower Danube (no. 418) (including Dalmatia (no. 419)) ecoregions of Abell et al.'s (2008) FEOWs. The second-most spatially extensive bioregion in the current study (Bioregion 1 in north America, Fig. 5.5) combines a multitude of FEOWs. It might also reflect increased connectivity of freshwater systems through human activities, as in Europe. With the current data, a few smaller bioregions in northern and central America could also be delimited. For example, Bioregion 23 (the northern part of the Yucatán Peninsula), which here is defined by two stygobiontic species collected from caves in the region, has also been proposed as a separate ecoregion (no. 175) in the FEOWs. It is characterised by extensive karst areas with plenty of grottos and cenotes. In the southern hemisphere, only the Australasian region had sufficient data points to be included in the analysis. New Zealand and its Subantarctic islands form a distinct bioregion in both the FEOWs (no. 811) and the current analysis (Bioregion 11). Australia is divided into several bioregions, some corresponding nicely to ecoregions delineated in the FEOWs, while others stretch over two or three ecoregions.

It is evident from the presented data that there are substantial geographical sampling gaps of freshwater isopods in South America, Africa, Asia and Indonesia. Moreover, most of the proposed bioregions in this analysis include less than a hundred occurrence records. More sampling of freshwater habitats is needed to get a better understanding of local, regional and global distributions of freshwater isopods to guide conservation efforts. Many freshwater species have very restricted distribution ranges. Therefore, studies on regional scales might be more informative than global studies, especially in data-scarce regions. Local habitat features and environmental heterogeneity play an important role in structuring species' distributions (Adlem & Timms, 2000; Cortés-Guzmán & Alcocer, 2022). In southwestern Virginian caves and springs, subterranean asellid species richness is high. Due to niche partitioning, it is not uncommon to find two or three asellid species co-occurring at the same site (Lewis et al., 2021). On the other hand, in the Western Carpathians, species richness of individual karst springs was found to be low (Cíbik et al., 2022). However, regional gamma diversity was high, resulting from high taxonomic turnover between springs. A study on benthic freshwater macroinvertebrates in tropical Mexican lakes also highlighted the importance of regional-scale conservation efforts. Each lake contained a unique species community; therefore, diversity was spread across the entire region instead of being concentrated in specific hotspots (Cortés-Guzmán & Alcocer, 2022). A genetic study of groundwater-associated *Haloniscus* species in Australia's central arid zone revealed high regional endemicity of 26 putative species, each restricted to a small geographical range (Guzik et al., 2019). High endemicity rates are also reported in this study, and wide-ranging freshwater species are rare. Only 19 species have been recorded in two or more of the delineated bioregions. Regional freshwater isopod faunas' uniqueness makes the need to close knowledge gaps more pressing.

6. General Discussion

6.1. Summary of the main findings

This thesis explored the global diversity and biogeography of the peracarid crustacean order Isopoda. It did not restrict itself to either aquatic or terrestrial environments but included information on isopods from all environments to analyse the worldwide biodiversity of the whole order.

After a detailed introduction to the natural history of isopods and an overview of what is known about their diversity and biogeography (Chapter 2), a data-driven approach focused on the current status of isopod taxonomy, examining species description rates and the number of people involved in the scientific inventory of isopod species (Chapter 3). Taking advantage of the wealth of taxonomic information stored in the World Register of Marine Species (WoRMS) database, a global list of accepted species names and their authorities has been compiled. This list includes 10,687 extant isopod species in 1,557 genera, 141 families, and 12 suborders. More than half of all named species are marine (6,151), while a considerable number thrive in the terrestrial environment (3,840), and the rest live in freshwater habitats (696). Over the past two and a half centuries, a cohort of 755 first authors has described these isopod species. The number of scientists involved in isopod taxonomy has increased over time, especially since the 1950s. This indicates that increasing effort is put into completing the global isopod inventory and contradicts, at least concerning the number of people involved, the notion that taxonomy is in crisis (Bacher, 2012; Gaston & May, 1992; Hopkins & Freckleton, 2002). Despite this significant effort, the description rate has slowed in recent decades. Given the current pace of the description rate, a statistical model estimates that approximately 660 more isopod species will be described by the end of this century, bringing the total number of named isopod species up to about 11,350. These data highlight the considerable progress that has already been made in the scientific description of the world's isopod species and provide a hopeful outlook for completing a global isopod inventory as an achievable task.

The second data-driven approach utilised the plentiful geo-referenced occurrence records, which are available through biodiversity databases like the Ocean Biodiversity Information System (OBIS) and the Global Biodiversity Information Facility (GBIF). In the first step, these data were used to examine the latitudinal diversity gradient in species richness (Chapter 4). The gradient was determined for the order as a whole and several environmental and ecological subgroups. The results confirmed the claim that bimodality with a dip in species richness within equatorial regions is the most commonly observed pattern of latitudinal diversity (Cerezer et al., 2022; Chaudhary et al., 2016, 2017), in contrast to the long-lasting paradigm that the latitudinal diversity gradient in species richness is generally unimodal with increasing diversity from the poles towards the tropics (Stehli et al., 1969). Within almost all subgroups, except marine isopods (where it was higher in the southern hemisphere), higher species richness was found in the northern hemisphere, leading to asymmetric latitudinal diversity gradients. Although considerable sampling bias towards the extra-tropical northern hemisphere could be detected in the data when accounted for, it did not markedly affect the overall shape of the latitudinal pattern in species richness (also concluded by Boltovskoy & Correa, 2017; Chaudhary et al., 2017; Rivadeneira & Poore, 2020). This was especially evident within marine isopods, where higher species richness was found in the southern hemisphere despite four times more occurrence records in the northern hemisphere. The southern hemisphere peak in marine diversity coincided with the location of some very species-rich biogeographic regions like Australia, New Zealand, and South Africa (which were determined in Chapter 5).

In a second step, the dataset of occurrence records compiled in Chapter 4 was used to identify distinct biogeographic regions within the marine, terrestrial, and freshwater realms (Chapter 5). Cluster analysis identified 33 distinct bioregions for marine isopods, in which endemicity ranged from 27% to 87%. The most widespread species, *Idotea metallica* Bosc, 1801, recorded in 14 of the 33 bioregions, uses rafts of floating vegetation as a dispersal mechanism (Gutow et al., 2006). Within the terrestrial realm, 28 bioregions were recognised. Compared with marine isopods, more of the terrestrial species tended to be widespread. A few terrestrial species are cosmopolitan, occurring in most of the delineated bioregions. Terrestrial endemism rates ranged from 12% to 95%. Within the 23 delineated bioregions in the freshwater realm, regional endemicity ranged from 62% to 100%. This analysis also illustrated the geographical gaps in isopods' distribution data. Vast areas of South America, Africa, Asia, Indonesia, and parts of Australia lack occurrence records of isopod species, as does most of the deep sea floor. Closing these gaps (along with taxonomical gaps) will help refine the outcomes of biogeographical analyses and increase the value of resulting maps as base maps to inform conservation efforts.

6.2. Limitations

The analyses performed in this thesis face the apparent limitations of the available data in the utilised databases. The global species list compiled and analysed in Chapter 3 cannot claim to be complete. Only data made available through WoRMS were used, i.e., only species names entered into the database and verified by a taxonomic editor. In many cases, newly described species are entered into the database with considerable delay, some species have been overlooked, and validating species names and accompanying information takes time. Therefore, there are likely unrecognised synonyms in the dataset and species missing from the list. However, this is not expected to significantly affect the observed rate of description or the other variables examined. Predictions on future species numbers will change, though, once a more complete dataset can be analysed. Already discovered but yet unnamed species will be added to the list in time, as will newly discovered species from future sampling events.

Utilising data from public databases is always accompanied by some pitfalls. For instance, since the temporal range of the observations is very broad both in OBIS and GBIF, and citizen scientists can upload data, too, there is always the risk of misidentifications. To minimise the impact of low-quality data, downloaded datasets should be thoroughly cleaned before analysis, as has been done here. However, even after extensive quality checks, the current dataset of geo-referenced occurrence records is taxonomically and spatially incomplete. Some of the missing data have been previously reported in the scientific literature but have yet to be uploaded to the databases. Given the global and broad taxonomic scope of the current analysis, it was not feasible to fill in gaps by extracting additional occurrence records from the scientific literature. However, for local or limited regional scale and narrow taxonomic scale studies, it would be highly recommended to increase coverage by searching for and adding additional records from the literature.

The bioregionalisations produced in this thesis are a hypothesis, which needs to be tested further and needs to be refined. The delineated bioregions are based only on differences in species compositions of isopods. Furthermore, the resolution used in this analysis is very coarse. In reality, biogeographic regions are not formed by only a single taxon but by a community of various animal and plant species (as well as fungi, protozoans, bacteria, etc.). Additionally, the extent of biogeographic regions and their boundaries underly a complex interplay of environmental factors. Environmental drivers, such as temperature, salinity, productivity, precipitation, etc., and dispersal barriers like ocean trenches or mountain ranges play a pivotal role in defining biogeographic boundaries. More research needs to be conducted to empirically relate those environmental drivers to the observed biogeographic patterns of species distributions within isopods. Nevertheless, the global patterns observed in here match other proposed global bioregionalisations, whether they are based on taxon-specific occurrence records or include a multitude of animal and plant species occurrences, and regardless of whether the results were correlated with environmental variables or not. Several studies agree that even with incomplete datasets and without the inclusion of environmental drivers one can produce meaningful global bioregionalisations. Bioregions resulting from a global analysis with a coarse resolution should better be viewed as biogeographic realms that can be subdivided into smaller biogeographic provinces, which better reflect regional differences. Conducting the global analysis in here on a finer scale is not recommendable as the spatial coverage of the data is highly variable and resulting regions might more strongly reflect sampling bias in the data. If a research question focuses on regional or local differences within one of the delineated bioregions, it is recommended to re-analyse a regional subset of the data with a finer resolution, so smaller provinces can be detected.

6.3. Future directions

This thesis integrated taxonomic and biogeographic knowledge of freshwater, terrestrial, and marine isopods to form a basic understanding of the global diversity and distribution of the order Isopoda. In the process, it also revealed gaps in our knowledge. Despite the ongoing efforts of a considerable workforce describing newly discovered isopod species, a substantial task remains in the scientific documentation of the world's isopods. However, even though the world's biodiversity faces numerous threats, the scientific community is believed to stand a good chance of naming most species before they go extinct (Costello, May, et al., 2013). Only once a species is adequately described and documented will it be included in threat assessments and conservation plans. The achievement of a comprehensive global species inventory entails the thorough examination and description of isopod species previously discovered during sampling events in species-rich locations, presumed to be new to science. Possible species complexes need to be identified and resolved, requiring integrating a morphological approach with molecular methods. Further fieldwork must be conducted in inadequately sampled regions to uncover and add previously unknown diversity to the species list. This is crucial for addressing not only taxonomic gaps but also gaps in isopods' distribution data. Chapter 5 underscores these geographic gaps across diverse environments, emphasising the need for focused research efforts in these regions. Finally, all collected information should be added to digital data repositories to facilitate widespread access to global datasets among scientists.

References

Abell, R., Thieme, M. L., Revenga, C., Bryer, M., Kottelat, M., Bogutskaya, N., Coad, B., Mandrak, N., Balderas, S. C., Bussing, W., Stiassny, M. L. J., Skelton, P., Allen, G. R., Unmack, P., Naseka, A., Ng, R., Sindorf, N., Robertson, J., Armijo, E., … Petry, P. (2008). Freshwater ecoregions of the world: A new map of biogeographic units for freshwater biodiversity conservation. *BioScience*, *58*(5), 403–414. https://doi.org/10.1641/B580507

Achouri, M. S., Hamaied, S., & Charfi-Cheikhrouha, F. (2008). The diversity of terrestrial Isopoda in the Berkoukech area, Kroumirie, Tunisia. *Crustaceana*, *81*(8), 917–929. https://doi.org/10.1163/156854008X354948

Adlard, R. D., & Lester, R. J. G. (1995). The life cycle and biology of *Anilocra pomacentri* (Isopoda: Cymothoidae), an ectoparasitic isopod of the coral reef fish, *Chromis nitida* (Perciformes: Pomacentridae). *Australian Journal of Zoology*, *43*(3), 271–281. https://doi.org/10.1071/ZO9950271

Adlem, L. T., & Timms, B. V. (2000). Biogeography of the freshwater Peracarida (Crustacea) from Barrington Tops, NSW. *Proceedings of the Linnean Society of New South Wales*, *122*, 131–141.

Agnarsson, I., & Kuntner, M. (2007). Taxonomy in a changing world: Seeking solutions for a science in crisis. *Systematic Biology*, *56*(3), 531–539. https://doi.org/10.1080/10635150701424546

Ahadi, N., Sharifi, Z., Hossaini, S. M. T., Rostami, A., & Renella, G. (2020). Remediation of heavy metals and enhancement of fertilizing potential of a sewage sludge by the synergistic interaction of woodlice and earthworms. *Journal of Hazardous Materials*, *385*, Article 121573. https://doi.org/10.1016/j.jhazmat.2019.121573

Ahyong, S. T., Boyko, C. B., Bailly, N., Bernot, J., Bieler, R., Brandão, S. N., Daly, M., De Grave, S., Gofas, S., Hernandez, F., Hughes, L., Neubauer, T. A., Paulay, G., Boydens, B., Decock, W., Dekeyzer, S., Vandepitte, L., Vanhoorne, B., Adlard, R., … Zullini, A. (2022). *World Register of Marine Species*. Available from https://www.marinespecies.org at VLIZ. Accessed 2022-08-24. https://doi.org/10.14284/170

Ahyong, S. T., Boyko, C. B., Bailly, N., Bernot, J., Bieler, R., Brandão, S. N., Daly, M., De Grave, S., Gofas, S., Hernandez, F., Hughes, L. E., Neubauer, T. A., Paulay, G., Boydens, B., Decock, W., Dekeyzer, S., Vandepitte, L., Vanhoorne, B., Adlard, R. D., … Zullini, A. (2023). *World Register of Marine Species*. Available from https://www.marinespecies.org at VLIZ. Accessed 2023-03-29. https://doi.org/10.14284/170

Ali, N. G., El-Sayed Ali, T., Kamel, M. F., Saleh, R., Sherif, A. H., & Aboyadak, I. M. (2022). Eradication of *Livoneca redmanii* infestation in cultured *Argyrosomus regius*. *Aquaculture*, *558*, Article 738373. https://doi.org/10.1016/j.aquaculture.2022.738373

Allen, A. P., & Gillooly, J. F. (2006). Assessing latitudinal gradients in speciation rates and biodiversity at the global scale. *Ecology Letters*, *9*(8), 947–954. https://doi.org/10.1111/j.1461-0248.2006.00946.x

Aneesh, P. T., & Kappalli, S. (2020). Protandrous hermaphroditic reproductive system in the adult phases of *Mothocya renardi* (Bleeker, 1857) (Cymothoidae: Isopoda: Crustacea) - Light and electron microscopy study. *Zoological Studies*, *59*, Article 61. https://doi.org/10.6620/ZS.2020.59-61

Aneesh, P. T., Kottarathil, H. A., & Kumar, A. B. (2022). Simultaneous double parasitism by the parasitic cymothoids (Crustacea: Isopoda) of two genera on a single host fish *Tenualosa toli* from India. *Nauplius*, *30*, Article e2022013. https://doi.org/10.1590/2358-2936e2022013

Appeltans, W., Ahyong, S. T., Anderson, G., Angel, M. V., Artois, T., Bailly, N., Bamber, R. N., Barber, A., Bartsch, I., Berta, A., Błażewicz-Paszkowycz, M., Bock, P., Boxshall, G., Boyko, C. B., Brandão, S. N., Bray, R., Bruce, N. L., Cairns, S., Chan, T. Y., … Costello, M. J. (2012). The magnitude of global marine species diversity. *Current Biology*, *22*(23), 2189–2202. https://doi.org/10.1016/j.cub.2012.09.036

Arfianti, T., & Costello, M. J. (2020). Global biogeography of marine amphipod crustaceans: Latitude, regionalization, and beta diversity. *Marine Ecology Progress Series*, *638*, 83–94. https://doi.org/10.3354/meps13272

Arfianti, T., Wilson, S. P., & Costello, M. J. (2018). Progress in the discovery of amphipod crustaceans. *PeerJ*, *6*, Article e5187. https://doi.org/10.7717/peerj.5187

Artim, J. M., Hook, A., Grippo, R. S., & Sikkel, P. C. (2017). Predation on parasitic gnathiid isopods on coral reefs: A comparison of Caribbean cleaning gobies with non-cleaning microcarnivores. *Coral Reefs*, *36*, 1213–1223. https://doi.org/10.1007/s00338-017-1613-6

Ashton, G. V., Freestone, A. L., Duffy, J. E., Torchin, M. E., Sewall, B. J., Tracy, B., Albano, M., Altieri, A. H., Altvater, L., Bastida-Zavala, R., Bortolus, A., Brante, A., Bravo, V., Brown, N., Buschmann, A. H., Buskey, E., Barrera, R. C., Cheng, B., Collin, R., … Ruiz, G. M. (2022). Predator control of marine communities increases with temperature across 115 degrees of latitude. *Science*, *376*(6598), 1215–1219. https://doi.org/10.1126/science.abc4916

Athanassopoulou, F., Pappas, I. S., & Bitchava, K. (2009). An overview of the treatments for parasitic disease in Mediterranean aquaculture. In C. Rogers & B. Basurco (Eds.), *The use of veterinary drugs and vaccines in Mediterranean aquaculture* (pp. 65–83). CIHEAM, Zaragoza.

Ax, P. (2000). Pancarida - Peracarida. In *Multicellular Animals*. Springer, Berlin, Heidelberg. https://doi.org/10.1007/978-3-662-10396-8_50

Ayari, A., Ghemari, C., & Nasri-Ammar, K. (2021). Reproductive adaption as a survival strategy to life in an arid environment: The terrestrial crustacean *Hemilepistus reaumurii* as a model. *Zoologischer Anzeiger*, *294*, 10–19. https://doi.org/10.1016/j.jcz.2021.07.003

Bacher, S. (2012). Still not enough taxonomists: Reply to Joppa et al. *Trends in Ecology & Evolution*, *27*(2), 65–66. https://doi.org/10.1016/j.tree.2011.11.003

Bakalem, A., Hassam, N., Oulmi, Y., Martinez, M., & Dauvin, J.-C. (2020). Diversity and geographical distribution of soft-bottom macrobenthos in the bay of Bou Ismail (Algeria, Mediterranean Sea). *Regional Studies in Marine Science*, *33*, Article 100938. https://doi.org/10.1016/j.rsma.2019.100938

Bánki, O., Roskov, Y., Vandepitte, L., DeWalt, R. E., Remsen, D., Schalk, P., Orrell, T., Keping, M., Miller, J., Aalbu, R., Adlard, R. D., Adriaenssens, E., Aedo, C., Aescht, E., Akkari, N., Alonso-Zarazaga, M. A., Alvarez, B., Alvarez, F., Anderson, G., … von Konrat, M. (2021). Catalogue of Life Checklist (Annual Checklist 2021). *Catalogue of Life*. https://doi.org/10.48580/d4sb

Barham, E. G., & Pickwell, G. V. (1969). The giant isopod, *Anuropus*: A scyphozoan symbiont. *Deep Sea Research and Oceanographic Abstracts*, *16*, 525–529.

Barker, P. F., Filippelli, G. M., Florindo, F., Martin, E. E., & Scher, H. D. (2007). Onset and role of the Antarctic Circumpolar Current. *Deep Sea Research Part II: Topical Studies in Oceanography*, *54*(21–22), 2388–2398. https://doi.org/10.1016/j.dsr2.2007.07.028

Bebber, D. P., Marriott, F. H. C., Gaston, K. J., Harris, S. A., & Scotland, R. W. (2007). Predicting unknown species numbers using discovery curves. *Proceedings of the Royal Society B: Biological Sciences*, *274*(1618), 1651–1658. https://doi.org/10.1098/rspb.2007.0464

Beck, J. T. (1980). The effects of an isopod castrator, *Probopyrus pandalicola*, on the sex characters of one of its caridean shrimp hosts, *Palaemonetes paludosus*. *The Biological Bulletin*, *158*(1), 1–15.

Belanger, C. L., Jablonski, D., Roy, K., Berke, S. K., Krug, A. Z., & Valentine, J. W. (2012). Global environmental predictors of benthic marine biogeographic structure. *Proceedings of the National Academy of Sciences*, *109*(35), 14046–14051. https://doi.org/10.1073/pnas.1212381109

Boag, T. H., Gearty, W., & Stockey, R. G. (2021). Metabolic tradeoffs control biodiversity gradients through geological time. *Current Biology*, *31*(13), 1–8. https://doi.org/10.1016/j.cub.2021.04.021

Bober, S., Brix, S., Riehl, T., Schwentner, M., & Brandt, A. (2018). Does the Mid-Atlantic Ridge affect the distribution of abyssal benthic crustaceans across the Atlantic Ocean? *Deep Sea Research Part II: Topical Studies in Oceanography*, *148*, 91–104. https://doi.org/10.1016/j.dsr2.2018.02.007

Boltovskoy, D., & Correa, N. (2017). Planktonic equatorial diversity troughs: Fact or artifact? Latitudinal diversity gradients in Radiolaria. *Ecology*, *98*(1), 112–124. https://doi.org/10.1002/ecy.1623

Boos, H., Scalco, A. C. S., & Araujo, P. B. (2021). Biological and ecological traits of *Bathynomus giganteus* and *Bathynomus miyarei* (Crustacea: Isopoda): Contribution to the conservation of deep-sea in southern Brazil. *Aquatic Conservation: Marine and Freshwater Ecosystems*, *31*(8), 2084–2094. https://doi.org/10.1002/aqc.3583

Borges, L. M. S., Merckelbach, L. M., & Cragg, S. M. (2014). Biogeography of wood-boring crustaceans (Isopoda: Limnoriidae) established in European coastal waters. *PloS ONE*, *9*(10), Article e109593. https://doi.org/10.1371/journal.pone.0109593

Bortolini Rosales, J. L., Mejía Estrada, J. A., del Pilar Alonso Reyes, M., Romero-Rodríguez, J., & Baeza, J. A. (2021). Reproductive biology of the bopyrid isopod *Robinione overstreeti*, a branchial parasite of the ghost shrimp *Callichirus islagrande* (Decapoda: Callichiridae) in the Gulf of Mexico. *Marine Biology Research*, *17*(3), 247–259. https://doi.org/10.1080/17451000.2021.1928221

Botosaneanu, L. (2001). Morphological rudimentation and novelties in stygobitic Cirolanidae (Isopoda, Cymothoidea). *Vie et Milieu*, *51*(1–2), 37–54.

Bouchet, P. (2006). The magnitude of marine biodiversity. In C. M. Duarte (Ed.), *The exploration of marine biodiversity: scientific and technological challenges* (pp. 31–62). Fundación BBVA, Bilbao, Spain. http://www.vliz.be/imisdocs/publications/ocrd/114391.pdf

Bouchon, D., Rigaud, T., & Juchault, P. (1998). Evidence for widespread *Wolbachia* infection in isopod crustaceans: Molecular identification and host feminization. *Proceedings of the Royal Society B: Biological Sciences*, *265*(1401), 1081–1090. https://doi.org/10.1098/rspb.1998.0402

Boxshall, G., & Self, D. (2011). *UK Taxonomy & Systematics Review - 2010.*

Boyko, C. B., & Williams, J. D. (2023). Nomenclatural and taxonomic changes in parasitic isopods (Isopoda: Epicaridea), including two new families and note on the questionable association between monogeneans and bopyrids. *Zootaxa*, *5258*(3), 251–269. https://doi.org/10.11646/zootaxa.5258.3.1

Boyko, C. B., & Wolff, C. (2014). Isopoda and Tanaidacea. In J. W. Martin, J. Olesen, & J. T. Høeg (Eds.), *Atlas of Crustacean Larvae* (pp. 210–215). Johns Hopkins University Press.

Brad, T., Iepure, S., & Sarbu, S. M. (2021). The chemoautotrophically based Movile Cave groundwater ecosystem, a hotspot of subterranean biodiversity. *Diversity*, *13*, Article 128. https://doi.org/10.3390/d13030128

Brandt, A., Błażewicz-Paszkowycz, M., Bamber, R. N., Mühlenhardt-Siegel, U., Malyutina, M. V., Kaiser, S., de Broyer, C., & Havermans, C. (2012). Are there widespread peracarid species in the deep sea (Crustacea: Malacostraca)? *Polish Polar Research*, *33*(2), 139–162. https://doi.org/10.2478/v10183-012-0012-5

Brandt, A., Brix, S., Brökeland, W., Choudhury, M., Kaiser, S., & Malyutina, M. V. (2007). Deep-sea isopod biodiversity, abundance, and endemism in the Atlantic sector of the Southern Ocean — Results from the ANDEEP I–III expeditions. *Deep Sea Research Part II: Topical Studies in Oceanography*, *54*(16–17), 1760–1775. https://doi.org/10.1016/J.DSR2.2007.07.015

Brandt, A., Crame, J. A., Polz, H., & Thomson, M. R. A. (1999). Late Jurassic tethyan ancestry of recent southern high-latitude marine isopods (Crustacea, Malacostraca). *Palaeontology*, *42*(4), 663–675. https://doi.org/10.1111/1475-4983.00090

Brandt, A., De Broyer, C., De Mesel, I., Ellingsen, K. E., Gooday, A. J., Hilbig, B., Linse, K., Thomson, M. R. A., & Tyler, P. A. (2007). The biodiversity of the deep Southern Ocean benthos. *Philosophical Transactions of the Royal Society B: Biological Sciences*, *362*(1477), 39–66. https://doi.org/10.1098/rstb.2006.1952

Brandt, A., Linse, K., Ellingsen, K. E., & Somerfield, P. J. (2016). Depth-related gradients in community structure and relatedness of bivalves and isopods in the Southern Ocean. *Progress in Oceanography*, *144*, 25–38. https://doi.org/10.1016/j.pocean.2016.03.003

Brandt, A., & Poore, G. C. B. (2003). Higher classification of the flabelliferan and related Isopoda based on a reappraisal of relationships. *Invertebrate Systematics*, *17*, 893–923. https://doi.org/10.1071/IS02032

Brayard, A., Escarguel, G., & Bucher, H. (2005). Latitudinal gradient of taxonomic richness: Combined outcome of temperature and geographic mid-domains effects? *Journal of Zoological Systematics and Evolutionary Research*, *43*(3), 178–188. https://doi.org/10.1111/j.1439-0469.2005.00311.x

Bribiesca-Contreras, G., Verbruggen, H., Hugall, A. F., & O'Hara, T. D. (2019). Global biogeographic structuring of tropical shallow-water brittle stars. *Journal of Biogeography*, *46*(7), 1287–1299. https://doi.org/10.1111/jbi.13620

Briggs, J. C. (1974). *Marine zoogeography*. McGraw-Hill Book Company.

Brökeland, W., Guðmundsson, G., & Svavarsson, J. (2010). Diet of four species of deep-sea isopods (Crustacea: Malacostraca: Peracarida) in the South Atlantic and the Southern Ocean. *Marine Biology*, *157*, 177–187. https://doi.org/10.1007/s00227-009-1308-9

Broly, P., Deville, P., & Maillet, S. (2013). The origin of terrestrial isopods (Crustacea: Isopoda: Oniscidea). *Evolutionary Ecology*, *27*, 461–476. https://doi.org/10.1007/s10682-012-9625-8

Brook, H. J., Rawlings, T. A., & Davies, R. W. (1994). Protogynous sex change in the intertidal isopod *Gnorimoshpaeroma oregonense* (Crustacea: Isopoda). *The Biological Bulletin*, *187*(1), 99–111. https://doi.org/10.2307/1542169

Browne, J. G., Pitt, K. A., & Norman, M. D. (2017). Temporal patterns of association between the jellyfish *Catostylus mosaicus* and a sphaeromatid isopod and parasitic anemone. *Marine and Freshwater Research*, *68*(9), 1771–1777. https://doi.org/10.1071/MF16076

Bruce, N. L. (1986). Cirolanidae (Crustacea: Isopoda) of Australia. *Records of the Australian Museum, Supplement 6*.

Brusca, R. C. (1983a). A monograph on the isopod family Aegidae in the tropical eastern Pacific. I. The genus *Aega*. *Allan Hancock Monographs in Marine Biology*, *12*, 1–39.

Brusca, R. C. (1983b). Two new idoteid isopods from Baja California and the Gulf of California (Mexico) and an analysis of the evolutionary history the genus *Colidotea* (Crustacea: Isopoda: Idoteidae). *Transactions of the San Diego Society of Natural History*, *20*(4), 69–79.

Brusca, R. C. (1984). Phylogeny, evolution and biogeography of the marine isopod subfamily Idoteinae (Crustacea: Isopoda: Idoteidae). *Transactions of the San Diego Society of Natural History*, *20*(7), 99–134.

Brusca, R. C. (1987). Biogeographic relationships of Galapagos marine isopod crustaceans. *Bulletin of Marine Science*, *41*(2), 268–281.

Brusca, R. C., & Gilligan, M. R. (1983). Tongue replacement in a marine fish (*Lutjanus guttatus*) by a parasitic isopod (Crustacea: Isopoda). *Copeia*, *3*, 813–816.

Brusca, R. C., & Wallerstein, B. R. (1979). Zoogeographic patterns of idoteid isopods in the northeast Pacific, with a review of shallow water zoogeography of the area. *Bulletin of the Biological Society of Washington*, *3*, 67–105.

Brusca, R. C., & Wilson, G. D. F. (1991). A phylogenetic analysis of the Isopoda with some classificatory recommendations. *Memoirs of the Queensland Museum*, *31*, 143–204.

Burbanck, M. P., & Burbanck, W. D. (1974). Sex reversal of female *Cyathura polita* (Stimpson, 1855) (Isopoda, Anthuridae). *Crustaceana*, *26*(1), 110–112.

Burbanck, W. D. (1962). An ecological study of the distribution of the isopod *Cyathura polita* (Stimpson) from brackish waters of Cape Cod, Massachusetts. *The American Midland Naturalist*, *67*(2), 449–476.

Campos-Filho, I. S., Monticelli Cardoso, G., & Aguiar, J. O. (2018). New species and first record of *Alloniscus* Dana, 1854 (Isopoda: Oniscidae: Alloniscidae) from Brazil. *Nauplius*, *26*, Article e2018014. https://doi.org/10.1590/2358-2936e2018014

Carpenter, J. H. (2021). Forty-year natural history study of *Bahalana geracei* Carpenter, 1981, an anchaline cave-dwelling isopod (Crustacea, Isopoda, Cirolanidae) from San Salvador Island, Bahamas: Reproduction, growth, longevity, and population structure. *Subterranean Biology*, *37*, 105–156. https://doi.org/10.3897/subtbiol.37.60653

Cartes, J. E., & Figueroa, D. F. (2020). Deep sea isopods from the western Mediterranean: Distribution and habitat. *Progress in Oceanography*, *188*, Article 102415. https://doi.org/10.1016/j.pocean.2020.102415

Castelló, J., Bitar, G., & Zibrowius, H. (2020). Isopoda (Crustacea) from the Levantine Sea with comments on the biogeography of Mediterranean isopods. *Mediterranean Marine Science*, *21*(2), 308–339. https://doi.org/10.12681/mms.20329

Cerezer, F. O., Machac, A., Rangel, T. F., & Dambros, C. S. (2022). Exceptions to the rule: Relative roles of time, diversification rates and regional energy in shaping the inverse latitudinal diversity gradient. *Global Ecology and Biogeography*, *31*(9), 1794–1809. https://doi.org/10.1111/geb.13559

Cericola, M. J., & Williams, J. D. (2015). Prevalence, reproduction and morphology of the parasitic isopod *Athelges takanoshimensis* Ishii, 1914 (Isopoda: Bopyridae) from Hong Kong hermit crabs. *Marine Biology Research*, *11*(3), 236–252. https://doi.org/10.1080/17451000.2014.928415

Ceriello, H., Lopes, C. S. S., Reimer, J. D., Bakken, T., Fukuda, M. V., Cunha, C. M., & Stampar, S. N. (2020). Knock knock, who's there?: Marine invertebrates in tubes of Ceriantharia (Cnidaria: Anthozoa). *Biodiversity Data Journal*, *8*, Article e47019. https://doi.org/10.3897/BDJ.8.e47019

Chapman, J. W., Dumbauld, B. R., Itani, G., & Markham, J. C. (2012). An introduced Asian parasite threatens northeastern Pacific estuarine ecosystems. *Biological Invasions*, *14*, 1221–1236. https://doi.org/10.1007/s10530-011-0151-3

Chaudhary, C., Richardson, A. J., Schoeman, D. S., & Costello, M. J. (2021). Global warming is causing a more pronounced dip in marine species richness around the equator. *Proceedings of the National Academy of Sciences*, *118*(15), Article e2015094118. https://doi.org/10.1073/pnas.2015094118

Chaudhary, C., Saeedi, H., & Costello, M. J. (2016). Bimodality of latitudinal gradients in marine species richness. *Trends in Ecology & Evolution*, *31*(9), 670–676. https://doi.org/10.1016/j.tree.2016.06.001

Chaudhary, C., Saeedi, H., & Costello, M. J. (2017). Marine species richness is bimodal with latitude: A reply to Fernandez and Marques. *Trends in Ecology & Evolution*, *32*(4), 234–237. https://doi.org/10.1016/j.tree.2017.02.007

Chong, Y. T., Hatai, K., & Ransangan, J. (2015). Life cycle of *Caecognathia coralliophila* (Crustacea, Isopoda, Gnathiidae) in hatchery reared tiger grouper, *Epinephelus fuscogutattus*. *Bulletin of the European Association of Fish Pathologists*, *35*(5), 177–184.

Christenhusz, M. J. M., & Byng, J. W. (2016). The number of known plants species in the world and its annual increase. *Phytotaxa*, *261*(3), 201–217. https://doi.org/10.11646/phytotaxa.261.3.1

Churchfield, S. (1982). Food availability and the diet of the Common shrew, *Sorex araneus*, in Britain. *Journal of Animal Ecology*, *51*, 15–28.

Cíbik, J., Beracko, P., Bulánková, E., Čiamporová Zaťovičová, Z., Gregušová, K., Kodada, J., Krno, I., Mišíková Elexová, E., Navara, T., Rogánska, A., & Derka, T. (2022). Are springs hotspots of benthic invertebrate diversity? Biodiversity and conservation priority of rheocrene springs in the karst landscape. *Aquatic Conservation: Marine and Freshwater Ecosystems*, *32*(5), 843–858. https://doi.org/10.1002/aqc.3802

Clarke, A., & Crame, J. A. (1997). Diversity, latitude and time: Patterns in the shallow sea. In R. F. G. Ormond, J. D. Gage, & M. V. Angel (Eds.), *Marine Biodiversity - Patterns and Processes* (pp. 122–147). Cambridge University Press. https://doi.org/10.1017/CBO9780511752360.007

Cohen, B. F., & Poore, G. C. B. (1994). Phylogeny and biogeography of the Gnathiidae (Crustacea: Isopoda) with descriptions of new genera and species, most from south-eastern Australia. *Memoirs of the Museum of Victoria*, *54*(2), 271–397. https://doi.org/10.24199/j.mmv.1994.54.13

Čolak, S., Kolega, M., Mejdandžić, D., Župan, I., Šarić, T., Piplović, E., & Mustać, B. (2018). Prevalence and effects of the cymothoid isopod (*Ceratothoa oestroides*, Risso 1816) on cultured meagre (*Argyrosomus regius*, Asso 1801) in the eastern Adriatic Sea. *Aquaculture Research*, *49*, 1001–1007. https://doi.org/10.1111/are.13547

Coleman, C. O. (2015). Taxonomy in times of the taxonomic impediment – Examples from the community of experts on amphipod crustaceans. *Journal of Crustacean Biology*, *35*(6), 729–740. https://doi.org/10.1163/1937240X-00002381

Condamine, F. L., Sperling, F. A. H., Wahlberg, N., Rasplus, J.-Y., & Kergoat, G. J. (2012). What causes latitudinal gradients in species diversity? Evolutionary processes and ecological constraints on swallowtail biodiversity. *Ecology Letters*, *15*(3), 267–277. https://doi.org/10.1111/j.1461-0248.2011.01737.x

Corral, J. M., Henmi, Y., Shiozaki, Y., & Itani, G. (2019). Parasitic effects of the bopyrid *Megacepon goetici* (Crustacea: Isopoda) on the varunid crab *Gaetice depressus*. *Diseases of Aquatic Organisms*, *135*(1), 71–75. https://doi.org/10.3354/dao03380

Cortés-Guzmán, D., & Alcocer, J. (2022). Turnover drives high benthic macroinvertebrates' beta diversity in a tropical karstic lake district. *Diversity*, *14*(4), Article 259. https://doi.org/10.3390/d14040259

Costello, M. J. (2015). Biodiversity: The known, unknown, and rates of extinction. *Current Biology*, *25*(9), PR368-R371. https://doi.org/10.1016/J.CUB.2015.03.051

Costello, M. J. (2016). Parasite rates of discovery, global species richness and host specificity. *Integrative and Comparative Biology*, *56*(4), 588–599. https://doi.org/10.1093/icb/icw084

Costello, M. J., Corkrey, R., Bates, A. E., Burrows, M. T., Chaudhary, C., Edgar, G. E., Stuart-Smith, R. D., Yasuhara, M., & Wei, C.-L. (2023). The universal evolutionary and ecological significance of 20 °C. *Frontiers of Biogeography*, *15*(4), Article e61673. https://doi.org/10.21425/F5FBG61673

Costello, M. J., Emblow, C. S., & Picton, B. E. (1996). Long term trends in the discovery of marine species new to science which occur in Britain and Ireland. *Journal of the Marine Biological Association of the United Kingdom*, *76*(1), 255–257. https://doi.org/10.1017/S0025315400029234

Costello, M. J., Houlding, B., & Wilson, S. P. (2014). As in other taxa, relatively fewer beetles are being described by an increasing number of authors: Response to Löbl and Leschen. *Systematic Entomology*, *39*(3), 395–399. https://doi.org/10.1111/syen.12068

Costello, M. J., Lane, M., Wilson, S. P., & Houlding, B. (2015). Factors influencing when species are first named and estimating global species richness. *Global Ecology and Conservation*, *4*, 243–254. https://doi.org/10.1016/j.gecco.2015.07.001

Costello, M. J., May, R. M., & Stork, N. E. (2013). Can we name Earth's species before they go extinct? *Science*, *339*, 413–416. https://doi.org/10.112/science.1230318

Costello, M. J., Tsai, P., Wong, P. S., Cheung, A. K. L., Basher, Z., & Chaudhary, C. (2017). Marine biogeographic realms and species endemicity. *Nature Communications*, *8*, Article 1057. https://doi.org/10.1038/s41467-017-01121-2

Costello, M. J., Vanhoorne, B., & Appeltans, W. (2015). Conservation of biodiversity through taxonomy, data publication, and collaborative infrastructures. *Conservation Biology*, *29*(4), 1094–1099. https://doi.org/10.1111/cobi.12496

Costello, M. J., Wilson, S. P., & Houlding, B. (2012). Predicting total global species richness using rates of species description and estimates of taxonomic effort. *Systematic Biology*, *61*(5), 871–883. https://doi.org/10.1093/sysbio/syr080

Costello, M. J., Wilson, S. P., & Houlding, B. (2013). More taxonomists describing significantly fewer species per unit effort may indicate that most species have been discovered. *Systematic Biology*, *62*(4), 616–624. https://doi.org/10.1093/sysbio/syt024

Crame, J. A. (1999). An evolutionary perspective on marine faunal connections between southernmost South America and Antarctica. *Scientia Marina*, *63*, 1–14.

Crame, J. A. (2000). Evolution of taxonomic diversity gradients in the marine realm: Evidence from the composition of recent bivalve faunas. *Paleobiology*, *26*(2), 188–214.

Crame, J. A. (2020). Early Cenozoic evolution of the latitudinal diversity gradient. *Earth-Science Reviews*, *202*, Article 103090. https://doi.org/10.1016/j.earscirev.2020.103090

Crame, J. A. (2023). Late Cenozoic evolution of the latitudinal diversity gradient. *Journal of Biogeography*, *50*(7), 1213–1220. https://doi.org/10.1111/jbi.14620

Cruz-Motta, J. J., Miloslavich, P., Guerra-Castro, E., Hernández-Agreda, A., Herrera, C., Barros, F., Navarrete, S. A., Sepúlveda, R. D., Glasby, T. M., Bigatti, G., Cardenas-Calle, M., Carneiro, P. B. M., Carranza, A., Flores, A. A. V., Gil-Kodaka, P., Gobin, J., Gutiérrez, J. L., Klein, E., Krull, M., … Romero, L. (2020). Latitudinal patterns of species diversity on South American rocky shores: Local processes lead to contrasting trends in regional and local species diversity. *Journal of Biogeography*, *47*(9), 1966–1979. https://doi.org/10.1111/jbi.13869

Csonka, D., Halasy, K., Buczkó, K., & Hornung, E. (2018). Morphological traits – desiccation resistance – habitat characteristics: A possible key for distribution in woodlice (Isopoda, Oniscidea). *ZooKeys*, *801*, 481–499. https://doi.org/10.3897/zookeys.801.23088

Culp, J. M., Lento, J., Curry, R. A., Luiker, E., & Halliwell, D. (2019). Arctic biodiversity of stream macroinvertebrates declines in response to latitudinal change in the abiotic template. *Freshwater Science*, *38*(3), 465–479. https://doi.org/10.1086/704887

Culver, D. C., Deharveng, L., Bedos, A., Lewis, J. J., Madden, M., Reddell, J. R., Sket, B., Trontelj, P., & White, D. (2006). The mid-latitude biodiversity ridge in terrestrial cave fauna. *Ecography*, *29*(1), 120–128. https://doi.org/10.1111/j.2005.0906-7590.04435.x

Culver, S. J., & Buzas, M. A. (2000). Global latitudinal species diversity gradient in deep-sea benthic foraminifera. *Deep Sea Research Part I: Oceanographic Research Papers*, *47*(2), 259–275. https://doi.org/10.1016/S0967-0637(99)00055-2

Currie, D. J., Francis, A. P., & Kerr, J. T. (1999). Some general propositions about the study of spatial patterns of species richness. *Écoscience*, *6*(3), 392–399. https://doi.org/10.1080/11956860.1999.11682541

Dantas, A., & Fonseca, C. R. (2023). Global biogeographical patterns of ants and their abiotic determinants. *Perspectives in Ecology and Conservation*, *21*(3), 237–246. https://doi.org/10.1016/j.pecon.2023.07.003

Davidson, T. M. (2012). Boring crustaceans damage polystyrene floats under docks polluting marine waters with microplastic. *Marine Pollution Bulletin*, *64*, 1821–1828. https://doi.org/10.1016/j.marpolbul.2012.06.005

De Smedt, P., & Henrard, A. (2022). Observations of *Trachyzelotes pedestris* (C.L. Koch, 1837) hunting for terrestrial isopods in Belgium. *Journal of the Belgian Arachnological Society*, *37*(1), 41–43.

Del Carmen Espinosa-Pérez, M., & Hendrickx, M. E. (2006). A comparative analysis of biodiversity and distribution of shallow-water marine isopods (Crustacea: Isopoda) from polar and temperate waters in the East Pacific. *Belgian Journal of Zoology*, *136*(2), 219–247.

Delaney, P. M. (1989). Phylogeny and biogeography of the marine isopod family Corallanidae (Crustacea, Isopoda, Flabellifera). *Contributions in Science*, *409*, 1–75.

Delaney, P. M., & Brusca, R. C. (1985). Two new species of *Tridentella* Richardson, 1905 (Isopoda: Flabellifera: Tridentellidae) from California, with a rediagnosis and comments on the family, and a key to the genera of Tridentellidae and Corallanidae. *Journal of Crustacean Biology*, *5*(4), 728–742.

Deng, J., Li, K., Chen, C., Wu, S., & Huang, X. (2016). Discovery pattern and species number of scale insects (Hemiptera: Coccoidea). *PeerJ*, *4*, Article e2526. https://doi.org/10.7717/peerj.2526

DeWalt, R. E., & Ower, G. D. (2019). Ecosystem services, global diversity, and rate of stonefly species descriptions (Insecta: Plecoptera). *Insects*, *10*, 1–13. https://doi.org/10.3390/insects10040099

Di Franco, D., Linse, K., Griffiths, H. J., Haas, C., Saeedi, H., & Brandt, A. (2020). Abundance and distributional patterns of benthic peracarid crustaceans from the Atlantic Sector of the Southern Ocean and Weddell Sea. *Frontiers in Marine Science*, *7*, Article 554663. https://doi.org/10.3389/fmars.2020.554663

Dias, N., Sprung, M., & Hassall, M. (2005). The abundance and life histories of terrestrial isopods in a salt marsh of the Ria Formosa lagoon system, southern Portugal. *Marine Biology*, *147*, 1343–1352. https://doi.org/10.1007/s00227-005-0033-2

Dimitriou, A. C., Taiti, S., & Sfenthourakis, S. (2019). Genetic evidence against monophyly of Oniscidea implies a need to revise scenarios for the origin of terrestrial isopods. *Scientific Reports*, *9*, Article 18508. https://doi.org/10.1038/s41598-019-55071-4

Dinerstein, E., Olson, D. M., Joshi, A., Vynne, C., Burgess, N. D., Wikramanayake, E. D., Hahn, N., Palminteri, S., Hedao, P., Noss, R., Hansen, M., Locke, H., Ellis, E. C., Jones, B., Barber, C. V., Hayes, R., Kormos, C., Martin, V., Crist, E., … Saleem, M. (2017). An ecoregion-based approach to protecting half the terrestrial realm. *BioScience*, *67*(6), 534–545. https://doi.org/10.1093/biosci/bix014

Dodds, W. K., Bruckerhoff, L., Batzer, D., Schechner, A., Pennock, C., Renner, E., Tromboni, F., Bigham, K., & Grieger, S. (2019). The freshwater biome gradient framework: Predicting macroscale properties based on latitude, altitude, and precipitation. *Ecosphere*, *10*(7), Article e02786. https://doi.org/10.1002/ecs2.2786

Dodge-Wan, D., & Nagarajan, R. (2020). Boring of intertidal sandstones by isopod *Sphaeroma triste* in NW Borneo (Sarawak, Malaysia). *Journal of Coastal Research*, *36*(2), 238–248. https://doi.org/10.2112/JCOASTRES-D-19-00066.1

Doti, B. L., Chiesa, I. L., & Roccatagliata, D. (2020). Biodiversity of Isopoda and Cumacea (Peracarida, Crustacea) from the Marine Protected Area Namuncurá-Burdwood Bank, South-West Atlantic. *Polar Biology*, *43*(10), 1519–1534. https://doi.org/10.1007/s00300-020-02725-z

Dreyer, H., & Wägele, J. W. (2001). Parasites of crustaceans (Isopoda: Bopyridae) evolved from fish parasites: Molecular and morphological evidence. *Zoology*, *103*, 157–178.

Dreyer, H., & Wägele, J. W. (2002). The Scutocoxifera tax. Nov. and the information content of nuclear ssu rDNA sequences for reconstruction of isopod phylogeny (Peracarida: Isopoda). *Journal of Crustacean Biology*, *22*(2), 217–234.

Durand, S., Braquart-Varnier, C., & Beltran-Bech, S. (2020). Promiscuity and sex ratio in the terrestrial isopod *Armadillidium vulgare* and consequences on genetic diversity. *Behavioural Processes*, *171*, Article 104030. https://doi.org/10.1016/j.beproc.2019.104030

Edler, D., Guedes, T., Zizka, A., Rosvall, M., & Antonelli, A. (2017). Infomap Bioregions: Interactive mapping of biogeographical regions from species distributions. *Systematic Biology*, *66*(2), 197–204. https://doi.org/10.1093/sysbio/syw087

Ejdung, G., & Elmgren, R. (2001). Predation by the benthic isopod *Saduria entomon* on two Baltic Sea deposit-feeders, the amphipod *Monoporeia affinis* and the bivalve *Macoma balthica*. *Journal of Experimental Marine Biology and Ecology*, *266*, 165–179.

Ekman, S. (1953). *Zoogeography of the sea*. Sidgwick and Jackson Limited.

Ellis, P., & Williams, W. D. (1970). The biology of *Haloniscus searlei* Chilton, an oniscoid isopod living in Australian salt lakes. *Australian Journal of Marine and Freshwater Research*, *21*, 51–69.

Ellis, R. J. (1971). Notes on the biology of the isopod *Asellus tomalensis* Harford in an intermittent pond. *Transactions of the American Microscopical Society*, *90*(1), 51–61. https://doi.org/10.2307/3224897

Ellison, A. M., & Farnsworth, E. J. (1990). The ecology of Belizean mangrove-root fouling communities. I. Epibenthic fauna are barriers to isopod attack of red mangrove roots. *Journal of Experimental Marine Biology and Ecology*, *142*, 91–104.

Elsner, N. O., Golovan, O. A., Malyutina, M. V., & Brandt, A. (2013). Alone in the dark: Distribution, population structure and reproductive mode of the dominant isopod *Eurycope spinifrons* Gurjanova, 1933 (Isopoda: Asellota: Munnopsidae) from bathyal and abyssal depths of the Sea of Japan. *Deep Sea Research Part II: Topical Studies in Oceanography*, *86–87*, 103–110. https://doi.org/10.1016/j.dsr2.2012.07.043

Ercoli, F., Lefebvre, F., Delangle, M., Godé, N., Caillon, M., Raimond, R., & Souty-Grosset, C. (2019). Differing trophic niches of three French stygobionts and their implications for conservation of endemic stygofauna. *Aquatic Conservation: Marine and Freshwater Ecosystems*, *29*(12), 2193–2203. https://doi.org/10.1002/aqc.3227

Eschmeyer, W. N., Fricke, R., Fong, J. D., & Polack, D. A. (2010). Marine fish diversity: History of knowledge and discovery (Pisces). *Zootaxa*, *2525*(1), 19–50. https://www.mapress.com/j/zt/article/viewFile/zootaxa.2525.1.2/17829

Essl, F., Rabitsch, W., Dullinger, S., Moser, D., & Milasowszky, N. (2013). How well do we know species richness in a well-known continent? Temporal patterns of endemic and widespread species descriptions in the European fauna. *Global Ecology and Biogeography*, *22*(1), 29–39. https://doi.org/10.1111/J.1466-8238.2012.00787.X

Etter, W. (2014). A well-preserved isopod from the Middle Jurrassic of southern Germany and implications for the isopod fossil record. *Palaeontology*, *57*(5), 931–949. https://doi.org/10.1111/pala.12095

Fenton, I. S., Aze, T., Farnsworth, A., Valdes, P., & Saupe, E. E. (2023). Origination of the modern-style diversity gradient 15 million years ago. *Nature*, *614*(7949), 708–712. https://doi.org/10.1038/s41586-023-05712-6

Fernandez, M. O., & Marques, A. C. (2017). Diversity of diversities: A response to Chaudhary, Saeedi, and Costello. *Trends in Ecology & Evolution*, *32*(4), 232–234. https://doi.org/10.1016/j.tree.2016.10.013

Ficetola, G. F., Mazel, F., & Thuiller, W. (2017). Global determinants of zoogeographical boundaries. *Nature Ecology & Evolution*, *1*, Article 0089. https://doi.org/10.1038/s41559-017-0089

Fischer, L., Covatti Ale, M., Deli Antoni, M., Díaz de Astarloa, J. M., & Delpiani, G. (2022). Feeding ecology of the longtail southern cod, *Patagonotothen ramsayi* (Regan, 1913) (Notothenioidei) in the Marine Proteced Area Namuncurá-Burdwood Bank, Argentina. *Polar Biology*, *45*, 1483–1494. https://doi.org/10.1007/s00300-022-03082-9

Fisher, M. A., Vinson, J. E., Gittleman, J. L., & Drake, J. M. (2018). The description and number of undiscovered mammal species. *Ecology and Evolution*, *8*(7), 3628–3635. https://doi.org/10.1002/ece3.3724

Fogelman, R. M., Kuris, A. M., & Grutter, A. S. (2009). Parasitic castration of a vertebrate: Effect of the cymothoid isopod, *Anilocra apogonae*, on the five-lined cardinalfish, *Cheilodipterus quinquelineatus*. *International Journal for Parasitology*, *39*(5), 577–583. https://doi.org/10.1016/j.ijpara.2008.10.013

Fontaine, B., Perrard, A., & Bouchet, P. (2012). 21 years of shelf life between discovery and description of new species. *Current Biology*, *22*(22), R943–R944. https://doi.org/10.1016/j.cub.2012.10.029

Freestone, A. L., Torchin, M. E., Jurgens, L. J., Bonfim, M., López, D. P., Repetto, M. F., Schlöder, C., Sewall, B. J., & Ruiz, G. M. (2021). Stronger predation intensity and impact on prey communities in the tropics. *Ecology*, *102*(8), Article e03428. https://doi.org/10.1002/ecy.3428

Frutos, I., & Sorbe, J. C. (2010). *Politolana sanchezi* sp. Nov. (Crustacea: Isopoda: Cirolanidae), a new benthic bioturbating scavenger from bathyal soft-bottoms of the southern Bay of Biscay (northeastern Atlantic Ocean). *Zootaxa*, *2640*, 20–34. https://doi.org/10.11646/zootaxa.2640.1.2

Fuller, N., Ford, A. T., Lerebours, A., Gudkov, D. I., Nagorskaya, L. L., & Smith, J. T. (2019). Chronic radiation exposure at Chernobyl shows no effect on genetic diversity in the freshwater crustacean, *Asellus aquaticus* thirty years on. *Ecology and Evolution*, *9*(18), 10135–10144. https://doi.org/10.1002/ece3.5478

Fuller, N., Ford, A. T., Nagorskaya, L. L., Gudkov, D. I., & Smith, J. T. (2018). Reproduction in the freshwater crustacean *Asellus aquaticus* along a gradient of radionuclide contamination at Chernobyl. *Science of the Total Environment*, *628–629*, 11–17. https://doi.org/10.1016/j.scitotenv.2018.01.309

Fuller, N., Smith, J. T., Nagorskaya, L. L., Gudkov, D. I., & Ford, A. T. (2017). Does Chernobyl-derived radiation impact the developmental stability of *Asellus aquaticus* 30 years on? *Science of the Total Environment*, *576*, 242–250. https://doi.org/10.1016/j.scitotenv.2016.10.097

Gaillard, C., Hantzpergue, P., Vannier, J., Margerard, A.-L., & Mazin, J.-M. (2005). Isopod trackways from the Crayssac Lagerstätte, Upper Jurassic, France. *Palaeontology*, *48*(5), 947–962.

Garcia, L. (2020). Description of *Mica iberica* sp. Nov. and *Porcellio cibioi* sp. Nov., two new terrestrial isopods previously confused with *Porcellio ingenuus* Budde-Lund, 1885 (Isopoda: Oniscidea: Porcellionidae). *Bolletí de La Societat d'Història Natural de Les Balears*, *63*, 159–173. https://www.raco.cat/index.php/BolletiSHNBalears/article/download/385135/478236

García-Padrón, L. Y. (2021). Diet of a community of frogs in an agroecosystem in western Cuba. *Caribbean Herpetology*, *76*, 1–8. https://doi.org/10.31611/ch.76

Garzón-Ferreira, J. (1990). An isopod, *Rocinela signata* (Crustacea: Isopoda: Aegidae), that attacks humans. *Bulletin of Marine Science*, *46*(3), 813–815.

Gaston, K. J. (2000). Global patterns in biodiversity. *Nature*, *405*, 220–227. https://doi.org/10.1038/35012228

Gaston, K. J., & May, R. M. (1992). Taxonomy of taxonomists. *Nature*, *356*(6367), 281–282. https://doi.org/10.1038/356281a0

GBIF. (2022). *GBIF Occurrence Download*. Retrieved from www.gbif.org. Accessed 2022-06-10. https://doi.org/10.15468/dl.zqvdpd

Gentile, G., & Argano, R. (2005). Island biogeography of the Mediterranean Sea: The species-area relationship for terrestrial isopods. *Journal of Biogeography*, *32*(10), 1715–1726. https://doi.org/10.1111/j.1365-2699.2005.01329.x

Gentile, G., Argano, R., & Taiti, S. (2022). Evaluating the correlation between area, environmental heterogeneity, and species richness using terrestrial isopods (Oniscidea) from the Pontine Islands (West Mediterranean). *Organisms Diversity & Evolution*, *22*(1), 275–284. https://doi.org/10.1007/s13127-021-00523-x

Girling, M. A. (1979). Calcium carbonate-replaced arthropods from archaeological deposits. *Journal of Archaeological Science*, *6*, 309–320.

Glazier, D. S., & Kleynhans, E. (2015). Arboreal herbivory by a semi-terrestrial South African isopod crustacean, *Tylos capensis* Krauss (Isopoda: Tylidae), on the bietou bush, *Chrysanthemoides monilifera* (L.) Norlindh. *African Invertebrates*, *56*(3), 729–738. https://doi.org/10.5733/afin.056.0315

Glynn, P. W. (1968). Ecological studies on the associations of chitons in Puerto Rico, with special reference to shpaeromid isopods. *Bulletin of Marine Science*, *18*(3), 572–626.

Godfray, H. C. J. (2002). Challenges for taxonomy. *Nature*, *417*, 17–19. https://doi.org/10.1038/417017a

Gopalakrishnan, A., Raja, K., Trilles, J. P., Rajkumar, M., Rahman, M. M., & Saravanakumar, A. (2017). Bopyrid isopods parasitizing on the cultured fresh water prawn, *Macrobrachium malcolmsonii* in South India. *Journal of Parasitic Diseases*, *41*(1), 93–96. https://doi.org/10.1007/s12639-016-0756-7

Grassle, J. F., & Maciolek, N. J. (1992). Deep-sea species richness: Regional and local diversity estimates from quantitative bottom samples. *American Naturalist*, *139*(2), 313–341. https://doi.org/10.1086/285329

Gray, K. W., & Rabeling, C. (2023). Global biogeography of ant social parasites: Exploring patterns and mechanisms of an inverse latitudinal diversity gradient. *Journal of Biogeography*, *50*(2), 316–329. https://doi.org/10.1111/jbi.14528

Grieneisen, M. L., Zhan, Y., Potter, D., & Zhang, M. (2014). Biodiversity, taxonomic infrastructure, international collaboration, and new species discovery. *BioScience*, *64*(4), 322–332. https://doi.org/10.1093/biosci/biu035

Grutter, A. S. (1997). Spatiotemporal variation and feeding selectivity in the diet of the cleaner fish *Labroides dimidiatus*. *Copeia*, *2*, 346–355.

Gutow, L., Strahl, J., Wiencke, C., Franke, H.-D., & Saborowski, R. (2006). Behavioural and metabolic adaptations of marine isopods to the rafting life style. *Marine Biology*, *149*(4), 821–828. https://doi.org/10.1007/s00227-006-0257-9

Guzik, M. T., Stringer, D. N., Murphy, N. P., Cooper, S. J. B., Taiti, S., King, R. A., Humphreys, W. F., & Austin, A. D. (2019). Molecular phylogenetic analysis of Australian arid-zone oniscidean isopods (Crustacea: *Haloniscus*) reveals strong regional endemicity and new putative species. *Invertebrate Systematics*, *33*(3), 556–574. https://doi.org/10.1071/IS18070

Hadfield, K. A., & Smit, N. J. (2020). Review of the global distribution and hosts of the economically important fish parasitic isopod genus *Ceratothoa* (Isopoda: Cymothoidae), including the description of *Ceratothoa springbok* n. sp. From South Africa. *International Journal for Parasitology*, *50*(10–11), 899–919. https://doi.org/10.1016/j.ijpara.2020.07.001

Harrison, K. (1984). The morphology of the sphaeromatid brood pouch (Crustacea: Isopoda: Sphaeromatidae). *Zoological Journal of the Linnean Society*, *82*, 363–407.

Hartebrodt, L. (2019). *World list of isopod species and their authorities.xlsx*. The University of Auckland, Auckland, New Zealand. https://doi.org/10.17608/k6.auckland.9927278.v3

Hartebrodt, L. (2020). The biology, ecology, and societal importance of marine isopods. In *Encyclopedia of the World's Biomes* (Vols. 4–5). Elsevier Inc. https://doi.org/10.1016/B978-0-12-409548-9.11682-3

Hartebrodt, L. (2023a). *Global occurrences of Isopoda.xlsx*. The University of Auckland, Auckland, New Zealand. https://doi.org/10.17608/k6.auckland.24452059.v1

Hartebrodt, L. (2023b). *World list of isopod species and their authorities_updated_2023.xlsx*. The University of Auckland, Auckland, New Zealand. https://doi.org/10.17608/k6.auckland.23258675.v1

Hayashi, C., Tanaka, K., & Hirose, E. (2020). Larvae of female *Caecognathia* sp. (Isopoda: Gnathiidae) are attracted to male adults and prolong their larval phase in the absence of males. *Journal of Crustacean Biology*, *40*(2), 156–161. https://doi.org/10.1093/jcbiol/ruz094

Held, C. (2000). Phylogeny and biogeography of serolid isopods (Crustacea, Isopoda, Serolidae) and the use of ribosomal expansion segments in molecular systematics. *Molecular Phylogenetics and Evolution*, *15*(2), 165–178. https://doi.org/10.1006/mpev.1999.0739

Held, C. (2003). Molecular evidence for cryptic speciation within the widespread Antarctic crustacean *Ceratoserolis trilobitoides* (Crustacea, Isopoda). In *Antarctic Biology in a Global Context*. Backhuys Publishers. https://epic.awi.de/id/eprint/14270/

Hendrick, G. C., Nicholson, M. D., Pagan, J. A., Artim, J. M., Dolan, M. C., & Sikkel, P. C. (2023). Blood meal identification reveals extremely broad host range and host-bias in a temporary ectoparasite of coral reef fishes. *Oecologia*, *203*, 349–360. https://doi.org/10.1007/s00442-023-05468-w

Hernáez, P., Fenberg, P. B., & Rivadeneira, M. M. (2021). Departing from an ideal: An asymmetric, bimodal and non-equatorial latitudinal gradient of marine diversity in Western Atlantic burrowing shrimps (Decapoda: Axiidea and Gebiidea). *Journal of Biogeography*, *48*(3), 650–661. https://doi.org/10.1111/jbi.14030

Hessler, R. R. (1993). Swimming morphology in *Eurycope cornuta* (Isopoda: Asellota). *Journal of Crustacean Biology*, *13*(4), 667–674. https://doi.org/10.1163/193724093X00237

Hessler, R. R., & Sanders, H. L. (1967). Faunal diversity in the deep-sea. *Deep Sea Research and Oceanographic Abstracts*, *14*, 65–78. https://doi.org/10.1016/0011-7471(67)90029-0

Hessler, R. R., & Strömberg, J.-O. (1989). Behavior of janiroidean isopods (Asellota), with special reference to deep-sea genera. *Sarsia*, *74*, 145–159. https://doi.org/10.1080/00364827.1989.10413424

Hessler, R. R., Wilson, G. D. F., & Thistle, D. (1979). The deep-sea isopods: A biogeographic and phylogenetic overview. *Sarsia*, *64*(1–2), 67–75. https://doi.org/10.1080/00364827.1979.10411365

Higgs, N. D. (2016). Taxonomy in trouble? An ocean science perspective. *Ocean Challenge*, *21*(2), 10–11.

Higgs, N. D., & Attrill, M. J. (2015). Biases in biodiversity: Wide-ranging species are discovered first in the deep sea. *Frontiers in Marine Science*, *2*, Article 61. https://doi.org/10.3389/fmars.2015.00061

Hillebrand, H. (2004a). On the generality of the latitudinal diversity gradient. *The American Naturalist*, *163*(2), 192–211. https://doi.org/10.1086/381004

Hillebrand, H. (2004b). Strength, slope and variability of marine latitudinal gradients. *Marine Ecology Progress Series*, *273*, 251–267. https://doi.org/10.3354/meps273251

Holsinger, J. R., Hubbard, D. A., & Bowman, T. E. (1994). Biogeographic and ecological implications of newly discovered populations of the stygobiont isopod crustacean

Antrolana lira Bowman (Cirolanidae). *Journal of Natural History*, *28*(5), 1047–1058. https://doi.org/10.1080/00222939400770551

Holt, B. G., Lessard, J.-P., Borregaard, M. K., Fritz, S. A., Araújo, M. B., Dimitrov, D., Fabre, P.-H., Graham, C. H., Graves, G. R., Jønsson, K. A., Nogués-Bravo, D., Wang, Z., Whittaker, R. J., Fjeldså, J., & Rahbek, C. (2013). An update of Wallace's zoogeographic regions of the world. *Science*, *339*(6115), 74–78. https://doi.org/10.1126/science.1228282

Hopkins, G. W., & Freckleton, R. P. (2002). Declines in the numbers of amateur and professional taxonomists: Implications for conservation. *Animal Conservation*, *5*(3), 245–249. https://doi.org/10.1017/S1367943002002299

Hornung, E. (2011). Evolutionary adaptation of oniscidean isopods to terrestrial life: Structure, physiology and behavior. *Terrestrial Arthropod Reviews*, *4*(2), 95–130. https://doi.org/10.1163/187498311X576262

Hornung, E., Vilisics, F., & Sólymos, P. (2008). Low alpha- and high beta-diversity in terrestrial isopod assemblages in the Transdanubian region of Hungary. In M. Zimmer, F. Charfi-Cheikhrouha, & S. Taiti (Eds.), *Proceedings of the International Symposium on Terrestrial Isopod Biology - ISTIB-07* (pp. 1–11).

Horváthová, T., & Bauchinger, U. (2019). Biofilm improves isopod growth independent of the dietary cellulose content. *Physiological and Biochemical Zoology*, *92*(6), 531–543. https://doi.org/10.1086/705441

Hsieh, T. C., Ma, K. H., & Chao, A. (2022). *iNEXT: iNterpolation and EXTrapolation for species diversity* (R package version 3.0.0). http://chao.stat.nthu.edu.tw/wordpress/software-download/

Hughes, L. E., Bruce, N. L., & Osborn, K. J. (2020). *Aegiochus gracilipes* (Hansen, 1895) a senior synonym of *Aegiochus tara* Bruce, 2009 (Crustacea: Isopoda: Aegidae). *Zootaxa*, *4803*(2), 388–392. https://doi.org/10.11646/ZOOTAXA.4803.2.10

Hurtado, L. A., Mateos, M., Mattos, G., Liu, S., Haye, P. A., & Paiva, P. C. (2016). Multiple transisthmian divergences, extensive cryptic diversity, occasional long-distance dispersal, and biogeographic patterns in a marine coastal isopod with an amphi-American distribution. *Ecology and Evolution*, *6*(21), 7794–7808. https://doi.org/10.1002/ECE3.2397

Ikeda, H., Callaham Jr., M. A., Shefferson, R. P., Wenk, E. S., & Fragoso, C. (2020). A comparison of latitudinal species diversity patterns between riverine and terrestrial earthworms from the North American temperate zone. *Journal of Biogeography*, *47*(6), 1373–1382. https://doi.org/10.1111/jbi.13826

Jablonski, D., Huang, S., Roy, K., & Valentine, J. W. (2017). Shaping the latitudinal diversity gradient: New perspectives from a synthesis of paleobiology and biogeography. *The American Naturalist*, *189*(1), 1–12. https://doi.org/10.1086/689739

Jacobson, P., Bergström, U., & Eklöf, J. (2019). Size-dependent diet composition and feeding of Eurasian perch (*Perca fluviatilis*) and northern pike (*Esox lucius*) in the Baltic Sea. *Boreal Environment Research*, *24*, 137–153.

Janssen, A., Stuckas, H., Vink, A., & Martinez Arbizu, P. (2019). Biogeography and population structure of predominant macrofaunal taxa (Annelida and Isopoda) in abyssal polymetallic nodule fields: Implications for conservation and management. *Marine Biodiversity*, *49*(6), 2641–2658. https://doi.org/10.1007/s12526-019-00997-1

Jass, J., & Klausmeier, B. (2000). Endemics and immigrants: North American terrestrial isopods (Isopoda, Oniscidea) north of Mexico. *Crustaceana*, *73*(7), 771–799. https://doi.org/10.1163/156854000504804

Jennings, R. M., Golovan, O. A., & Brix, S. (2020). Integrative species delimitation of desmosomatid and nannoniscid isopods from the Kuril-Kamchatka trench, with description of a hadal species. *Progress in Oceanography*, *182*, Article 102236. https://doi.org/10.1016/J.POCEAN.2019.102236

Johannsen, N., Lins, L., Riehl, T., & Brandt, A. (2020). Changes in species composition of Haploniscidae (Crustacea: Isopoda) across potential barriers to dispersal in the Northwest Pacific. *Progress in Oceanography*, *180*, Article 102233. https://doi.org/10.1016/j.pocean.2019.102233

Johnson, W. S. (1976). Biology and population dynamics of the intertidal isopod *Cirolana harfordi*. *Marine Biology*, *36*, 343–350.

Joppa, L. N., Roberts, D. L., & Pimm, S. L. (2011a). How many species of flowering plants are there? *Proceedings of the Royal Society B: Biological Sciences*, *278*(1705), 554–559. https://doi.org/10.1098/rspb.2010.1004

Joppa, L. N., Roberts, D. L., & Pimm, S. L. (2011b). The population ecology and social behaviour of taxonomists. *Trends in Ecology & Evolution*, *26*(11), 551–553. https://doi.org/10.1016/j.tree.2011.07.010

Jormalainen, V., Merilaita, S., & Härdling, R. (2000). Dynamics of intersexual conflict over precopulatory mate guarding in two populations of the isopod *Idotea baltica*. *Animal Behaviour*, *60*(1), 85–93. https://doi.org/10.1006/anbe.2000.1429

Jormalainen, V., & Shuster, S. M. (1997). Microhabitat segregation and cannibalism in an endangered freshwater isopod, *Thermosphaeroma thermophilum*. *Oecologia*, *111*, 271–279.

Kaiser, S., Lins, L., Malyutina, M. V., Mills, S., & Lörz, A.-N. (2020). Diversity and composition of benthic asellote Isopoda from two different New Zealand continental margin habitats - implications of habitat heterogeneity, productivity and depth. *Deep Sea Research Part I: Oceanographic Research Papers*, *165*, Article 103368. https://doi.org/10.1016/j.dsr.2020.103368

Kakizaki, T., Saito, T., Ohtaka, A., & Nagasawa, K. (2003). Effects of *Acanthocephalus* sp. (Acanthocephala: Echinorhynchidae) on the body size and reproduction of isopods (*Asellus hilgendorfi*). *Limnology*, *4*, 43–46. https://doi.org/10.1007/s10201-002-0090-x

Kakui, K., Fukuchi, J., & Ohta, M. (2023). *Diexanthema hakuhomaruae* sp. Nov. (Copepoda: Siphonostomatoida: Nicothoidae) from the hadal zone in the northwestern Pacific, with an 18S molecular phylogeny. *Acta Parasitologica*, *68*, 413–419. https://doi.org/10.1007/s11686-023-00676-z

Kakui, K., Fukuchi, J., & Shimada, D. (2021). First report of marine horsehair worms (Nematomorpha: *Nectonema*) parasitic in isopod crustaceans. *Parasitology Research*, *120*, 2357–2362. https://doi.org/10.1007/s00436-021-07213-9

Kamenev, G. M., Fadeev, V. I., Selin, N. I., Tarasov, V. G., & Malakhov, V. V. (1993). Composition and distribution of macro- and meiobenthos around sublittoral hydrothermal vents in the Bay of Plenty, New Zealand. *New Zealand Journal of Marine and Freshwater Research*, *27*(4), 407–418. https://doi.org/10.1080/00288330.1993.9516582

Kato, N., Chen, C., Watanabe, H. K., Yamamoto, M., & Shimomura, M. (2022). The first bopyrid isopod from hydrothermal vents: *Pleurocryptella shinkai* sp. Nov. (Isopoda: Epicaridea) parasitizing *Shinkaia crosnieri* (Decapoda: Anomura). *Zoological Science*, *39*(3), 293–306. https://doi.org/10.2108/ZS210117

Kavanat Beerahassan, R., Dileep, N., & Pillai, D. (2021). Changes in the proximate and elemental composition of *Alitropus typus* (Crustacea: Flabellifera: Aegidae) exposed to lethal dose of bacterial consortium. *Journal of Parasitic Diseases*, *45*, 859–868. https://doi.org/10.1007/s12639-021-01374-1

Kemp, J. L., Ballot, A., Nilssen, J. P., Spikkeland, I., & Eriksen, T. E. (2020). Distribution, identification and range expansion of the common Asellidae in Northern Europe, featuring the first record of *Proasellus meridianus* in the Nordic countries. *Fauna Norvegica*, *40*, 93–108. https://doi.org/10.5324/fn.v40i0.3353

Kensley, B. (1984). The role of isopod crustaceans in the reef crest community at Carrie Bow Cay, Belize. *Marine Ecology*, *5*(1), 29–44. https://doi.org/10.1111/j.1439-0485.1984.tb00305.x

Kensley, B. (2001). Biogeography of the marine Isopoda of the Indian Ocean, with a check-list of species and records. In B. Kensley & R. C. Brusca (Eds.), *Isopod Systematics and Evolution. Crustacean Issues, 13* (pp. 205–264). Balkema: Rotterdam.

Ketmaier, V., Joyce, D. A., Horton, T., & Mariani, S. (2008). A molecular phylogenetic framework for the evolution of parasitic strategies in cymothoid isopods (Crustacea). *Journal of Zoological Systematics and Evolutionary Research*, *46*(1), 19–23. https://doi.org/10.1111/j.1439-0469.2007.00423.x

Kim, M.-J., Kim, H.-W., Lee, S.-R., Kim, N.-Y., Lee, Y.-J., Joo, H.-T., Kwak, S.-N., & Lee, S.-H. (2022). Feeding strategy of the wild Korean seahorse (*Hippocampus haema*). *Journal of Marine Science and Engineering*, *10*, Article 357. https://doi.org/10.3390/jmse10030357

Kitaura, J., & Nunomura, N. (2019). Life history of an intertidal boring isopod, *Sphaeroma sieboldii* Dollfus, 1889. *Crustacean Research*, *48*, 39–49. https://doi.org/10.18353/crustacea.48.0_39

Klapow, L. A. (1970). Ovoviviparity in the genus *Excirolana* (Crustacea: Isopoda). *Journal of Zoology*, *162*(3), 359–369. https://doi.org/10.1111/j.1469-7998.1970.tb01271.x

Klompmaker, A. A., Artal, P., van Bakel, B. W. M., Fraaije, R. H. B., & Jagt, J. W. M. (2014). Parasites in the fossil record: A Cretaceous fauna with isopod-infested decapod crustaceans, infestation patterns through time, and a new ichnotaxon. *PloS ONE*, *9*(3), Article e92551. https://doi.org/10.1371/journal.pone.0092551

Knauber, H., Kohlenbach, K., Brandt, A., & Saeedi, H. (2023). Crustaceans of the Northwest Pacific Ocean: Species richness and distribution patterns. *Journal of Sea Research*, *191*, Article 102332. https://doi.org/10.1016/j.seares.2022.102332

Kniesz, K., Brandt, A., & Riehl, T. (2018). Peritrich epibionts on the hadal isopod species *Macrostylis marionae* n. sp. From the Puerto Rico Trench used as indicator for sex-specific behaviour. *Deep Sea Research Part II: Topical Studies in Oceanography*, *148*, 105–129. https://doi.org/10.1016/j.dsr2.2017.10.007

Kocsis, Á. T., Reddin, C. J., & Kiessling, W. (2018). The stability of coastal benthic biogeography over the last 10 million years. *Global Ecology and Biogeography*, *27*(9), 1106–1120. https://doi.org/10.1111/geb.12771

Krug, A. Z., Jablonski, D., & Valentine, J. W. (2007). Contrarian clade confirms the ubiquity of spatial origination patterns in the production of latitudinal diversity gradients. *Proceedings of the National Academy of Sciences*, *104*(46), 18129–18134. https://doi.org/10.1073/pnas.0709202104

Kucska, B., Ngọc, Q. N., Havasi, M., Staszny, Á., Ivánovics, B., Vranovics, K., Griffitts, J. D., Urbányi, B., & Müller, T. (2022). *Asellus aquaticus* removal of unfertilized fish eggs and possible use in aquaculture as a biological control organism. *Research Square*. https://doi.org/10.21203/rs.3.rs-1818617/v1

Kussakin, O. G. (1973). Peculiarities of the geographical and vertical distribution of marine isopods and the problem of deep-sea fauna origin. *Marine Biology*, *23*(1), 19–34. https://doi.org/10.1007/BF00394108

Kuznetsova, D. M., & Gongalsky, K. B. (2012). Cartographic analysis of woodlice fauna of the former USSR. *ZooKeys*, *176*, 1–11. https://doi.org/10.3897/zookeys.176.2372

Larsen, B. B., Miller, E. C., Rhodes, M. K., & Wiens, J. J. (2017). Inordinate fondness multiplied and redistributed: The number of species on Earth and the new pie of life. *The Quarterly Review of Biology*, *92*(3), 229–265. https://doi.org/10.1086/693564

Lavaut, E., Guillemin, M.-L., Colin, S., Faure, A., Coudret, J., Destombe, C., & Valero, M. (2022). Pollinators of the sea: A discovery of animal-mediated fertilization in seaweed. *Science*, *377*(6605), 528–530. https://doi.org/10.1126/science.abo6661

Lawrence, M. J., & Keast, M. A. (1990). A guide to the identification of benthic Isopoda from the southern Beaufort Sea. *Canadian Manuscrpit Report of Fisheries and Aquatic Sciences 2048*, 76 p.

Lee, W. L. (1966). Color change and the ecology of the marine isopod *Idothea (Pentidothea) montereyensis* Maloney, 1933. *Ecology*, *47*(6), 930–941. https://doi.org/10.2307/1935640

Lee, W. L., & Gilchrist, B. M. (1972). Pigmentation, color change and the ecology of the marine isopod *Idotea resecata* (Stimpson). *Journal of Experimental Marine Biology and Ecology*, *10*, 1–27. https://doi.org/10.1016/0022-0981(72)90089-5

Leese, F., Agrawal, S., & Held, C. (2010). Long-distance island hopping without dispersal stages: Transportation across major zoogeographic barriers in a Southern Ocean isopod. *Naturwissenschaften*, *97*(6), 583–594. https://doi.org/10.1007/s00114-010-0674-y

Leonardsson, K. (1991). Effects of cannibalism and alternative prey on population dynamics of *Saduria entomon* (Isopoda). *Ecology*, *72*(4), 1273–1285.

Lewis, S. L., Lewis, J. J., & Orndorff, W. (2021). *Caecidotea burkensis*, new species, a unique subterranean isopod from Burke's Garden, with a synthesis of the biogeography and evolution of southwestern Virginia asellids. *Journal of Cave and Karst Studies*, *83*(2), 78–87. https://doi.org/10.4311/2020LSC0126

Lin, H. -Y., Corkrey, R., Kaschner, K., Garilao, C., & Costello, M. J. (2021). Latitudinal diversity gradients for five taxonomic levels of marine fish in depth zones. *Ecological Research*, *36*(2), 266–280. https://doi.org/10.1111/1440-1703.12193

Lindquist, N., Barber, P. H., & Weisz, J. B. (2005). Episymbiotic microbes as food and defence for marine isopods: Unique symbioses in a hostile environment. *Proceedings of the Royal Society B: Biological Sciences*, *272*, 1209–1216. https://doi.org/10.1098/rspb.2005.3082

Lins, L. S. F., Ho, S. Y. W., & Lo, N. (2017). An evolutionary timescale for terrestrial isopods and a lack of molecular support for the monophyly of Oniscidea (Crustacea: Isopoda). *Organisms Diversity & Evolution*, *17*, 813–820. https://doi.org/10.1007/s13127-017-0346-2

Lins, L. S. F., Ho, S. Y. W., Wilson, G. D. F., & Lo, N. (2012). Evidence for Permo-Triassic colonization of the deep sea by isopods. *Biology Letters*, *8*(6), 979–982. https://doi.org/10.1098/rsbl.2012.0774

Linse, K., Jackson, J. A., Malyutina, M. V., & Brandt, A. (2014). Shallow-water northern hemisphere *Jaera* (Crustacea, Isopoda, Janiridae) found on whale bones in the Southern Ocean deep sea: Ecology and description of *Jaera tyleri* sp. Nov. *PloS ONE*, *9*(3), Article e93018. https://doi.org/10.1371/journal.pone.0093018

Linsenmair, K. E. (1984). Comparative studies on the social behaviour of the desert isopod *Hemilepistus reaumuri* and of a *Porcellio* species. *Zoological Symposium*, *53*, 423–453.

Liu, J., Slik, F., Zheng, S., & Lindenmayer, D. B. (2022). Undescribed species have higher extinction risk than known species. *Conservation Letters*, *15*(3), Article e12876. https://doi.org/10.1111/CONL.12876

Lo Valvo, M., & Pieri, D. (2021). Nesting of the Spotless Starling, *Sturnus unicolor*, on the island of Favignana (Aegadian Islands, Sicily). *Rivista Italiana Di Ornitologia - Research in Ornithology*, *91*(1), 59–60. https://doi.org/10.4081/rio.2021.521

López-Orozco, C. M., Carpio-Díaz, Y. M., Borja-Arrieta, R., Navas-S., G. R., Campos-Filho, I. S., Taiti, S., Mateos, M., Olazaran, A., Caballero, I. C., Jotty, K., Gómez-Estrada, H., & Hurtado, L. A. (2022). A glimpse into a remarkable unknown diversity of oniscideans along the Caribbean coasts revealed on a tiny island. *European Journal of Taxonomy*, *793*(1), 1–50. https://doi.org/10.5852/EJT.2022.793.1643

Lourie, S. A., & Vincent, A. C. J. (2004). Using biogeography to help set priorities in marine conservation. *Conservation Biology*, *18*(4), 1004–1020. https://doi.org/10.1111/j.1523-1739.2004.00137.x

Lovejoy, T. E., Brouillet, L., Doolittle, W. F., Gonzalez, A., Green, D. M., Hall, P., Hebert, P., Herrmann, T. M., Hyde, D., Lee, J., Maddison, W. P., Otto, S. P., Sperling, F. A. H., & Thompson, R. P. (2010). Canadian taxonomy: Exploring biodiversity, creating opportunity. In *Report of the Expert Panel on Biodiversity Science: Vol. November*.

Lowry, J. K., & Myers, A. A. (2017). A phylogeny and classification of the Amphipoda with the establishment of the new order Ingolfiellida (Crustacea: Peracarida). *Zootaxa*, *4265*(1), 001–089. https://doi.org/10.11646/ZOOTAXA.4265.1.1

Lu, J., Taiti, S., Li, S., Lu, Y., Zhuo, D., Wang, X., & Bai, M. (2023). First fossil of Tylidae (Isopoda: Oniscidea) in Kachin amber, Myanmar, with a list of all Oniscidea fossil records. *Fossils*, *1*, 15–33. https://doi.org/10.3390/fossils1010003

Lupetti, P., Montesanto, G., Ciolfi, S., Marri, L., Gentile, M., Paccagnini, E., & Lombardo, B. M. (2013). Iridovirus infection in terrestrial isopods from Sicily (Italy). *Tissue and Cell*, *45*(5), 321–327. https://doi.org/10.1016/j.tice.2013.05.001

Malek-Hosseini, M. J., Jugovic, J., Fatemi, Y., Kuntner, M., Kostanjšek, R., Douady, C. J., & Malard, F. (2022). A new obligate groundwater species of *Asellus* (Isopoda, Asellidae) from Iran. *Subterranean Biology*, *42*, 97–124. https://doi.org/10.3897/SUBTBIOL.42.79447

Malyutina, M. V., & Brandt, A. (2020). Munnopsidae (Crustacea, Isopoda, Asellota) from the Kuril–Kamchatka Trench with a regional and inter-ocean comparison of their biogeographic and richness patterns. *Progress in Oceanography*, *183*, Article 102289. https://doi.org/10.1016/j.pocean.2020.102289

Malyutina, M. V., Frutos, I., & Brandt, A. (2018). Diversity and distribution of the deep-sea Atlantic *Acanthocope* (Crustacea, Isopoda, Munnopsidae), with description of two new species. *Deep Sea Research Part II: Topical Studies in Oceanography*, *148*, 130–150. https://doi.org/10.1016/j.dsr2.2017.11.003

Malyutina, M. V., & Golovan, O. A. (2022). The first record of Asellota (Isopoda) from hydrothermal vent biotopes of the submarine Piip Volcano, Bering Sea, with descriptions of two new species of Munnopsidae. *Deep Sea Research Part II: Topical Studies in Oceanography*, *202*, Article 105137. https://doi.org/10.1016/j.dsr2.2022.105137

Manes, S., Costello, M. J., Beckett, H., Debnath, A., Devenish-Nelson, E., Grey, K., Jenkins, R., Khan, T. M., Kiessling, W., Krause, C., Maharaj, S. S., Midgley, G. F., Price, J., Talukdar, G., & Vale, M. M. (2021). Endemism increases species' climate change risk in areas of global biodiversity importance. *Biological Conservation*, *257*, Article 109070. https://doi.org/10.1016/J.BIOCON.2021.109070

Markham, J. C. (1986). Evolution and zoogeography of the Isopoda Bopyridae, parasites of Crustacea Decapoda. In R. H. Gore & K. L. Heck (Eds.), *Crustacean Biogeography. Crustacean Issues, 4* (pp. 143–164). Balkema: Rotterdam.

Martin, J. W. (2014). Introduction to the Peracarida. In J. W. Martin, J. Olesen, & J. T. Høeg (Eds.), *Atlas of Crustacean Larvae* (p. 194). Johns Hopkins University Press.

Martin, J. W., & Davis, G. E. (2001). An updated classification of the recent Crustacea. *Natural History Museum of Los Angeles County, Science Series*, *39*, 1–124.

McClain, C. R., Nunnally, C., Dixon, R., Rouse, G. W., & Benfield, M. (2019). Alligators in the abyss: The first experimental reptilian food fall in the deep ocean. *PloS ONE*, *14*(12), Article e0225345. https://doi.org/10.1371/journal.pone.0225345

Menegotto, A., & Rangel, T. F. (2018). Mapping knowledge gaps in marine diversity reveals a latitudinal gradient of missing species richness. *Nature Communications*, *9*, Article 4713. https://doi.org/10.1038/s41467-018-07217-7

Menzies, R. J., & Glynn, P. W. (1968). The common marine isopod Crustacea of Puerto Rico - A handbook for marine biologists. *Studies on the Fauna of Curaçao and Other Caribbean Islands*, *27*(1), 1–133.

Menzies, R. J., & Kruczynski, W. L. (1983). Isopod Crustacea (exclusive of Epicaridea). *Memoirs of the Hourglass Cruises*, *VI*(Part I), 1–126.

Messelink, G. J., & Bloemhard, C. M. J. (2007). Woodlice (Isopoda) and millipedes (Diplopoda): Control of rare greenhouse pests. *Proceedings of the Netherlands Entomological Society Meeting*, *18*, 43–49.

Mészárosné Póss, A., Südiné Fehér, A., Tóthné Bogdányi, F., & Tóth, F. (2022). The spread of the soil-borne pathogen *Fusarium solani* in stored potato can be controlled by terrestrial woodlice (Isopoda: Oniscidea). *Agriculture*, *12*, Article 45. https://doi.org/10.3390/agriculture12010045

Mezhov, B. V. (1993). Three new species of *Macrostylis* G. O. Sars, 1864 (Crustacea Isopoda Asellota, Macrostylidae) from the Indian Ocean. *Arthropoda Selecta*, *2*(3), 3–9.

Montesanto, G., Musarra Pizzo, G., Caruso, D., & Lombardo, B. M. (2012). The postmarsupial development of *Porcellio siculoccidentalis*, with some data on reproductive biology (Crustacea, Isopoda, Oniscidea). *ZooKeys*, *176*, 87–101. https://doi.org/10.3897/zookeys.176.2369

Monticelli Cardoso, G., Bastos-Pereira, R., & Lopes Ferreira, R. (2022). A new species of *Chaimowiczia* from the karstic Serra do Ramalho plateau, Brazil (Oniscidea, Synocheta, Styloniscidae). *Subterranean Biology*, *42*, 139–149. https://doi.org/10.3897/SUBTBIOL.42.80274

Mora, C., Tittensor, D. P., Adl, S., Simpson, A. G. B., & Worm, B. (2011). How many species are there on Earth and in the ocean? *PloS Biology*, *9*(8), Article e1001127. https://doi.org/10.1371/journal.pbio.1001127

Muggeo, V. M. R. (2008). Segmented: An R package to fit regression models with broken-line relationships. *R News*, *8*(1), 20–25. http://cran.r-project.org/doc/Rnews/

Nagler, C., Hyžný, M., & Haug, J. T. (2017). 168 million years old "marine lice" and the evolution of parasitism within isopods. *BMC Evolutionary Biology*, *17*, Article 76. https://doi.org/10.1186/s12862-017-0915-1

Nakamachi, T., & Asakura, A. (2020). Reproductive aggregations of *Dynoides dentisinus* (Crustacea: Peracarida), an intertidal isopod with remarkable sexual dimorphism. *The Biological Bulletin*, *239*, 40–50. https://doi.org/10.1086/710080

Newman, B. K., Wooldridge, T. H., & Cockcroft, A. C. (2007). Aspects of the biology and ecology of the estuarine cirolanid isopod, *Cirolana fluviatilis*. *African Zoology*, *42*(1), 12–22. https://doi.org/10.1080/15627020.2007.11407372

Nicholson, M. D., Hendrick, G. C., Packard, A. J., Strobel, D. L., Vondriska, C., & Sikkel, P. C. (2020). Vertical limits of host infestation by gnathiid isopods (Isopoda: Gnathiidae) parasitic on Caribbean coral reef fishes. *Journal of Crustacean Biology*, *40*(6), 866–871. https://doi.org/10.1093/jcbiol/ruaa067

Oanh, L. T. K., & Boyko, C. B. (2020). *Cancrion khanhensis* sp. Nov. (Crustacea: Isopoda: Entoniscidae) infesting *Monomia haanii* (Stimpson, 1858) (Crustacea: Brachyura: Portunidae) from Nha Trang Bay, Khanh Hoa, Vietnam, with remarks on larval stages of entoniscids and description of a new family, genus and two new species of hyperparasites infesting entoniscids. *Zootaxa*, *4894*(3), 366–386. https://doi.org/10.11646/zootaxa.4894.3.4

OBIS. (2022). [Distribution records of Isopoda (Latreille, 1817)] [Dataset]. Retrieved from *Ocean Biogeographic Information System*. Intergovernmental Oceanographic Commission of UNESCO. www.iobis.org. Accessed 2022-06-10.

O'Callaghan, I., Harrison, S., Fitzpatrick, D., & Sullivan, T. (2019). The freshwater isopod *Asellus aquaticus* as a model biomonitor of environmental pollution: A review. *Chemosphere*, *235*, 498–509. https://doi.org/10.1016/j.chemosphere.2019.06.217

O'Hara, T. D., Williams, A., Ahyong, S. T., Alderslade, P., Alvestad, T., Bray, D., Burghardt, I., Budaeva, N., Criscione, F., Crowther, A. L., Ekins, M., Eléaume, M., Farrelly, C. A., Finn, J. K., Georgieva, M. N., Graham, A., Gomon, M., Gowlett-Holmes, K., Gunton, L. M., … Bax, N. J. (2020). The lower bathyal and abyssal seafloor fauna of eastern Australia. *Marine Biodiversity Records*, *13*, Article 11. https://doi.org/10.1186/s41200-020-00194-1

Olson, D. M., Dinerstein, E., Wikramanayake, E. D., Burgess, N. D., Powell, G. V. N., Underwood, E. C., D'Amico, J. A., Itoua, I., Strand, H. E., Morrison, J. C., Loucks, C. J., Allnutt, T. F., Ricketts, T. H., Kura, Y., Lamoreux, J. F., Wettengel, W. W., Hedao, P., & Kassem, K. R. (2001). Terrestrial ecoregions of the world: A new map of life on Earth. *BioScience*, *51*(11), 933–938. https://doi.org/10.1641/0006-3568(2001)051[0933:TEOTWA]2.0.CO;2

Orr, M. C., Hughes, A. C., Chesters, D., Pickering, J., Zhu, C.-D., & Ascher, J. S. (2021). Global patterns and drivers of bee distribution. *Current Biology*, *31*(3), 451–458. https://doi.org/10.1016/j.cub.2020.10.053

Pagès-Escolà, M., Bock, P., Gordon, D. P., Wilson, S. P., Linares, C., Hereu, B., & Costello, M. J. (2020). Progress in the discovery of extant and fossil bryozoans. *Marine Ecology Progress Series*, *635*, 71–79. https://doi.org/10.3354/MEPS13201

Pamungkas, J., Glasby, C. J., & Costello, M. J. (2021). Biogeography of polychaete worms (Annelida) of the world. *Marine Ecology Progress Series*, *657*, 147–159. https://doi.org/10.3354/meps13531

Pamungkas, J., Glasby, C. J., Read, G. B., Wilson, S. P., & Costello, M. J. (2019). Progress and perspectives in the discovery of polychaete worms (Annelida) of the world. *Helgoland Marine Research*, *73*, Article 4. https://doi.org/10.1186/s10152-019-0524-z

Pante, E., Schoelinck, C., & Puillandre, N. (2015). From integrative taxonomy to species description: One step beyond. *Systematic Biology*, *64*(1), 152–160. https://doi.org/10.1093/SYSBIO/SYU083

Paris, O. H. (1963). The ecology of *Armadillidium vulgare* (Isopoda: Oniscoidea) in California grassland: Food, enemies, and weather. *Ecological Monographs*, *33*(1), 1–22.

Parmentier, T., Vanderheyden, A., Dekoninck, W., & Wenseleers, T. (2017). Body size in the ant-associated isopod *Platyarthrus hoffmannseggii* is host-dependent. *Biological Journal of the Linnean Society*, *121*(2), 305–311. https://doi.org/10.1093/biolinnean/blw052

Pascual, S., Vega, M. A., Rocha, F. J., & Guerra, A. (2002). First report of an endoparasitic epicaridean isopod infecting cephalopods. *Journal of Wildlife Diseases*, *38*(2), 473–477. https://doi.org/10.7589/0090-3558-38.2.473

Paula, J. R., Sun, D., Pissarra, V., Narvaez, P., Rosa, R., Grutter, A. S., & Sikkel, P. C. (2021). The role of corals on the abundance of a fish ectoparasite in the Great Barrier Reef. *Coral Reefs*, *40*, 535–542. https://doi.org/10.1007/s00338-021-02051-8

Pearman, W. S., Wells, S. J., Silander, O. K., Freed, N. E., & Dale, J. (2020). Concordant geographic and genetic structure revealed by genotyping-by-sequencing in a New Zealand marine isopod. *Ecology and Evolution*, *10*(24), 13624–13639. https://doi.org/10.1002/ece3.6802

Poinar Jr., G. O. (1981). *Thaumamermis cosgrovei* n. gen., n. sp. (Mermithidae: Nematoda) parasitizing terrestrial isopods (Isopoda: Oniscoidea). *Systematic Parasitology*, *2*, 261–266.

Poore, G. C. B. (2005). Peracarida: Monophyly, relationships and evolutionary success. *Nauplius*, *13*(1), 1–27.

Poore, G. C. B., Avery, L., Błażewicz-Paszkowycz, M., Browne, J. G., Bruce, N. L., Gerken, S., Glasby, C. J., Greaves, E., McCallum, A. W., Staples, D., Syme, A., Taylor, J., Walker-Smith, G., Warne, M., Watson, C., Williams, A., Wilson, R. S., & Woolley, S. (2015). Invertebrate diversity of the unexplored marine western margin of Australia: Taxonomy and implications for global biodiversity. *Marine Biodiversity*, *45*(2), 271–286. https://doi.org/10.1007/S12526-014-0255-Y/FIGURES/5

Poore, G. C. B., & Bruce, N. L. (2012). Global diversity of marine isopods (except Asellota and crustacean symbionts). *PloS ONE*, *7*(8), Article e43529. https://doi.org/10.1371/journal.pone.0043529

Poore, G. C. B., Just, J., & Cohen, B. F. (1994). Composition and diversity of Crustacea Isopoda of the southeastern Australian continental slope. *Deep Sea Research Part I: Oceanographic Research Papers*, *41*(4), 677–693. https://doi.org/10.1016/0967-0637(94)90049-3

Poore, G. C. B., & Wilson, G. D. F. (1993). Marine species richness. *Nature*, *361*, 597–598. https://doi.org/10.1038/361597a0

Pos, E., Guevara Andino, J. E., Sabatier, D., Molino, J. -F., Pitman, N., Mogollón, H., Neill, D., Cerón, C., Rivas, G., Di Fiore, A., Thomas, R., Tirado, M., Young, K. R., Wang, O., Sierra, R., García-Villacorta, R., Zagt, R., Palacios, W., Aulestia, M., & ter Steege, H. (2014). Are all species necessary to reveal ecologically important patterns? *Ecology and Evolution*, *4*(24), 4626–4636. https://doi.org/10.1002/ece3.1246

Poulin, R., & Pérez-Ponce de León, G. (2017). Global analysis reveals that cryptic diversity is linked with habitat but not mode of life. *Journal of Evolutionary Biology*, *30*(3), 641–649. https://doi.org/10.1111/JEB.13034

Powell, C. V. L., & Halcrow, K. (1982). The surface microstructure of marine and terrestrial Isopoda (Crustacea, Peracarida). *Zoomorphology*, *101*, 151–164.

R Core Team. (2021). *R: A Language and Environment for Statistical Computing*. R Foundation for Statistical Computing. https://www.r-project.org/

Rajkumar, M., Kumaraguru Vasagam, K. P., Perumal, P., & Trilles, J. P. (2005). First record of *Cymothoa indica* (Crustacea, Isopoda, Cymothoidae) infecting the cultured catfish *Mystus gulio* in India. *Diseases of Aquatic Organisms*, *65*, 269–272.

Raupach, M. J., Malyutina, M. V., Brandt, A., & Wägele, J. W. (2007). Molecular data reveal a highly diverse species flock within the munnopsoid deep-sea isopod *Betamorpha fusiformis* (Barnard, 1920) (Crustacea: Isopoda: Asellota) in the Southern Ocean. *Deep Sea Research Part II: Topical Studies in Oceanography*, *54*, 1820–1830. https://doi.org/10.1016/J.DSR2.2007.07.009

Raupach, M. J., Mayer, C., Malyutina, M. V., & Wägele, J. W. (2009). Multiple origins of deep-sea Asellota (Crustacea: Isopoda) from shallow waters revealed by molecular data. *Proceedings of the Royal Society B: Biological Sciences*, *276*, 799–808. https://doi.org/10.1098/rspb.2008.1063

Rayes, C. A., Beattie, J., & Duggan, I. C. (2015). Boring through history: An environmental history of the extent, impact and management of marine woodborers in a global and local context, 500 BCE to 1930s CE. *Environment and History*, *21*, 477–512. https://doi.org/10.3197/096734015X14414683716163

Recuero, E., & Rodríguez-Flores, P. C. (2019). On the geographic distribution of the uncommon Iberian endemic *Armadillidium mateui* Vandel, 1953 (Crustacea, Isopoda, Armadillidiidae). *Graellsia*, *75*(2), Article e096. https://doi.org/10.3989/graellsia.2019.v75.239

Reed, M. L., Hoback, W. W., & Long, J. M. (2018). Winter and spring diet of the Orangebelly Darter, *Etheostoma radiosum*, among tributaries of the Lower Mountain Fork River. *The Southwestern Naturalist*, *63*(2), 146–148. https://doi.org/10.1894/0038-4909-63-2-146

Rehm, A., & Humm, H. J. (1973). *Sphaeroma terebrans*: A threat to the mangroves of southwestern Florida. *Science*, *182*, 173–174.

Rex, M. A., Etter, R. J., & Stuart, C. T. (1997). Large-scale patterns of species diversity in the deep-sea benthos. In R. F. G. Ormond, J. D. Gage, & M. V. Angel (Eds.), *Marine Biodiversity - Patterns and Processes* (pp. 94–116). Cambridge University Press, United Kingdom. https://doi.org/10.1017/cbo9780511752360.006

Rex, M. A., Stuart, C. T., Hessler, R. R., Allen, J. A., Sanders, H. L., & Wilson, G. D. F. (1993). Global-scale latitudinal patterns of species diversity in the deep-sea benthos. *Nature*, *365*, 636–639. https://doi.org/10.1038/365636a0

Richter, S., & Scholtz, G. (2001). Phylogenetic analysis of the Malacostraca (Crustacea). *Journal of Zoological Systematics and Evolutionary Research*, *39*, 113–136. https://doi.org/10.1046/j.1439-0469.2001.00164.x

Riehl, T., Wölfl, A.-C., Augustin, N., Devey, C. W., & Brandt, A. (2020). Discovery of widely available abyssal rock patches reveals overlooked habitat type and prompts rethinking deep-sea biodiversity. *Proceedings of the National Academy of Sciences*, *117*(27), 15450–15459. https://doi.org/10.1073/pnas.1920706117

Riseman, S. F., & Brusca, R. C. (2002). Taxonomy, phylogeny and biogeography of *Politolana* Bruce, 1981 (Crustacea: Isopoda: Cirolanidae). *Zoological Journal of the Linnean Society*, *134*(1), 57–140. https://doi.org/10.1046/j.1096-3642.2002.00002.x

Rivadeneira, M. M., & Poore, G. C. B. (2020). Latitudinal gradient of diversity of marine crustaceans: Towards a synthesis. In G. C. B. Poore & M. Thiel (Eds.), *Evolution and Biogeography: Volume 8* (pp. 389–412). Oxford University Press. https://doi.org/10.1093/oso/9780190637842.003.0015

Rivadeneira, M. M., Thiel, M., González, E. R., & Haye, P. A. (2011). An inverse latitudinal gradient of diversity of peracarid crustaceans along the Pacific Coast of South America: out of the deep south. *Global Ecology and Biogeography*, *20*(3), 437–448. https://doi.org/10.1111/j.1466-8238.2010.00610.x

Robin, N., Gueriau, P., Luque, J., Jarvis, D., Daley, A. C., & Vonk, R. (2021). The oldest peracarid crustacean reveals a Late Devonian freshwater colonization by isopod relatives. *Biology Letters*, *17*, Article 20210226. https://doi.org/10.1098/rsbl.2021.0226

Rosa, R., Pissarra, V., Borges, F. O., Xavier, J., Gleadall, I. G., Golikov, A., Bello, G., Morais, L., Lishchenko, F., Roura, Á., Judkins, H., Ibáñez, C. M., Piatkowski, U., Vecchione, M., & Villanueva, R. (2019). Global patterns of species richness in coastal cephalopods. *Frontiers in Marine Science*, *6*, Article 469. https://doi.org/10.3389/fmars.2019.00469

Roslin, T., Hardwick, B., Novotny, V., Petry, W. K., Andrew, N. R., Asmus, A., Barrio, I. C., Basset, Y., Boesing, A. L., Bonebrake, T. C., Cameron, E. K., Dáttilo, W., Donoso, D. A., Drozd, P., Gray, C. L., Hik, D. S., Hill, S. J., Hopkins, T., Huang, S., … Slade, E. M. (2017). Higher predation risk for insect prey at low latitudes and elevations. *Science*, *356*(6339), 742–744. https://doi.org/10.1126/science.aaj1631

Roswell, M., Dushoff, J., & Winfree, R. (2021). A conceptual guide to measuring species diversity. *Oikos*, *130*(3), 321–338. https://doi.org/10.1111/oik.07202

Rundio, D. E., & Lindley, S. T. (2021). Importance of non-native isopods and other terrestrial prey resources to steelhead/rainbow trout *Oncorhynchus mykiss* in coastal streams in Big Sur, California. *Ecology of Freshwater Fish*, *30*(4), 419–432. https://doi.org/10.1111/eff.12594

Rusconi, J. M., Eliceche, D., Salas, A., Balcazar, D., Ibañez Shimabukuro, M., & Achinelly, M. F. (2023). *Agamermis* sp. (Nematoda: Mermithidae) parasitizing *Armadillidium* *vulgare* (Crustacea: Isopoda) in Argentina. *Journal of Helminthology*, *97*, Article e24. https://doi.org/10.1017/S0022149X23000068

Saeedi, H., Brandt, A., & Jacobsen, N. L. (2022). Biodiversity and distribution of Isopoda and Polychaeta along the Northwestern Pacific Ocean and the Arctic Ocean. *Biodiversity Informatics*, *17*, 10–26. https://doi.org/10.17161/bi.v17i.15581

Saeedi, H., Simões, M., & Brandt, A. (2020). Biodiversity and distribution patterns of deep-sea fauna along the temperate NW Pacific. *Progress in Oceanography*, *183*, Article 102296. https://doi.org/10.1016/j.pocean.2020.102296

Salemaa, H. (1986). Breeding biology and microhabitat utilization of the intertidal isopod *Idotea granulosa* Rathke, in the Irish Sea. *Estuarine, Coastal and Shelf Science*, *22*, 335–355.

Sangster, G., & Luksenburg, J. A. (2015). Declining rates of species described per taxonomist: Slowdown of progress or a side-effect of improved quality in taxonomy? *Systematic Biology*, *64*(1), 144–151. https://doi.org/10.1093/sysbio/syu069

Sanil, N. K., Vikas, P. A., Ratheesh, T. B., George, K. C., & Vijayan, K. K. (2009). Mortalities caused by the crustacean isopod, *Cirolana fluviatilis*, in tropical, cage-cultured Asian seabass, *Lates calcarifer*: A case study from the southwest coast of India. *Aquaculture Research*, *40*(14), 1626–1633. https://doi.org/10.1111/j.1365-2109.2009.02263.x

Saravanakumar, A., Balasubramanian, T., Raja, K., & Trilles, J. P. (2012). A massive infestation of sea snakes by cymothoid isopods. *Parasitology Research*, *110*, 2529–2531. https://doi.org/10.1007/s00436-011-2795-4

Saska, P. (2008). Granivory in terrestrial isopods. *Ecological Entomology*, *33*, 742–747. https://doi.org/10.1111/j.1365-2311.2008.01026.x

Schädel, M., Hörnig, M. K., Hyžný, M., & Haug, J. T. (2021). Mass occurrence of small isopodan crustaceans in 100-million-year-old amber: An extrodinary view on behaviour of extinct organisms. *PalZ*, *95*, 429–445. https://doi.org/10.1007/s12542-021-00564-9

Scheffers, B. R., Joppa, L. N., Pimm, S. L., & Laurance, W. F. (2012). What we know and don’t know about Earth’s missing biodiversity. *Trends in Ecology & Evolution*, *27*(9), 501–510. https://doi.org/10.1016/j.tree.2012.05.008

Schlick-Steiner, B. C., Seifert, B., Stauffer, C., Christian, E., Crozier, R. H., & Steiner, F. M. (2007). Without morphology, cryptic species stay in taxonomic crypsis following discovery. *Trends in Ecology & Evolution*, *22*(8), 391–392. https://doi.org/10.1016/J.TREE.2007.05.004

Schmidt, C., & Wägele, J. W. (2001). Morphology and evolution of respiratory structures in the pleopod exopodites of terrestrial Isopoda (Crustacea, Isopoda, Oniscidea). *Acta Zoologica*, *82*(4), 315–330. https://doi.org/10.1046/j.1463-6395.2001.00092.x

Schnurr, S., Brandt, A., Brix, S., Fiorentino, D., Malyutina, M. V., & Svavarsson, J. (2014). Composition and distribution of selected munnopsid genera (Crustacea, Isopoda, Asellota) in Icelandic waters. *Deep Sea Research Part I: Oceanographic Research Papers*, *84*, 142–155. https://doi.org/10.1016/j.dsr.2013.11.004

Schnurr, S., Osborn, K. J., Malyutina, M. V., Jennings, R. M., Brix, S., Driskell, A., Svavarsson, J., & Martinez Arbizu, P. (2018). Hidden diversity in two species complexes of munnopsid isopods (Crustacea) at the transition between the northernmost North Atlantic and the Nordic Seas. *Marine Biodiversity*, *48*(2), 813–843. https://doi.org/10.1007/S12526-018-0877-6

Schram, F. R. (1970). Isopod from the Pennsylvanian of Illinois. *Science*, *169*(3948), 854–855. https://doi.org/10.1126/science.169.3948.854

Selden, P. A., Wilson, G. D. F., Simonetto, L., & Dalla Vecchia, F. M. (2016). First fossil asellote (Isopoda: Asellota), from the Upper Triassic (Norian) of the Carnic Prealps (Friuli, northeastern Italy). *Journal of Crustacean Biology*, *36*(1), 68–86. https://doi.org/10.1163/1937240X-00002387

Sfenthourakis, S. (1996). A biogeographical analysis of terrestrial isopods (Isopoda, Oniscidea) from the central Aegean islands (Greece). *Journal of Biogeography*, *23*(5), 687–698. https://doi.org/10.1111/j.1365-2699.1996.tb00029.x

Sfenthourakis, S., & Giokas, S. (1998). A biogeographical analysis of Greek oniscidean endemism. *Israel Journal of Zoology*, *44*(3–4), 273–282.

Sfenthourakis, S., & Hornung, E. (2018). Isopod distribution and climate change. *ZooKeys*, *801*, 25–61. https://doi.org/10.3897/zookeys.801.23533

Sharifian, S., Kamrani, E., & Saeedi, H. (2020). Global biodiversity and biogeography of mangrove crabs: Temperature, the key driver of latitudinal gradients of species richness. *Journal of Thermal Biology*, *92*, Article 102692. https://doi.org/10.1016/j.jtherbio.2020.102692

Shuster, S. M. (1981). Sexual selection in the socorro isopod, *Thermosphaeroma thermophilum* (Cole) (Crustacea: Peracarida). *Animal Behaviour*, *29*(3), 698–707. https://doi.org/10.1016/S0003-3472(81)80004-8

Shuster, S. M. (1987). Alternative reproductive behaviors: Three discrete male morphs in *Paracerceis sculpta*, an intertidal isopod from the northern Gulf of California. *Journal of Crustacean Biology*, *7*(2), 318–327.

Si, A., Bellwood, O., & Alexander, C. G. (2002). Evidence for filter-feeding by the wood-boring isopod, *Sphaeroma terebrans* (Crustacea: Peracarida). *Journal of Zoology*, *256*(4), 463–471. https://doi.org/10.1017/S095283690200050X

Smit, N. J., Bruce, N. L., & Hadfield, K. A. (2014). Global diversity of fish parasitic isopod crustaceans of the family Cymothoidae. *International Journal for Parasitology: Parasites and Wildlife*, *3*, 188–197. https://doi.org/10.1016/j.ijppaw.2014.03.004

Songvorawit, N., Quicke, D. L. J., & Butcher, B. A. (2021). Taxonomic progress and diversity of ichneumonoid wasps (Hymenoptera: Ichneumonoidea) in Southeast Asia. *Tropical Natural History*, *21*(1), 78–93. https://li01.tci-thaijo.org/index.php/tnh/article/view/248068

Soto, J. M. R., & Mincarone, M. M. (2001). Distribution and morphology of the giant isopods *Bathynomus giganteus* and *Bathynomus miyarei* (Flabellifera, Cirolanidae) off southern Brazil. *Mare Magnum*, *1*(2), 141–145.

Spalding, M. D., Agostini, V. N., Rice, J., & Grant, S. M. (2012). Pelagic provinces of the world: A biogeographic classification of the world's surface pelagic waters. *Ocean & Coastal Management*, *60*, 19–30. https://doi.org/10.1016/j.ocecoaman.2011.12.016

Spalding, M. D., Fox, H. E., Allen, G. R., Davidson, N., Ferdaña, Z. A., Finlayson, M., Halpern, B. S., Jorge, M. A., Lombana, A., Lourie, S. A., Martin, K. D., McManus, E., Molnar, J., Recchia, C. A., & Robertson, J. (2007). Marine ecoregions of the world: A bioregionalization of coastal and shelf areas. *BioScience*, *57*(7), 573–583. https://doi.org/10.1641/B570707

Stehli, F. G., Douglas, R. G., & Newell, N. D. (1969). Generation and maintenance of gradients in taxonomic diversity. *Science*, *164*(3882), 947–949. https://doi.org/10.1126/science.164.3882.947

Stepien, C. A., & Brusca, R. C. (1985). Nocturnal attacks on nearshore fishes in southern California by crustacean zooplankton. *Marine Ecology Progress Series*, *25*, 91–105.

Stinnesbeck, E. S., Wägele, J. W., Herder, F., Rust, J., & Stinnesbeck, W. (2022). A fish-parasitic isopod (Cymothoidae) on the pachyrhizodont *Goulmimichthys roberti* from the lower Turonian (Upper Cretaceous) Vallecillo plattenkalk, NE Mexico. *Cretaceous Research*, *129*, Article 105019. https://doi.org/10.1016/j.cretres.2021.105019

Stransky, B., Svavarsson, J., Poore, G. C. B., & Kihara, T. C. (2020). Revision of *Pleuroprion* zur Strassen, 1903 (Holidoteidae) and re-evaluation of *Spectrarcturus* Schultz, 1981 (Arcturidae) (Crustacea, Isopoda, Valvifera). *Zootaxa*, *4894*(1), 001–052. https://doi.org/10.11646/ZOOTAXA.4894.1.1

Sutton, T. T., Clark, M. R., Dunn, D. C., Halpin, P. N., Rogers, A. D., Guinotte, J., Bograd, S. J., Angel, M. V., Perez, J. A. A., Wishner, K., Haedrich, R. L., Lindsay, D. J., Drazen, J. C., Vereshchaka, A., Piatkowski, U., Morato, T., Błachowiak-Samołyk, K., Robison, B. H., Gjerde, K. M., … Heino, M. (2017). A global biogeographic classification of the mesopelagic zone. *Deep Sea Research Part I: Oceanographic Research Papers*, *126*, 85–102. https://doi.org/10.1016/j.dsr.2017.05.006

Svane, I., & Barnett, J. (2008). The occurrence of benthic scavengers and their consumption at tuna farms off Port Lincoln, South Australia. *Journal of Experimental Marine Biology and Ecology*, *363*, 110–117. https://doi.org/10.1016/j.jembe.2008.06.028

Svavarsson, J. (2006). New species of Gnathiidae (Crustacea, Isopoda, Cymothoida) from seamounts off northern New Zealand. *Zootaxa*, *1173*, 39–56.

Tait, J. (1917). V.—Experiments and observations on Crustacea: Part II. Moulting of Isopods. *Proceedings of the Royal Society of Edinburgh*, *37*, 59–68. https://doi.org/10.1017/S0370164600023506

Taiti, S., & Monticelli Cardoso, G. (2020). New species and records of *Exalloniscus* Stebbing, 1911 from southern Asia (Malacostraca, Isopoda, Oniscidea). *Tropical Zoology*, *33*(4), 125–158. https://doi.org/10.4081/TZ.2020.83

Talley, T. S., Crooks, J. A., & Levin, L. A. (2001). Habitat utilization and alteration by the invasive burrowing isopod, *Sphaeroma quoyanum*, in California salt marshes. *Marine Biology*, *138*, 561–573.

Tanaka, K. (2007). Life history of gnathiid isopods - current knowledge and future directions. *Plankton & Benthos Research*, *2*(1), 1–11.

Tanaka, K., & Nishi, E. (2011). Male dimorphism in the harem-forming gnathiid isopod *Elaphognathia discolor* (Crustacea: Isopoda). *Zoological Science*, *28*, 587–592. https://doi.org/10.2108/zsj.28.587

Tancoigne, E., & Dubois, A. (2013). Taxonomy: No decline, but inertia. *Cladistics*, *29*, 567–570. https://doi.org/10.1111/cla.12019

Taxonomy Decadal Plan Working Group. (2018). *Discovering biodiversity: A decadal plan for taxonomy and biosystematics in Australia and New Zealand 2018–2027*.

Thiel, M. (1999). Reproductive biology of a wood-boring isopod, *Sphaeroma terebrans*, with extended parental care. *Marine Biology*, *135*, 321–333. https://doi.org/10.1007/s002270050630

Thiel, M. (2002). Reproductive biology of a small isopod symbiont living on a large isopod host: From the maternal marsupium to the protective grip of guarding males. *Marine Biology*, *141*(1), 175–183. https://doi.org/10.1007/s00227-002-0801-1

Thiel, M. (2003). Reproductive biology of *Limnoria chilensis*: Another boring peracarid species with extended parental care. *Journal of Natural History*, *37*(14), 1713–1726. https://doi.org/10.1080/00222930210125416

Thomas, P. W., & Thomas, H. W. (2022). Mycorrhizal fungi and invertebrates: Impacts on *Tuber melanosporum* ascospore dispersal and lifecycle by isopod mycophagy. *Food Webs*, *33*, Article e00260. https://doi.org/10.1016/j.fooweb.2022.e00260

Tiemensma, M., Bruce, N. L., & Willan, R. C. (2017). Post-mortem human cadaver scavenging by marine crustaceans (Isopoda: Cirolanidae) in tropical waters. *Forensic Science, Medicine and Pathology*, *13*, 515–517. https://doi.org/10.1007/s12024-017-9926-x

Toft, S., & Macías-Hernández, N. (2021). Prey acceptance and metabolic specialisations in some Canarian Dysdera spiders. *Journal of Insect Physiology*, *131*, Article 104227. https://doi.org/10.1016/j.jinsphys.2021.104227

Triantis, K. A., Sfenthourakis, S., & Mylonas, M. (2008). Biodiversity patterns of terrestrial isopods from two island groups in the Aegean Sea (Greece): Species—area relationship, small island effect, and nestedness. *Écoscience*, *15*(2), 169–181. https://doi.org/10.2980/15-2-3065

Tsai, M.-L., Li, J.-J., & Dai, C.-F. (1999). Why selection favors protandrous sex change for the parasitic isopod, *Ichthyoxenus fushanensis* (Isopoda: Cymothoidae). *Evolutionary Ecology*, *13*, 327–338.

Van As, L. L. (2019). Hypersymbionts and hyperparasites of parasitic Crustacea. In N. J. Smit & K. A. Hadfield (Eds.), *Parasitic Crustacea. Zoological Monographs, Vol. 3* (pp. 343–385). Springer, Cham. https://doi.org/10.1007/978-3-030-17385-2_8

Van Der Spuy, L., Erasmus, J. H., Nachev, M., Schaeffner, B. C., Sures, B., Wepener, V., & Smit, N. J. (2023). The use of fish parasitic isopods as element accumulation indicators in marine pollution monitoring. *Marine Pollution Bulletin*, *194*, Article 115385. https://doi.org/10.1016/j.marpolbul.2023.115385

Victorero, L., Samadi, S., O'Hara, T. D., Mouchet, M., Delavenne, J., Leprieur, F., & Leroy, B. (2023). Global benthic biogeographical regions and macroecological drivers for ophiuroids. *Ecography*, *2023*(9), Article e06627. https://doi.org/10.1111/ecog.06627

Vilisics, F., Sólymos, P., & Hornung, E. (2007). A preliminary study on habitat features and associated terrestrial isopod species. In K. Tajovský, J. Schlaghamerský, & V. Pižl (Eds.), *Contributions to Soil Zoology in Central Europe II* (pp. 195–199). České Budějovice.

Wägele, J. W. (1990). Aspects of the evolution and biogeography of stygobiontic Isopoda (Crustacea: Peracarida). *Bijdragen tot de Dierkunde*, *60*(3/4), 145–150.

Wägele, J. W., Voelz, N. J., & McArthur, J. V. (1995). Older than the Atlantic Ocean: Discovery of a fresh-water *Microcerberus* (Isopoda) in North America and erection of *Coxicerberus*, new genus. *Journal of Crustacean Biology*, *15*(4), 733–745. https://doi.org/10.1163/193724095X00136

Wallace, A. R. (1876). *The geographical distribution of animals: With a study of the relations of living and extinct faunas as elucidating the past changes of the Earth's surface: In two volumes*. Macmillan and Co.

Wallerstein, B. R., & Brusca, R. C. (1982). Fish predation: A preliminary study of its role in the zoogeography and evolution of shallow water idoteid isopods (Crustacea: Isopoda: Idoteidae). *Journal of Biogeography*, *9*(2), 135–150. https://doi.org/10.2307/2844698

Warburg, M. R. (1993). The integument and moult. In *Evolutionary biology of land isopods* (p. 161). Springer, Berlin, Heidelberg. https://doi.org/10.1007/978-3-662-21889-1_2

Wares, J. P. (2001). Intraspecific variation and geographic isolation in *Idotea balthica* (Isopoda: Valvifera). *Journal of Crustacean Biology*, *21*(4), 1007–1013. https://doi.org/10.1163/20021975-99990193

Watling, L., Guinotte, J., Clark, M. R., & Smith, C. R. (2013). A proposed biogeography of the deep ocean floor. *Progress in Oceanography*, *111*, 91–112. https://doi.org/10.1016/j.pocean.2012.11.003

Watling, L., & Lapointe, A. (2022). Global biogeography of the lower bathyal (700–3000 m) as determined from the distributions of cnidarian anthozoans. *Deep Sea Research Part I: Oceanographic Research Papers*, *181*, Article 103703. https://doi.org/10.1016/j.dsr.2022.103703

Wheeler, Q. D. (2014). Are reports of the death of taxonomy an exaggeration? *New Phytologist*, *201*(2), 370–371. https://doi.org/10.1111/NPH.12612

Wieser, W. (1978). Consumer strategies of terrestrial gastropods and isopods. *Oecologia*, *36*, 191–201.

Williams, J. D., & Boyko, C. B. (2012). The global diversity of parasitic isopods associated with crustacean hosts (Isopoda: Bopyroidea and Cryptoniscoidea). *PloS ONE*, *7*(4), Article e35350. https://doi.org/10.1371/journal.pone.0035350

Williams, J. D., Boyko, C. B., & Marin, I. N. (2020). A new species and depth record of bopyrid (Crustacea, Isopoda) from a squat lobster in the Kuril-Kamchatka Trench. *European Journal of Taxonomy*, *724*, 122–133. https://doi.org/10.5852/EJT.2020.724.1165

Williams, J. D., Escalante, M., & Shanks, A. L. (2022). Identification and observations of parasitic isopod larvae (Isopoda: Epicaridea) from the northeastern Pacific: pelagic distribution and association with copepod intermediate hosts. *Journal of Crustacean Biology*, *42*, 1–14. https://doi.org/10.1093/jcbiol/ruac045

Wills, M. A. (1998). A phylogeny of recent and fossil Crustacea derived from morphological characters. In R. A. Fortey & R. H. Thomas (Eds.), *Arthropod Relationships, Systematics Association Special Volume Series 55* (pp. 189–209). Chapman & Hall, London, UK.

Wilson, G. D. F. (1991). Functional morphology and evolution of isopod genitalia. In R. T. Bauer & J. W. Martin (Eds.), *Crustacean sexual biology* (p. 355). Columbia University Press, New York.

Wilson, G. D. F. (1998). Historical influences on deep-sea isopod diversity in the Atlantic Ocean. *Deep Sea Research Part II: Topical Studies in Oceanography*, *45*(1–3), 279–301. https://doi.org/10.1016/S0967-0645(97)00046-5

Wilson, G. D. F. (2008a). Global diversity of isopod crustaceans (Crustacea; Isopoda) in freshwater. In *Freshwater Animal Diversity Assessment* (pp. 231–240). Springer. https://doi.org/10.1007/978-1-4020-8259-7_26

Wilson, G. D. F. (2008b). Gondwanan groundwater: Subterranean connections of Australian phreatoicidean isopods (Crustacea) to India and New Zealand. *Invertebrate Systematics*, *22*(2), 301–310. https://doi.org/10.1071/IS07030

Wilson, G. D. F. (2009). The phylogenetic position of the Isopoda in the Peracarida (Crustacea: Malacostraca). *Arthropod Systematics & Phylogeny*, *67*(2), 159–198.

Wilson, G. D. F., & Edgecombe, G. D. (2003). The Triassic isopod *Protamphisopus wianamattensis* (Chilton) and comparison with extant taxa (Crustacea, Phreatoicidea). *Journal of Paleontology*, *77*(3), 454–470. https://doi.org/10.1017/s0022336000044176

Wilson, G. D. F., & Morel, N. (2022). Isopod crustacean fossils from the Cenomanian stratotype: Five new species in suborders Cymothoida, Asellota and Valvifera. *Annales de Paléontologie*, *108*(1), Article 102538. https://doi.org/10.1016/j.annpal.2022.102538

Wilson, S. P., & Costello, M. J. (2005). Predicting future discoveries of European marine species by using a non-homogeneous renewal process. *Journal of the Royal Statistical Society: Series C*, *54*(5), 897–918.

WoRMS. (2018). *Isopoda*. Accessed on 2018-07-19 At: http://www.marinespecies.org/aphia.php?p=taxdetails&id=1131

WoRMS. (2023). *Isopoda*. Accessed on 2023-02-20 At: http://www.marinespecies.org/aphia.php?p=taxdetails&id=1131

Xu, L., Li, Y., Liu, Y., Mi, H., Jiang, X., Sun, Y., Zhao, H., Chen, D., & Wang, L. (2021). A comprehensive evaluation of the potential of semiterrestrial isopods, *Ligia exotica*, as a new animal food. *Scientific Reports*, *11*, Article 7213. https://doi.org/10.1038/s41598-021-86561-z

Yesudas, A., Jayachandran, P. R., Parameswaran, U. V., Vidyalakshmi, D., & Priyaja, P. (2021). Report on the association of valviferan isopod *Synidotea variegata* Collinge 1917 and regular sea urchin *Stomopneustes variolaris* Lamarck, 1816 from rocky subtidal regions of Vizhinjam, southwest coast of India. *Symbiosis*, *84*, 105–110. https://doi.org/10.1007/s13199-021-00759-3

Yli-Renko, M., Pettay, J. E., Rothäusler, E., & Vesakoski, O. (2022). Lack of anti-predator recognition in a marine isopod under the threat of an invasive predatory crab. *Biological Invasions*, *24*, 3189–3198. https://doi.org/10.1007/s10530-022-02839-x

Yu, J., An, J., Li, Y., & Boyko, C. B. (2018). The first complete mitochondrial genome of a parasitic isopod supports Epicaridea Latreille, 1825 as a suborder and reveals the less conservative genome of isopods. *Systematic Parasitology*, *95*, 465–478. https://doi.org/10.1007/s11230-018-9792-2

Yue, Y., Zhang, Q., & Wang, J. (2019). Integrated gas chromatograph-mass spectrometry (GC/MS) and MS/MS-based molecular networking reveals the analgesic and anti-inflammatory phenotypes of the sea slater *Ligia exotica*. *Marine Drugs*, *17*, Article 395. https://doi.org/10.3390/md17070395

Zaixso, H. E., Stoyanoff, P., & Gil, D. G. (2009). Detrimental effects of the isopod, *Edotia doellojuradoi*, on gill morphology and host condition of the mussel, *Mytilus edulis platensis*. *Marine Biology*, *156*, 2369–2378. https://doi.org/10.1007/s00227-009-1265-3

Zimina, O. L., Strelkova, N. A., & Lyubina, O. S. (2019). Species composition and peculiarities of the distribution of benthic Peracarida (Crustacea, Malacostraca) in the Barents Sea, based on surveys 2003–2008. *Biology Bulletin*, *46*(8), 864–885. https://doi.org/10.1134/S1062359019080181

Zimmer, M. (2001). Why do male terrestrial isopods (Isopoda: Oniscidea) not guard females? *Animal Behaviour*, *62*(4), 815–821. https://doi.org/10.1006/anbe.2001.1845

Zimmer, M. (2002). Nutrition in terrestrial isopods (Isopoda: Oniscidea): An evolutionary-ecological approach. *Biological Reviews*, *77*, 455–493. https://doi.org/10.1017/S1464793102005912

Zizka, A., Silvestro, D., Andermann, T., Azevedo, J., Duarte Ritter, C., Edler, D., Farooq, H., Herdean, A., Ariza, M., Scharn, R., Svanteson, S., Wengtrom, N., Zizka, V., & Antonelli, A. (2019). *CoordinateCleaner: standardized cleaning of occurrence records from biological collection databases*. Methods in Ecology and Evolution, 10(5):744-751. https://doi.org/0.1111/2041-210X.13152

Appendices

Appendix A – Chapter 3

Table A1. A list of the most prolific authors (who described each more than 100 isopod species – as first authors). Together they described 4,619 species, approximately 43% of the total.

First author	Species described				Publication lifetime (years)	Species/ year
	Total	Multi-authored	First	Last		
Verhoeff	421	4	1896	1967	72	6
Vandel	332	0	1924	1981	58	6
Budde-Lund	332	0	1880	1913	34	10
Bruce, N.L.	313	35	1978	2022	45	7
Barnard, K.H.	305	0	1914	1965	52	6
Menzies	302	121	1950	1983	34	9
Kensley	289	102	1971	2009	39	7
Nunomura	260	21	1973	2019	47	6
Richardson	250	0	1897	1913	17	15
Taiti	235	229	1979	2020	42	6
Poore	211	129	1975	2013	39	5
Arcangeli	187	0	1911	1960	50	4
Ferrara	178	156	1971	1996	26	7
Kussakin	165	64	1955	2001	47	4
Dollfus, A.	142	1	1884	1905	22	6
Birstein	137	4	1932	1972	41	3
Schmalfuss	127	46	1972	2016	45	3
Hansen	115	0	1890	1916	27	4
Nierstrasz	112	90	1915	1941	27	4
Müller, H.G.	105	3	1988	1995	8	13
Schultz	101	5	1963	1995	33	3

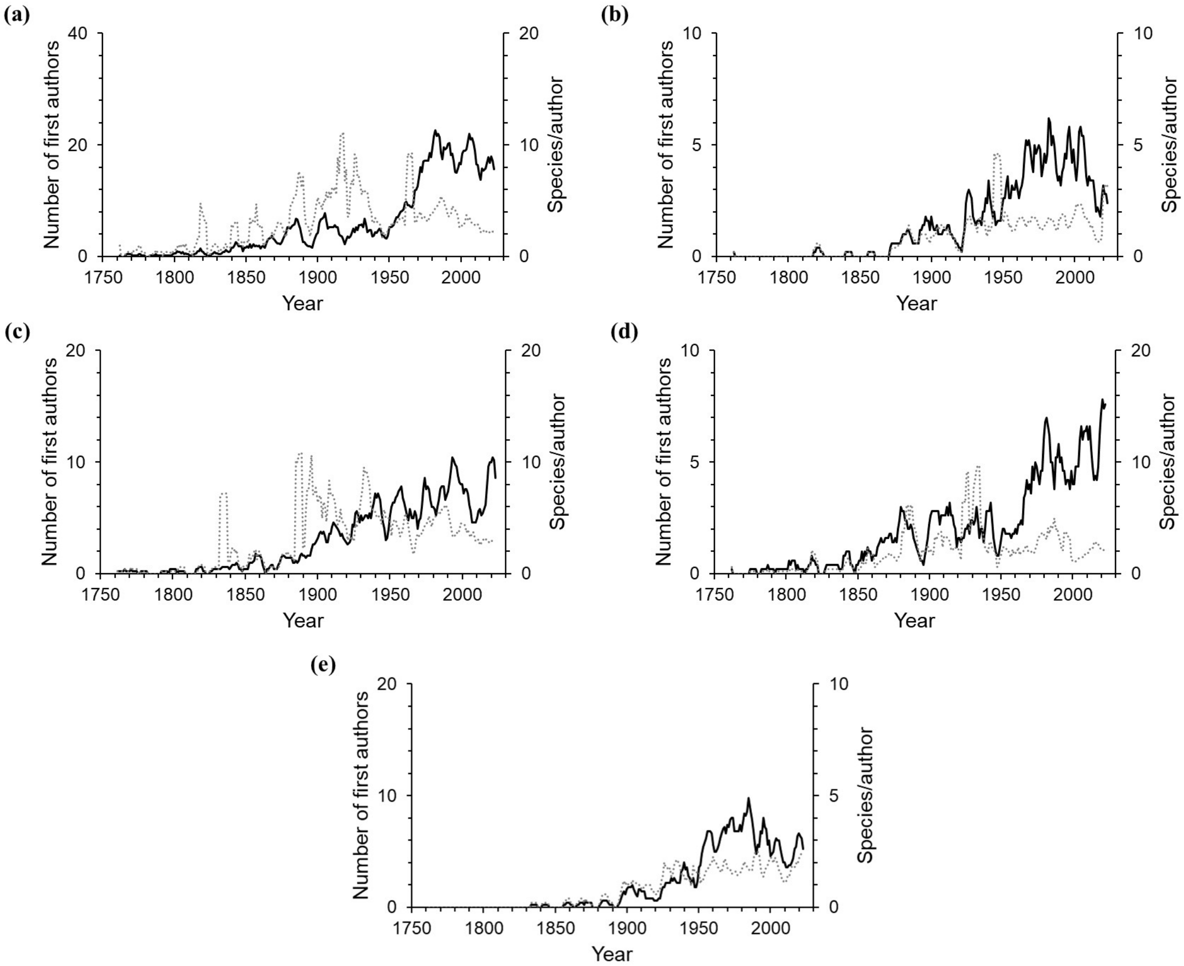

Figure A1. The number of first authors per year (solid line) and the average number of species described per author per year (dotted line) for the various subgroups. **(a)** marine, **(b)** freshwater, **(c)** terrestrial, **(d)** parasitic and **(e)** subterranean. The lines are 5-year moving averages. Note that the scales vary.

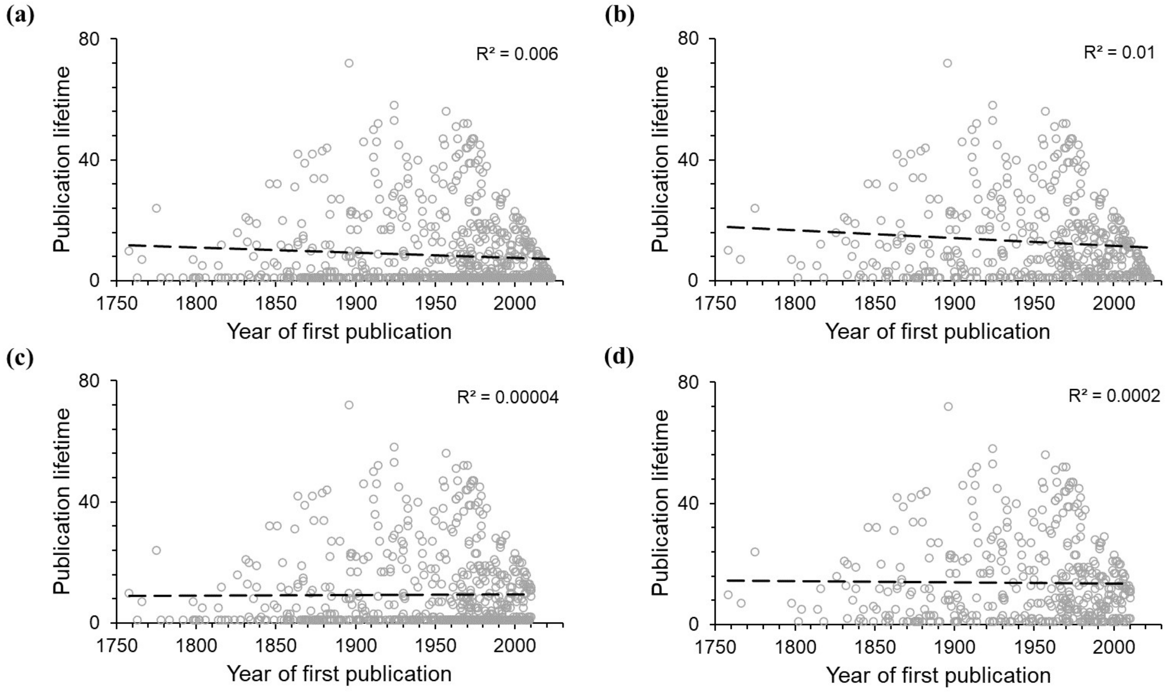

Figure A2. Linear regressions of authors' publication lifetimes against the year of the first publication (start of their publication lifetime). **(a)** for all first authors, **(b)** one-time authors excluded, **(c)** for all first authors, excluding the ones, who started publishing after 2010, **(d)** one-time authors and first authors, who started publishing after 2010, excluded.

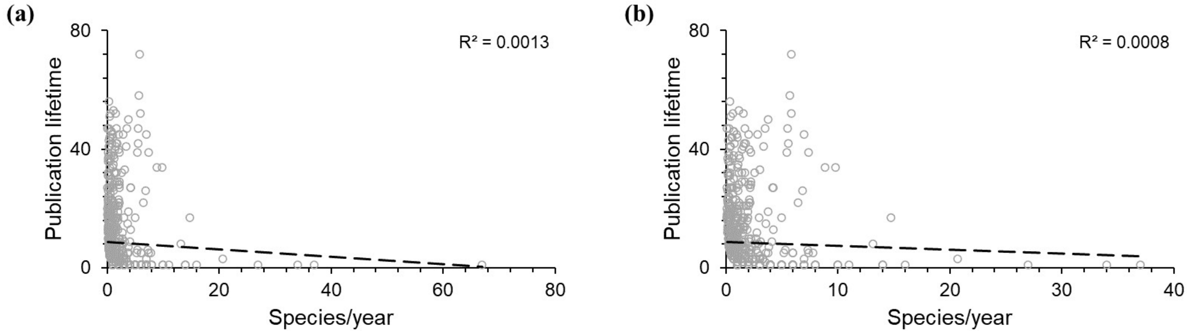

Figure A3. Linear regressions of publication lifetime against the average yearly number of species described by each author. **(a)** all first authors, **(b)** Vanhöffen, who described all 67 species in a single year, excluded.

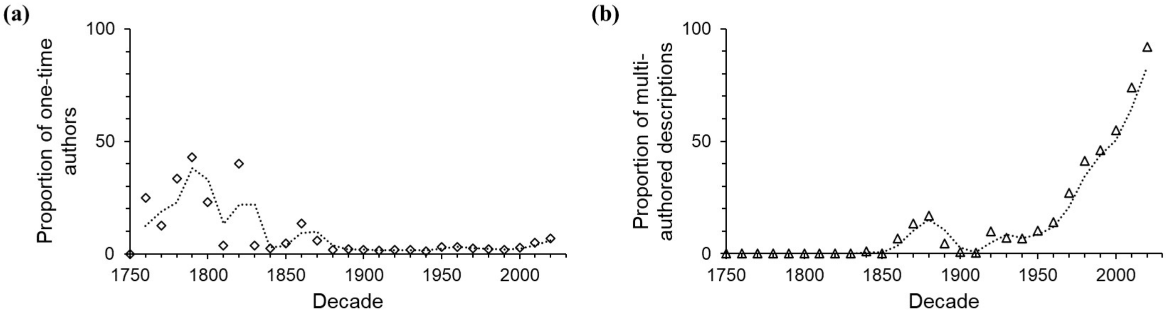

Figure A4. The percentage of contributions **(a)** by one-time authors, who described only a single species and **(b)** for multi-authored descriptions over time.

Table A2. A selection of 21 studies (selected by chance) which found and reported undescribed isopod species. Some studies described a few of the collected unnamed species right away, so the numbers given in the table below are the number of species that were left undescribed. Note that those species were undescribed at the time of publication of the respective study. It has not been checked whether any of the reported species have been formally described since and might now already be part of our dataset of globally described isopod species.

Study	Location	# collected but undescribed species
Brandt et al., 2007	Southern Ocean, deep sea	585
Poore et al., 1994	South-eastern Australian continental slope	318
Poore et al., 2015	Western and south-western Australian continental margin	118
Wilson, 2008	Gulf of Mexico, deep sea	60
Jennings et al., 2020 (genetic study)	Kuril-Kamchatka trench, deep sea	34
Guzik et al., 2019 (genetic study)	Australia, terrestrial	26
Golovan, 2018	Kuril Basin, deep sea	19
Kavanagh, 2009	Ireland, deep sea	15
Wetzer et al., 1997	Santa Maria Basin and Western Santa Barbara Channel	14
del Carmen Espinosa-Pérez & Hendrickx, 2001	Eastern Tropical Pacific	8
Schnurr et al., 2018 (genetic study)	Transition zone btw. North Atlantic and Nordic Seas	7

Table A2. Continued

Study	Location	# collected but undescribed species
Bunkley-Williams et al., 2006	Venezuela, marine	Potentially up to 4
Graening & Rogers, 2013	California, terrestrial	Potentially up to 4
Bluhm et al., 2005	Canada Basin, deep sea	3
Boyko & Williams, 2004	Bahamas, shallow marine waters	2
Merrin, 2006	Tasman Sea	2
Magrini et al., 2010	Brazil, terrestrial	1 or 2
Held, 2003 (genetic study)	Antarctic, shallow marine waters	1
Brockerhoff, 2004	New Zealand, intertidal	1
Xavier et al., 2012 (genetic study)	North-east Atlantic and Mediterranean	1
Golovan & Malyutina, 2022	Bering Sea, deep sea	1

References listed in Table A2

Bluhm, B. A., MacDonald, I. R., Debenham, C., & Iken, K. (2005). Macro- and megabenthic communities in the high Arctic Canada Basin: Initial findings. *Polar Biology*, *28*(3), 218–231.

Boyko, C. B., & Williams, J. D. (2004). New records of marine isopods (Crustacea: Peracarida) from the Bahamas, with descriptions of two new species of epicarideans. *Bulletin of Marine Science*, *74*(2), 353–383.

Brandt, A., Brix, S., Brökeland, W., Choudhury, M., Kaiser, S., & Malyutina, M. (2007). Deep-sea isopod biodiversity, abundance, and endemism in the Atlantic sector of the Southern Ocean — Results from the ANDEEP I–III expeditions. *Deep Sea Research Part II: Topical Studies in Oceanography*, *54*(16–17), 1760–1775. https://doi.org/10.1016/J.DSR2.2007.07.015

Brockerhoff, A. M. (2004). Occurrence of the internal parasite Portunion sp. (Isopoda: Entoniscidae) and its effect on reproduction in intertidal crabs (Decapoda: Grapsidae) from New Zealand. *Journal of Parasitology*, *90*(6), 1338–1344.

Bunkley-Williams, L., Williams, E. H., & Bashirullah, A. K. (2006). Isopods (Isopoda: Aegidae, Cymothoidae, Gnathiidae) associated with Venezuelan marine fishes (Elasmobranchii, Actinopterygii). *Revista de Biología Tropical*, *54*, 175–188.

del Carmen Espinosa-Pérez, M., & Hendrickx, M. E. (2001). Checklist of isopods (Crustacea: Peracarida: Isopoda) from the eastern tropical Pacific. *Belgian Journal of Zoology*, *131*(1), 43–56.

Golovan, O. A. (2018). Desmosomatidae (Isopoda: Asellota) from the Kuril Basin of the Sea of Okhotsk: First data on diversity with the description of the dominant species Mirabilicoxa biramosa sp. nov. *Deep Sea Research Part II: Topical Studies in Oceanography*, *154*, 292–307. https://doi.org/10.1016/J.DSR2.2018.01.008

Golovan, O. A., & Malyutina, M. (2022). The first record of the family Paramunnidae (Isopoda: Asellota) from the bathyal of the Bering Sea with descriptions of two new species of Munnogonium. *Deep Sea Research Part II: Topical Studies in Oceanography*, *105095*.

Graening, G., & Rogers, D. C. (2013). Checklist of inland aquatic Isopoda (Crustacea: Malacostraca) of California. *California Fish and Game*, *99*(4), 176–192.

Guzik, M. T., Stringer, D. N., Murphy, N. P., Cooper, S. J. B., Taiti, S., King, R. A., Humphreys, W. F., & Austin, A. D. (2019). Molecular phylogenetic analysis of Australian arid-zone oniscidean isopods (Crustacea: Haloniscus) reveals strong regional endemicity and new putative species. *Invertebrate Systematics*, *33*(3), 556–574. https://doi.org/10.1071/IS18070

Held, C. (2003). Molecular evidence for cryptic speciation within the widespread Antarctic crustacean Ceratoserolis trilobitoides (Crustacea, Isopoda). In *Antarctic Biology in a Global Context*. Backhuys Publishers.

Jennings, R. M., Golovan, O. A., & Brix, S. (2020). Integrative species delimitation of desmosomatid and nannoniscid isopods from the Kuril-Kamchatka trench, with description of a hadal species. *Progress in Oceanography*, *182*, 102236. https://doi.org/10.1016/J.POCEAN.2019.102236

Kavanagh, F. A. (2009). A catalogue of the Asellota (Crustacea: Isopoda) off the west coast of Ireland and Britain, from 100–5000 m. *Bulletin of the Irish Biogeographical Society*, *33*, 14–75.

Magrini, M. J., Araujo, P. B., & Uehara-Prado, M. (2010). Crustacea, Isopoda, Oniscidea Latreille, 1802: New continent record and distribution extension in Brazil. *Check List*, *6*(2), 217–219.

Merrin, K. L. (2006). The first record of the crustacean isopod genus Pseudarachna Sars, 1897 (Isopoda: Asellota: Munnopsidae) from the Southern Hemisphere, with description of a new species from New Zealand. *Zootaxa*, *1370*, 59–68. https://doi.org/10.11646/zootaxa.1370.1.5

Poore, G. C. B., Avery, L., Błażewicz-Paszkowycz, M., Browne, J., Bruce, N. L., Gerken, S., Glasby, C. J., Greaves, E., McCallum, A. W., Staples, D., Syme, A., Taylor, J., Walker-Smith, G., Warne, M., Watson, C., Williams, A., Wilson, R. S., & Woolley, S. (2015). Invertebrate diversity of the unexplored marine western margin of Australia: Taxonomy and implications for global biodiversity. *Marine Biodiversity*, *45*(2), 271–286. https://doi.org/10.1007/S12526-014-0255-Y/FIGURES/5

Poore, G. C. B., Just, J., & Cohen, B. F. (1994). Composition and diversity of Crustacea Isopoda of the southeastern Australian continental slope. *Deep Sea Research Part I: Oceanographic Research Papers*, *41*(4), 677–693. https://doi.org/10.1016/0967-0637(94)90049-3

Schnurr, S., Osborn, K. J., Malyutina, M., Jennings, R. M., Brix, S., Driskell, A., Svavarsson, J., & Martinez Arbizu, P. (2018). Hidden diversity in two species complexes of munnopsid isopods (Crustacea) at the transition between the northernmost North Atlantic and the Nordic Seas. *Marine Biodiversity*, *48*(2), 813–843. https://doi.org/10.1007/S12526-018-0877-6

Wetzer, R., Brusca, R. C., & Wilson, G. D. F. (1997). The Crustacea Part 2 - The Isopoda, Cumacea and Tanaidacea. In J. A. Blake & P. H. Scott (Eds.), *Taxonomic Atlas of the Benthic Fauna of the Santa Maria Basin and Western Santa Barbara Channel* (Vol. 11). Santa Barbara Museum of Natural History.

Wilson, G. D. F. (2008). Local and regional species diversity of benthic Isopoda (Crustacea) in the deep Gulf of Mexico. *Deep Sea Research Part II: Topical Studies in Oceanography*, *55*(24–26), 2634–2649.

Xavier, R., Santos, A. M., Harris, D. J., Sezgin, M., Machado, M., & Branco, M. (2012). Phylogenetic analysis of the north-east Atlantic and Mediterranean species of the genus Stenosoma (Isopoda, Valvifera, Idoteidae). *Zoologica Scripta*, *41*(4), 386–399.

Appendix B – Chapter 4

Table B1. List of datasets and associated digital object identifiers (DOI) used in this study. Data were downloaded from the Global Biodiversity Information Facility (GBIF) and the Ocean Biodiversity Information System (OBIS). The dataset names are listed as provided by the metadata from the GBIF and OBIS data downloads and are ranked by the number of records included in this study (for each source separately).

Dataset name	DOI	Source	# Records
iNaturalist Research-grade Observations	10.15468/ab3s5x	GBIF	63,931
Isopoda (Interim dataset)	10.15468/kyu8ob	GBIF	51,724
Dutch Foundation for Applied Water Research (STOWA) - Limnodata Neerlandica	10.15468/ennulm	GBIF	15,162
NMNH Extant Specimen Records (USNM, US)	10.15468/hnhrg3	GBIF	10,431
Environmental Monitoring database (MOD) DNV	10.15468/q8qykg	GBIF	9,669
Estonian Naturalists' Society	10.15468/bmk3ab	GBIF	7,878
Artportalen (Swedish Species Observation System)	10.15468/kllkyl	GBIF	7,724
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Franck NOEL	10.15468/8myaks	GBIF	7,678
Azorean Biodiversity Portal	10.15468/j0iceo	GBIF	6,108
Inventory of the terrestrial isopods in Belgium (2011-2020)	10.15468/mw9c66	GBIF	6,078
The Danish Environmental Portal, species and habitats-database "Danmarks Miljøportals Naturdatabase"	10.15468/ku2f82	GBIF	4,673
Collection Crustacea SMF	10.15468/mc7ysi	GBIF	4,229
SOUTHERN OCEAN ISOPODA	10.15468/laitod	GBIF	3,413
Limnodata	10.15468/3bqngy	GBIF	2,953
Museums Victoria provider for OZCAM	10.15468/lp1ctu	GBIF	2,936
Inventaire des Crustacés Isopodes terrestres de France métropolitaine - Inventaire des Crustacés Isopodes de France métropolitaine	10.15468/xqesji	GBIF	2,776
Verified marine records from Indicia-based surveys	10.15468/yfyeyg	GBIF	2,619
Welsh Invertebrate Database (WID)	10.15468/bv8fcj	GBIF	2,516
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes d'Emmanuel SECHET	10.15468/v1y0bl	GBIF	2,504
Marine Nature Conservation Review (MNCR) and associated benthic marine data held and managed by JNCC	10.15468/kcx3ca	GBIF	2,372
Biofokus	10.15468/jxbhqx	GBIF	2,371
National Benthic Inventory	10.15468/1dg6fe	GBIF	2,213

Table B1. Continued

Dataset name	DOI	Source	# Records
Suffolk Biodiversity Information Service (SBIS) Dataset	10.15468/ab4vwo	GBIF	2,103
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de CERCOPE (Jean-Louis PRATZ)	10.15468/ba63zj	GBIF	2,100
Leicestershire and Rutland Environmental Records Centre records pre 2000	10.15468/res3cx	GBIF	2,007
Australian Museum provider for OZCAM	10.15468/e7susi	GBIF	1,958
International Barcode of Life project (iBOL)	10.15468/inygc6	GBIF	1,912
(Table A-2.1 to A-2.7) Abundance of macrobenthos in surface sediments in the Arctic Ocean	10.1594/pangaea.734952	GBIF	1,824
Observation.org, Nature data from around the World	10.15468/5nilie	GBIF	1,777
naturgucker	10.15468/uc1apo	GBIF	1,545
Canadian Museum of Nature Crustacea Collection	10.15468/thiry8	GBIF	1,514
imr_mareano_rpsledge	10.15468/gecvl4	GBIF	1,510
Norwegian Biodiversity Information Centre - Other datasets	10.15468/tm56sc	GBIF	1,494
National Trust Species Records	10.15468/opc6g1	GBIF	1,416
FBIP:IZIKO-UCT:Historical Invertebrates (1930-1980)	10.15468/fk7bhk	GBIF	1,372
Norwegian Species Observation Service	10.15468/zjbzel	GBIF	1,342
Ifremer BIOCEAN database (Deep Sea Benthic Fauna)	10.15468/yxphxa	GBIF	1,292
mabik_cr	10.15468/gmn4sd	GBIF	1,231
Natural England Marine Monitoring surveys	10.15468/ysikg5	GBIF	1,145
BRERC species records from all years at full resolution excluding Notable Species within the last 10 years	10.15468/h1ln5p	GBIF	1,083
Queensland Museum provider for OZCAM	10.15468/lotsye	GBIF	1,081
Monitoring data from the federal state North Rhine-Westphalia (Germany)	10.13148/bfe91	GBIF	1,057
CLICNAT Base de données naturaliste picarde gérée par Picardie Nature	10.15468/pmodx6	GBIF	1,054
Marine benthic dataset (version 1) commissioned by UKOOA	10.15468/tqmrtv	GBIF	958
Biologiezentrum Linz	10.15468/ynjblx	GBIF	935
Données d'occurrences Espèces issues de l'inventaire des ZNIEFF	10.15468/ikshke	GBIF	897
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE293	10.1594/pangaea.745682	GBIF	861

Table B1. Continued

Dataset name	DOI	Source	# Records
Bibliographie de la faune, la flore et la fonge de France métropolitaine et outre-mer - Bulletin du Muséum d'Histoire Naturelle [1895-]	10.15468/d6eace	GBIF	837
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE206	10.1594/pangaea.745671	GBIF	834
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE275	10.1594/pangaea.745680	GBIF	831
INSDC Sequences	10.15468/sbmztx	GBIF	830
Collection Crustacea - ZMB	10.15468/fwghff	GBIF	820
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE272	10.1594/pangaea.745679	GBIF	816
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE278	10.1594/pangaea.745681	GBIF	816
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE241	10.1594/pangaea.745675	GBIF	813
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE255	10.1594/pangaea.745676	GBIF	813
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE258	10.1594/pangaea.745677	GBIF	813
All taxa records for Leicestershire and Rutland	10.15468/i46are	GBIF	789
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE262	10.1594/pangaea.745678	GBIF	786
DASSH Data Archive Centre - Statutory Surveys	10.15468/ytlexw	GBIF	749
(Table 2) Benthic isopod species of the Ross Sea, their bathymetry and distribution in the Southern Ocean	10.15468/5va88s	GBIF	728
Monitoring data from the federal state Hesse (Germany)	10.13148/bfe87	GBIF	728
imr_mareano_grab	10.15468/dlaxsw	GBIF	724
Invertebrate Zoology Division, Yale Peabody Museum	10.15468/0lkr3w	GBIF	715
Department of Agriculture Environment and Rural Affairs (DAERA) Marine and Fisheries Division Marine Survey Data	10.15468/zqaqwm	GBIF	704
Museum of Comparative Zoology, Harvard University	10.15468/p5rupv	GBIF	660
REBENT - Réseau national de surveillance des biocénoses benthiques côtières	10.15468/h74fvv	GBIF	632
Species data for Scottish waters held and managed by Scottish Natural Heritage, derived from benthic surveys 1993 to 2018	10.15468/faxvgd	GBIF	624
NIWA Invertebrate Collection	10.15468/6qgswo	GBIF	620

Table B1. Continued

Dataset name	DOI	Source	# Records
Programme d'observation naturaliste participative avec l'application mobile INPN Espèces - Application INPN Espèces: Observations naturalistes, participatives et opportunistes, fondées sur des photographies	10.15468/2wgzwe	GBIF	617
Invertebrate Site Register - England (1738-2005)	10.15468/7wbiu7	GBIF	599
Invertebrata varia (Luomus)	10.15468/h37nwv	GBIF	591
Marine data from Natural Resources Wales (NRW) Technical Support (Research & Monitoring) Contracts, Wales	10.15468/az7nw3	GBIF	578
Fife Nature Records Centre combined dataset	10.15468/ccc1ip	GBIF	576
KiEco Freshwater Ecology: River Macroinvertebrates	10.15468/qggn2f	GBIF	536
Crustacea collection (TSZCr) The Arctic University Museum of Norway	10.15468/y4kj3p	GBIF	531
imr_mareano_beamtrawl	10.15468/iomgfj	GBIF	508
Marine Records from Pembrokeshire Marine Species Atlas	10.15468/42yudm	GBIF	502
NBIS Records to December 2016	10.15468/jca5lo	GBIF	491
Marine invertebrate collection NTNU University Museum	10.15468/ddbs14	GBIF	487
Invertebrates excl. Entomology at the Natural History Museum of Denmark	10.15468/wodhis	GBIF	443
SA Fauna (BDBSA)	10.15468/xsderl	GBIF	438
Crustacea in surface sediments off Sylt collected during HEINCKE cruise HE218	10.1594/pangaea.745672	GBIF	414
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du/de la naturaliste Bottinelli Julien provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/qvzekx	GBIF	411
A national macroinvertebrate dataset collected for the biomonitoring of Ireland’s river network, 2007–2018 (EPA)	10.15468/c63xzc	GBIF	409
Edaphobase	10.15468/rk9xc7	GBIF	402
Diversidad bentónica del ambiente intermareal e infralitoral somero de Progreso, Yucatán	10.15468/k3kbuk	GBIF	397
HBRG Other Invertebrates Dataset	10.15468/r780rb	GBIF	394
Marine Invertebrate voucher specimens at the Florida Biodiversity Collection, Florida Fish and Wildlife Conservation Commission	10.15468/bfd6ci	GBIF	391
Tasmanian Museum and Art Gallery provider for OZCAM-Arthropoda	10.15468/f8ranh	GBIF	383
SEWBReC Myriapods, Isopods, and allied species (South East Wales)	10.15468/rvxsqs	GBIF	381

Table B1. Continued

Dataset name	DOI	Source	# Records
The crustaceans collection (IU) of the Muséum national d'Histoire naturelle (MNHN - Paris)	10.15468/qgvvhd	GBIF	375
CAS Invertebrate Zoology (IZ)	10.15468/tiac99	GBIF	371
NHMD Invertebrate Zoology Collection	10.15468/nuz79n	GBIF	368
NINA Vanndata øvrige arter	10.15468/s6zhid	GBIF	358
Argyll Biological Records Dataset	10.15468/ejve6c	GBIF	349
SHARK - Regional monitoring, recipient control and monitoring projects of zoobenthos in Sweden since 1972	10.15468/cesssx	GBIF	343
UF Invertebrate Zoology	10.15468/sm6qo6	GBIF	331
Royal BC Museum - Invertebrates Collection	10.5886/zh7n1e	GBIF	318
Natural History Museum (London) Collection Specimens	10.5519/0002965	GBIF	316
Merseyside BioBank (unverified)	10.15468/iou2ld	GBIF	313
Tasmanian Museum and Art Gallery provider for OZCAM	10.15468/ijp8p9	GBIF	301
Monitoring data from the federal state Bavaria (Germany)	10.13148/bfe88	GBIF	297
Gloucestershire Historic Wildlife Sightings prior to 1st Jan 2000	10.15468/dgf5es	GBIF	292
Marine Offshore Seabed Survey data held by JNCC	10.15468/skvdld	GBIF	282
Déterminations et observations du Forum "Le Monde des Insectes" (LMDI) - Données photographiques validées de la galerie du forum "Le Monde des Insectes"	10.15468/2vjhg8	GBIF	269
Porcupine Marine Natural History Society Dataset	10.15468/pcmg9q	GBIF	269
NRW Regional Data: South East Wales Non-sensitive species	10.15468/g7xxs8	GBIF	268
Coleção de Crustacea do Museu Nacional (MNRJ - CARCINO)	10.15468/zv0lvr	GBIF	263
Lund Museum of Zoology (MZLU)	10.15468/mw39rb	GBIF	260
Monitoring data from the federal state Saxony-Anhalt (Germany)	10.13148/bfe94	GBIF	255
Leicestershire and Rutland Environmental Records Centre records 2010-2014	10.15468/9n92x3	GBIF	248
Manscape	10.13148/6hacwh	GBIF	248
Northern Ireland Environment Agency (NIEA) Collated Species Records	10.15468/gefbqb	GBIF	244
SIO Benthic Invertebrate Collection	10.15468/4w9oc7	GBIF	244
Adenda a la colección de referencia de invertebrados de la Estación Mazatlán, UNAM y análisis de la fauna de crustáceos isópodos del Pacífico mexicano, julio 1996-julio 1997	10.15468/wzmgns	GBIF	237

Table B1. Continued

Dataset name	DOI	Source	# Records
Estonian University of Life Sciences Institute of Agricultural and Environmental Sciences Entomological Collection	10.15468/qn6223	GBIF	235
Monitoring data from the federal state Saxony (Germany)	10.13148/bfe93	GBIF	234
La Planète revisitée - Corse 2019-2022 - La Planète Revisitée Corse 2020: prospections Agriate, Saint-Florent et Cap Corse	10.15468/bwhm6a	GBIF	232
DEMNA-DNE: Occurrences of benthic macroinvertebrates in running waters of Wallonia, Belgium	10.15468/nnzqm5	GBIF	229
Programme CARTHAM: Inventaire biologique dans le cadre de Natura 2000 en Mer	10.15468/3isrct	GBIF	226
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de DESMOTS Didier	10.15468/r22czs	GBIF	212
TWIC Biodiversity Field Trip Data (1995-present)	10.15468/ljc0ke	GBIF	210
Collections and observation data National Museum of Natural History Luxembourg	10.15468/s2iu7d	GBIF	208
SHARK - National zoobenthos monitoring in Sweden since 1971	10.15468/fggzdr	GBIF	208
Northern Territory Museum and Art Gallery provider for OZCAM	10.15468/giro3a	GBIF	207
Collection Crustacea - SNSD	10.15468/zjvt2g	GBIF	206
Gwaii Haanas Invertebrates (OBIS Canada)	10.15468/9o6jbj	GBIF	205
LERN Records	10.15468/esxc9a	GBIF	201
DASSH Data Archive Centre Academic Surveys	10.15468/cwqszy	GBIF	199
Monitoring data from the federal state Rhineland-Palatinate (Germany)	10.13148/bfe92	GBIF	199
Museo Argentino de Ciencias Naturales "Bernardino Rivadavia" (MACN). Invertebrates National Collection (MACNIn)	10.15468/uuz636	GBIF	196
NRW Regional Data: all taxa (excluding sensitive species), West Wales	10.15468/q3d1hl	GBIF	196
ZFMK Crustacea collection	10.15468/s3fsbt	GBIF	191
Invertebrate Common Standards Monitoring and ISIS Test Data	10.15468/epw9ym	GBIF	189
Alien macroinvertebrates in Flanders, Belgium	10.15468/xjtfoo	GBIF	183
Marine Intertidal Phase 1 Species Dataset from the Countryside Council for Wales 1996-2005	10.15468/kflo7m	GBIF	179
Auckland Museum NZ Marine Collection	10.15468/plyefd	GBIF	174
DFO Quebec Region MLI museum collection	10.15468/tvrkn7	GBIF	164

Table B1. Continued

Dataset name	DOI	Source	# Records
Royal Belgian Institute of Natural Sciences Crustacea collection	10.15468/xtppux	GBIF	163
Earth Guardians Weekly Feed	10.15468/slqqt8	GBIF	160
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Monsieur LEFEBVRE François	10.15468/aylit9	GBIF	157
Bob Merritt invertebrate records for VCs 72-75	10.15468/a35rnz	GBIF	156
Invertebrate records from sites that are mainly across Scotland	10.15468/aaxvmc	GBIF	151
NMNH Material Samples (USNM)	10.15468/jb9tdf	GBIF	149
Alterra (NL) - Comparison of entomofauna in four different habitats	10.15468/mkoqqh	GBIF	148
Natural History Museum Rotterdam - Specimens	10.15468/kwqaay	GBIF	147
Abundance of benthic infauna in surface sediments from the North Sea sampled during two Michael Sars cruises in 2000	10.1594/pangaea.756785	GBIF	144
The Environmental Sample Collection of the Arthropoda Varia Section at the Zoologische Staatssammlung München	10.15468/biecak	GBIF	144
Lajitietokeskus/FinBIF - Notebook, general observations	10.15468/4g56tp	GBIF	141
South Australian Museum Adelaide provider for OZCAM	10.15468/wz4rrh	GBIF	141
Queen Victoria Museum Art Gallery provider for OZCAM	10.15468/tedfxn	GBIF	137
Yorkshire Wildlife Trust - Non-sensitive records from all taxonomic groups	10.15468/2razk5	GBIF	136
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Pierre FRAPA	10.15468/izrly0	GBIF	134
Biological data from the Soviet Antarctic Expedition (1955-1958)	10.15468/yr2tlh	GBIF	133
Arctic Ocean Diversity	10.15468/xrzanm	GBIF	131
Observations of three Idotea species (I. balthica, I. chelipes and I. granulosa) in Northern Europe, including the Baltic Sea - data derived from museum collections	10.14284/7	GBIF	130
Visiolittoral: conservatoire du littoral - Visiolittoral: surveillance de sites du littoral	10.15468/e8x2w5	GBIF	129

Table B1. Continued

Dataset name	DOI	Source	# Records
Abundance of benthic infauna in surface sediments from the North Sea sampled during cruise Cirolana00/5	10.1594/pangaea.756782	GBIF	128
Seasearch Marine Surveys in England	10.15468/kywx6m	GBIF	128
Miscellaneous records held by BIS	10.15468/mo7peo	GBIF	126
SER Species-based Surveys	10.15468/q8qen3	GBIF	126
Tasmanian Natural Values Atlas	10.15468/rtnb4m	GBIF	125
Monitoring data from the federal state Mecklenburg-Western Pomerania (Germany)	10.13148/bfe90	GBIF	124
Shropshire Ecological Data Network database	10.15468/5v5pvk	GBIF	124
Leicestershire and Rutland Environmental Records Centre records 2015-2019	10.15468/dues93	GBIF	123
Marine biological observation data from coastal and offshore surveys around New Zealand	10.15468/pzpgop	GBIF	123
n_niek_2021	10.15468/d7f5vk	GBIF	122
Abundance of megabenthic species in trawl catches per station in addition to table 2 during POLARSTERN cruise ARK-VIII/2 (EPOS)	10.1594/pangaea.815750	GBIF	120
Natural Resources Wales Regional Data: Mid-Wales	10.15468/whj6d7	GBIF	120
RBINS DaRWIN	10.15468/qxy4mc	GBIF	119
Commissioned surveys and staff surveys and reports for Scottish Wildlife Trust reserves - Verified data	10.15468/a6snhl	GBIF	117
Formación de una base de datos de la biodiversidad de fauna marina y costera en el Golfo de California	10.15468/t9mitt	GBIF	117
Marine sites, habitats and species data collected during the BioMar survey of Ireland.	10.15468/nwlt7a	GBIF	116
Centre for Biodiversity Genomics - Canadian Specimens	10.15468/mbwnw9	GBIF	113
Collection Crustacea - ZIM Hamburg	10.15468/zxrapv	GBIF	113
Dr Mary Gillham Archive Project	10.15468/ajv47f	GBIF	113
Species recordings from the Danish National portal Arter.dk	10.15468/q3yy4u	GBIF	112
UAM Invertebrate Collection (Arctos)	10.15468/wrvy1y	GBIF	112
Bernice P. Bishop Museum	10.15468/s6ctus	GBIF	107
Marine Invertebrata specimen database of Osaka Museum of Natutal History	10.15468/zhubgk	GBIF	105
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Monsieur BECHEAU Félix	10.15468/fgzzs7	GBIF	104
Biodiversity Research and Teaching Collections - TCWC Marine Invertebrates	10.15468/dfrwoh	GBIF	103

Table B1. Continued

Dataset name	DOI	Source	# Records
A glimpse into a remarkable unknown diversity of oniscideans along the Caribbean coasts revealed on a tiny island	10.15468/78vddj	GBIF	102
Ireland's BioBlitz	10.15468/aiiz3z	GBIF	102
Naturalis Biodiversity Center (NL) - Crustacea	10.15468/vjoltu	GBIF	102
Structures and Nutrition Requirements of Macrozoobenthic Communities in the area of the Lomonossov Ridge, 1995-1998	10.15468/rz66mz	GBIF	99
Intertidal Biodiversity along the Portuguese coast (2001-2002)	10.15468/mbg5p3	GBIF	98
Abundance of benthic infauna in surface sediments from the North Sea sampled during HEINCKE cruise HE133	10.1594/pangaea.756768	GBIF	97
Scottish river macro-invertebrate records from 2007 collected by SEPA	10.15468/l82tvb	GBIF	97
Bringing Reedbeds to Life Invertebrate Survey of three key reedbed sites in England in 2009, 2010	10.15468/919amh	GBIF	94
Kent Wildlife Trust Shoresearch Intertidal Survey 2004 onwards	10.15468/zyxxue	GBIF	92
Programme d’acquisition et de valorisation de données naturalistes BioObs - Observations naturalistes des Amis de BioObs.	10.15468/ldch7a	GBIF	92
1778-1998 Ivor Rees North Wales Marine Fauna Ad-hoc sightings shore and ship-based surveys	10.17031/35prlf	GBIF	91
Invertebrados del Parque Nacional Arrecife Puerto Morelos	10.15468/gbg82q	GBIF	86
CEDaR Online Recording	10.15468/ke4kw8	GBIF	84
Miscellaneous records held on the Cofnod database	10.15468/hcgqsi	GBIF	84
Riverfly Census - Aquatic invertebrate species occurrence, for the calculation of pressure biometric scores in English and Welsh rivers covering the period 2015 - 2018	10.15468/dz1qvj	GBIF	84
CLICNAT- Base de données naturaliste picarde - Données de terrain du CPIE des Pays de l'Aisne	10.15468/jjazaz	GBIF	82
Merseyside BioBank Active Naturalists (unverified)	10.15468/smzyqf	GBIF	81
Species lists for benthic communities of Norwegian fjords from environmental surveys (data used in Sen et al. Estuarine, Coastal and Shelf Science 2022)	10.15468/nvcpqd	GBIF	80
Waarnemingen.be - Non-native animal occurrences in Flanders and the Brussels Capital Region, Belgium	10.15468/k2aiak	GBIF	80

Table B1. Continued

Dataset name	DOI	Source	# Records
Bibliographie de la faune, la flore et la fonge de France métropolitaine et outre-mer - Bulletin mensuel de la société linnéenne de Lyon [1932-]	10.15468/riwjrl	GBIF	79
FBIP:IZIKO-UCT:Historical Survey (1930-1980)	10.15468/zmnk0m	GBIF	79
Réserves Naturelles de France (RNF) - RNF - Données de l'association Vivarmor Nature	10.15468/aqixrm	GBIF	79
University of Amsterdam (NL) – Benthos monitoring of the North Sea research database	10.15468/smncop	GBIF	78
BIOMAERL.Maerl Biodiversity.Functional Structure And Antropogenic Impacts (1996-1998).	10.25607/zp0vwl	GBIF	76
Rotherham Biological Records Centre - Non-sensitive Records from all taxonomic groups	10.15468/d3tufo	GBIF	76
niek_2022	10.15468/9axxge	GBIF	75
New species and records of terrestrial Isopoda (Crustacea, Oniscidea) from Socotra Island, Yemen	10.3897/zookeys.31.140	GBIF	74
Natural history museum data on Canadian Arctic marine benthos	10.5886/nb9hje	GBIF	73
Données naturalistes de ROMET Nicolas	10.15468/qtt1ke	GBIF	72
SILENE-FAUNE-PACA - Parc_National_de_Port_Cros_2017_12_18	10.15468/oatraz	GBIF	72
Ulster Museum Marine Surveys	10.15468/bj5xdk	GBIF	71
River macroinvertebrate data for 2005 and 2006	10.15468/knxcqi	GBIF	70
Computarización de material complementario en la Colección Regional de Invertebrados del Pacífico mexicano, Instituto de Ciencias del Mar y Limnología, Unidad Académica Mazatlán, UNAM	10.15468/yl5jhd	GBIF	69
Données Faune Base SIRFF - FNE Centre-Val de Loire - Système d'Information Régional sur la Faune et la Flore - FNE Centre-Val de Loire - Données 2016	10.15468/pobntk	GBIF	69
Leicestershire and Rutland Environmental Records Centre records 2000-2009	10.15468/cs2zzf	GBIF	66
Natural History Collections of the Faculty of Biology AMU	10.15468/54hgbz	GBIF	66
Zoobenthos data from the Southern Beaufort Sea, 1971-1975	10.15468/hrvlij	GBIF	66
Irish Lagoon Surveys 2016 - 2017	10.15468/2y63zg	GBIF	65
Hypogean macro-Crustacea records	10.15468/jzjzcr	GBIF	64
Invertebrates (except insects), Outer Hebrides	10.15468/hpavud	GBIF	63
Terrestrial isopods of the genus Porcellio Latreille, 1804 (Isopoda; Oniscidea) in Iran, with a description of a new species	10.11646/zootaxa.4311.1.9	GBIF	63
Agri-Food and Biosciences Institute Marine Surveys	10.15468/d2mwmv	GBIF	61
IndOBIS, Indian Ocean Node of OBIS	10.15468/tbedgi	GBIF	61

Table B1. Continued

Dataset name	DOI	Source	# Records
The Arthropoda Varia Collection at the Zoologische Staatssammlung München	10.15468/hrzzrc	GBIF	60
The First Comprehensive Description of the Biodiversity and Biogeography of Antarctic and Sub-Antarctic Intertidal Communities	10.15468/doyfzk	GBIF	60
Biodiversidad de la macrofauna bentónica en la microcuenca de Chabihau, Yucatán	10.15468/jnkwlg	GBIF	58
1915-2016 Department for Environment Food & Rural Affairs (Defra), Marine Strategy Framework Directive (MSFD) Collation of invasive non-indigenous species	10.17031/f0vfo3	GBIF	56
Abundance of benthic infauna in surface sediments from the North Sea sampled during cruise DeHolland1986	10.1594/pangaea.757633	GBIF	56
Invertebrates (Type Specimens) of the Swedish Museum of Natural History	10.15468/uadgyw	GBIF	56
Limnic freshwater benthic invertebrates biogeographical mapping/inventory NTNU University Museum	10.15468/k1pumk	GBIF	55
NaGISA Project	10.15468/3gdwj2	GBIF	55
AAD Benthic Sampling Database	10.15468/j075qn	GBIF	54
Inventaire des invertébrés marins benthiques et des algues de la Guadeloupe: expédition Karubenthos 2012 - Expédition Karubenthos 2012: inventaire des Mollusques et des Crustacés de la Guadeloupe	10.15468/zxgvvp	GBIF	54
Marine Nature Conservation Review (MNCR) and associated benthic marine data held and managed by English Nature	10.15468/2vttzr	GBIF	53
Système d’évaluation de l’état des Eaux (SEEE) - Données hydrobiologiques sur l'état des eaux de surface - Système d’évaluation de l’état des Eaux (SEEE) - Invertébrés	10.15468/qnez2y	GBIF	53
ATBI Parc national du Mercantour/Parco naturale Alpi Marittime - Jeux de données provenant de l'ATBI Mercantour	10.15468/jtlspu	GBIF	52
CNCR/Colección Nacional de Crustaceos	10.15468/b3huws	GBIF	51
Données du CEN Picardie concernant la Faune, la Flore et la Fonge - Données faune, flore et fonge du Conservatoire des espaces naturels de Picardie	10.15468/eogcjy	GBIF	51
(Table 2a and b) Median abundances of macrobenthos in surface sediments	10.1594/pangaea.734774	GBIF	48
Biodiversidad de macroinvertebrados bénticos de la región marina Tijuana-Ensenada Baja California, México	10.15468/qb1kdr	GBIF	48

Table B1. Continued

Dataset name	DOI	Source	# Records
ATBI Parc national du Mercantour/Parco naturale Alpi Marittime - EXPLOR'NATURE 2018, inventaire biologique de la commune de Sospel	10.15468/s1cjxq	GBIF	47
BioFresh Pond Data	10.13148/bf76	GBIF	47
Marine Species Records from Skomer Marine Conservation Zone (MCZ) Marine Monitoring Programme	10.15468/207iog	GBIF	47
SER Site-based Surveys	10.15468/h2yko0	GBIF	47
Stackpole National Nature Reserve Species Inventory and Ad-hoc Sightings from Across Pembrokeshire	10.15468/k6hvb8	GBIF	47
Manx Wildlife Trust - Records	10.15468/4hydf5	GBIF	46
Alterra (NL) - Entomofauna inventory in peat swamps	10.15468/ibom6z	GBIF	45
National Trust for Scotland Species Records	10.15468/a5y1cz	GBIF	45
Yorkshire Naturalists Union Marine and Coastal Section Records	10.15468/ajwexx	GBIF	45
A checklist of the marine Anthuroidea (Crustacea: Isopoda: Cymothoida) from the reefs of Peninsular Malaysia, with some new distributional data	10.3897/bdj.8.e54748	GBIF	44
Biodiversity4all Research-Grade Observations	10.15468/njmmp7	GBIF	44
ATBI Parc national du Mercantour/Parco naturale Alpi Marittime - EXPLOR'NATURE 2017, inventaire biologique de la commune de Barcelonnette	10.15468/ru5aks	GBIF	43
Bob Merritt dataset of Nottinghamshire invertebrates	10.15468/1pcd8f	GBIF	43
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste DEHALLEUX Axel provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/7eccel	GBIF	43
Patrick Roper’s Notebooks	10.15468/ntnedq	GBIF	41
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Tillier Pierre provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/7do6za	GBIF	41
Diversidad bentónica de la laguna Celestún, Yucatán	10.15468/o0mytw	GBIF	39
Galathea II, Danish Deep Sea Expedition 1950-52	10.15468/ouseij	GBIF	39
Isle of Man Historical Wildlife Records pre-1990	10.15468/n7ybfm	GBIF	39
Mesopelagic Crustaceans of the North Western Portuguese Coast between 1998 and 2000	10.15468/3mer8v	GBIF	39
Isle of Man historical wildlife records 1990 to 1994	10.15468/aru16v	GBIF	38
NCSM Non-molluscan Invertebrates Collection	10.36102/dwc.8	GBIF	38

Table B1. Continued

Dataset name	DOI	Source	# Records
The Rock Pool Project database - intertidal species records from rocky shore habitats - from February 2019	10.15468/bqpd1n	GBIF	38
Invertebrate surveys of various ponds in Scotland between 2010 and 2015	10.15468/c66cp5	GBIF	37
Effect of short-term meteorological disturbance on submergem aquatic vegetation and associated fauna	10.15468/v2dd3g	GBIF	36
La Planète revisitée - Corse 2019-2022 - La Planète Revisitée Corse 2021: prospections Côte orientale et Cap Corse	10.15468/g5astt	GBIF	36
Weddell Sea macrozoobenthos EASIZ I	10.15468/0ozw5m	GBIF	36
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Monsieur DAMOISEAU Sébastien	10.15468/kqswg4	GBIF	35
RHS monitoring of native and naturalised plants and animals at its gardens and surrounding areas	10.15468/mjksef	GBIF	35
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Contribution de la Ligue Insulaire Spéléogique de Corse à l'inventaire des arthropodes cavernicoles de Corse, dans le cadre de l'appel à projets INPN 2019	10.15468/24bs45	GBIF	34
Field Museum of Natural History (Zoology) Invertebrate Collection	10.15468/6q5vuc	GBIF	34
Invertebrates Collection of the Swedish Museum of Natural History	10.15468/eyda6l	GBIF	34
Monitoring data from the federal state Berlin (Germany)	10.13148/bfe89	GBIF	34
Bay of Fundy Species List	10.15468/nztqk6	GBIF	33
Macro benthos in surface sediments sampled during POLARSTERN cruise ANT-XXIII/8	10.1594/pangaea.718106	GBIF	33
New species and new records of terrestrial isopods (Crustacea, Isopoda, Oniscidea) of the families Philosciidae and Scleropactidae from Brazilian caves	10.5852/ejt.2020.606	GBIF	33
Radnorshire Wildlife Trust records held by BIS	10.15468/hxqokw	GBIF	33
Sbp-BioBlitz2017_60years-BiodiversityData	10.15468/jgnjsa	GBIF	33
Seasearch Marine Surveys in Scotland	10.15468/0hyjxi	GBIF	33
The Deepwater Program: Northern Gulf of Mexico Continental Slope Habitat and Benthic Ecology - DgoMB: Trawls	10.15468/qjow1m	GBIF	33

Table B1. Continued

Dataset name	DOI	Source	# Records
Ibaraki Nature Museum, Arthropoda collection	10.15468/auw14q	GBIF	32
SILENE-FAUNE-PACA - Parc_National_du_Mercantour_2017_12_18	10.15468/2ccdre	GBIF	32
Données LPO Réserves Naturelles Nationales - Données des réserves naturelles nationales co-gérées par la Ligue pour la protection des oiseaux (LPO) et l'Office français de la biodiversité (OFB)	10.15468/zpi2ni	GBIF	31
SILENE-FAUNE-PACA - Amis_des_Marais_du_Vigueirat_2017_12_18	10.15468/tj0rsp	GBIF	31
Banco de Datos de la Biodiversidad de la Comunitat Valenciana	10.15468/b4yqdy	GBIF	30
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Xavier JAPIOT	10.15468/hswza8	GBIF	30
Stoke-on-Trent Environmental Survey results (1982-1984)	10.15468/8gryb6	GBIF	30
A review of the genus Parapenaeon Richardson, 1904 (Crustacea: Isopoda: Bopyridae: Orbioninae), with description of three new species from China	10.1080/00222933.2015.1023227	GBIF	29
NRW Regional Data: North Wales	10.15468/krljpu	GBIF	29
Review of the species of the Cirolana ‘parva -group’ (Cirolanidae: Isopoda: Crustacea) in Indonesian and Singaporean waters	10.11646/zootaxa.4317.3.1	GBIF	29
TestWat - Macroinvertebrates and macrophytes of freshwater bodies in Flanders, Belgium	10.15468/xzpcvv	GBIF	29
USGS Nonindigenous Aquatic Species database	10.15468/ijccz9	GBIF	29
MBON POLE TO POLE: SANDY BEACH BIODIVERSITY OF YUCATAN COAST	10.15468/g7kwfh	GBIF	28
NatureMapr	10.15468/uye32x	GBIF	28
Benthic invertebrate surveys conducted between 2009-2011 as part of the Sydney Tar Ponds Cleanup and Coke Ovens Remediation Project.	10.15468/mzmzvn	GBIF	27
Computarización de la Colección Nacional de Crustáceos del Instituto de Biología, UNAM y elaboración de su catálogo	10.15468/9wpc6o	GBIF	27
SEFSC CAGES Alabama Fish Length Data with CPUE	10.15468/vs1k7e	GBIF	27
Survey and monitoring records for Scottish Wildlife Trust reserves from reserve convenors and Trust volunteers - Verified data	10.15468/yyd4b9	GBIF	27
Colección de Artropodos del Museo de Historia Natural Marina de Colombia - Makuriwa	10.15472/eateut	GBIF	26

Table B1. Continued

Dataset name	DOI	Source	# Records
Six new species of Anthuridae (Peracarida: Isopoda) from the southern Mexican Pacific	10.5852/ejt.2021.760.1441	GBIF	26
Ocean Genome Legacy Collection	10.15468/jzgqtc	GBIF	25
Riparia	10.15468/wunv9t	GBIF	25
Collection Crustacea ZMG	10.15468/1p3n36	GBIF	24
Explore Your Shore	10.14284/563	GBIF	24
FBIP: Offshore Benthic Macrofauna Data	10.15468/aaspie	GBIF	24
Abundance of benthic infauna in surface sediments from the North Sea sampled during Walther Herwig cruise WH220	10.1594/pangaea.756783	GBIF	23
Invertebrados y aves playeras de la Laguna Madre de Tamaulipas, México (Crustáceos)	10.15468/zbfnoa	GBIF	23
Invertebrate data from Selected Grazing Marshes	10.15468/88fiys	GBIF	23
AM: Freshwater Invertebrates (1900-2005)	10.15468/spzgor	GBIF	22
Crustáceos estomatópodos, anfípodos, isópodos y decápodos del litoral de Quintana Roo	10.15468/tbpto6	GBIF	22
Biodiversitätsdatenbank Salzburg	10.15468/3pf855	GBIF	21
Invertebrate Collection of IBIOMAR (CCT CONICET-CENPAT) (CNP-INV)	10.15468/n7mdpb	GBIF	21
MDFRC macroinvertebrate survey	10.15468/paioao	GBIF	21
Redescriptions and new species in the ‘ Austrosignum-Munnogonium’ complex sensu Just & Wilson (2007), mainly from the Southern Hemisphere (Crustacea Isopoda: Paramunnidae)	10.11646/zootaxa.4952.3.1	GBIF	21
South American Antarctic Marine Biodiversity Literature	10.15468/kxnpuq	GBIF	21
Système d'Information sur la Nature et les Paysages d'Ile de France - Étude biodiversité toitures végétalisées d'Île-de-France réalisée par Natureparif/ARB	10.15468/6zdm6k	GBIF	21
Système d'Information sur la Nature et les Paysages d'Ile de France - Inventaire éclair de Natureparif 2017	10.15468/l5zxvh	GBIF	21
ALA species sightings and OzAtlas	10.15468/jayxmn	GBIF	19
Macrobenthos of the Western Scheldt (Ossenisse, Valkenisse, Terneuzen and Vlissingen) on 27 and 28 September 1978	10.14284/231	GBIF	19
Nonindigenous Aquatic Species (NAS) Database Non-freshwater Specimens	10.15468/e9lcrw	GBIF	19
WTSWW Data: All Taxa (West Wales)	10.15468/gaakk2	GBIF	19
Isle of Man historical wildlife records 1995 to 1999	10.15468/lo2tge	GBIF	18
Maritimes Summer Research Vessel Surveys	10.15468/93x7ec	GBIF	18
New gnathiid isopod crustaceans (Cymothoida) from Heron Island and Wistari Reef, southern Great Barrier Reef	10.11646/zootaxa.4609.1.2	GBIF	18

Table B1. Continued

Dataset name	DOI	Source	# Records
NIEK_NES	10.15468/u9dkvz	GBIF	18
SeamountsOnline	10.15468/2juyaz	GBIF	18
Galiano Island BC Canada Marine Zoology 1893–2021	10.15468/gv9cy5	GBIF	17
GEO-Hauptveranstaltung (Duisburg)	10.15468/mhmojc	GBIF	17
Macro- and megafauna from the North Aegean Sea from 1997-1998	10.15468/8jbgwk	GBIF	17
On the composition of the benthic fauna of the western Fram Strait	10.15468/lbi9s0	GBIF	17
Registro de macrofauna bentónicas submareales de fondos blandos, variables físico-químicos en la zona sur – Registro 5	10.15468/3ng7tv	GBIF	17
Sizing Ocean Giants	10.15468/mfxiws	GBIF	17
Taxonomy of Alpioniscus (Illyrionethes): A. magnus and three new species from the Dinaric Karst (Isopoda: Oniscidea: Trichoniscidae)	10.11646/zootaxa.4657.3.4	GBIF	17
ZooplanktonBeaufortSeaNOGAP1	10.15468/lhtmpo	GBIF	17
Inventario faunístico de dos regiones terrestres prioritarias de la península de Baja California: San Telmo-San Quintín y Planicies de Magdalena (Ejemplares)	10.15468/mn8cr9	GBIF	16
IPOE_Benthos_Steffens	10.15468/mttec8	GBIF	16
Projet éolien en mer de Dieppe Le Tréport - Etat initial benthique et sédimentaire	10.15468/aatgex	GBIF	16
Réserves Naturelles de France (RNF) - RNF - Données de la Fédération des Réserves Catalanes	10.15468/ufryrd	GBIF	16
Swiss Occurrence Records of Non-Native Species of Various Faunal Groups	10.15468/z3vjjw	GBIF	16
Système d'Information sur la Nature et les Paysages d'Ile de France - Inventaire éclair de Natureparif 2016	10.15468/yitdzl	GBIF	16
Woodmeadow Invertebrate Survey 2018	10.15468/f4n4wf	GBIF	16
Data from Defra Family Organisations supplied to Staffordshire Ecological Record	10.15468/giebpp	GBIF	15
iRecord Surveys	10.15468/i7x5ca	GBIF	15
Alterra (NL) - Entomofauna inventory in dead wood	10.15468/2pz1c0	GBIF	14
Manx Biological Recording Partnership VERIFIED Isle of Man records between 14/02/2017 and 05/09/2019	10.15468/dudjqq	GBIF	14
Marine invertabrate(ARTHROPODA) specimen database of Osaka Museum of Natural History	10.15468/npb02o	GBIF	14
NE Scotland other invertebrate records 1800-2010	10.15468/ifjfxz	GBIF	14

Table B1. Continued

Dataset name	DOI	Source	# Records
New genera and species of the marine isopod family Serolidae (Crustacea, Sphaeromatidea) from the southwestern Pacific	10.3897/zookeys.18.96	GBIF	14
Benthic communities and environmental parameters in Amvrakikos Wetlands: Mazoma, Tsopeli,Tsoukalio, Rodia and Logarou lagoons (September 2010 – July 2011)	10.15468/dffi6y	GBIF	13
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Benoit MARTHA	10.15468/bsbtam	GBIF	13
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Brendan ALLIGAND	10.15468/ojaotz	GBIF	13
DASSH Data Archive Centre expert sightings records	10.15468/tggq3w	GBIF	13
GEO-Hauptveranstaltung in "Wildtierland"	10.15468/ebnnbs	GBIF	13
Macro-invertebrates of the Desna river basin	10.15468/cwjh3n	GBIF	13
Manx Biological Recording Partnership UNVERIFIED Isle of Man records between 14/02/2017 and 05/09/2019	10.15468/6stnx8	GBIF	13
Museu de Ciències Naturals de Barcelona: MCNB-Art	10.15468/pewzzr	GBIF	13
Observations naturalistes indépendantes d'origine privée partagées sur la base de données Kollect Nouvelle-Aquitaine - Observations faunistiques de Jean-Christophe BARTOLUCCI	10.15468/svmtj4	GBIF	13
Peracarida of Bernardo O'Higgins National Park (S Chile)	10.15468/m32pbu	GBIF	13
Registros faunísticos de invertebrados marinos en el SE del Golfo de California	10.15468/tjvs4i	GBIF	13
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure ANVL (Association des Naturalistes de la Vallée du Loing et du Massif de Fontainebleau) provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/ruvxhy	GBIF	13
Two new species and new records of terrestrial isopods (Crustacea, Isopoda, Oniscidea) from Brazilian caves	10.11646/zootaxa.4564.2.6	GBIF	13
Woodmeadow Invertebrate Survey 2019	10.15468/tnp8ek	GBIF	13
Zooplankton NOGAP32b 1986	10.15468/z7cj59	GBIF	13

Table B1. Continued

Dataset name	DOI	Source	# Records
Atlantic Reference Centre Museum of Canadian Atlantic Organisms - Invertebrates and Fishes Data	10.15468/wsxvo6	GBIF	12
Bibliographie de la faune, la flore et la fonge de France métropolitaine et outre-mer - Bibliographique de la faune, la flore et la fonge de France métropolitaine et outre-mer	10.15468/cthncb	GBIF	12
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de DUSOULIER François	10.15468/vfew3b	GBIF	12
Entomological collections, UiB	10.15468/irppio	GBIF	12
FBIP: SeaKeys_SANBI: Marine images iSpot_2013	10.15468/xu84gq	GBIF	12
Macrobenthos and Meiobenthos Tuktoyaktuk Harbor and Mason Bay 1985-1988 NOGAP	10.15468/9wnx7f	GBIF	12
Marine fauna survey of the Vestfold Hills and Rauer Island, 1981-82	10.15468/jauvuu	GBIF	12
PlutoF platform reference-based occurrences	10.15468/e15jve	GBIF	12
SeaWatch-B: citizens monitoring the Belgian North Sea from the beach (2014-2018)	10.14284/401	GBIF	12
SNH Invertebrate Site Condition Monitoring 2015-16: Culbin Sands, Culbin Forest and Findhorn Bay SSSI	10.15468/bg1iit	GBIF	12
Biological survey of the intertidal chalk reefs between Folkestone Warren and Kingsdown, Kent 2009-2011	10.15468/opmkmp	GBIF	11
Brecknock Wildlife Trust (Now WTSWW Brecknockhire) records held by BIS	10.15468/hd7pvq	GBIF	11
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Julien BIRARD [Inventaire Eclair 18 juin]	10.15468/rrps8r	GBIF	11
Cirolanides wassenichae sp. nov., a freshwater, subterranean Cirolanidae (Isopoda, Cymothoida) with additional records of other species from Texas, United States	10.11646/zootaxa.4543.4.2	GBIF	11
Crustacea specimens of Ryukyu University Museum (Fujukan)	10.15468/vdwqzo	GBIF	11
Crustacean collection of the National Museum of Nature and Science	10.15468/xdcsbl	GBIF	11

Table B1. Continued

Dataset name	DOI	Source	# Records
DNA barcoding and morphological studies confirm the occurrence of three Atarbolana (Crustacea: Isopoda: Cirolanidae) species along the coastal zone of the Persian Gulf and Gulf of Oman	10.11646/zootaxa.4200.1.7	GBIF	11
Données Faune Base SIRFF - FNE Centre-Val de Loire - Système d'Information Régional sur la Faune et la Flore - FNE Centre-Val de Loire - Données saisies entre 01-01-2017 et 31-03-2018	10.15468/aotvir	GBIF	11
Gesamtartenliste Bremerhaven, Helgoland und Sylt	10.15468/85zmqv	GBIF	11
Isle of Man wildlife records from 01/01/2000 to 13/02/2017	10.15468/mopwow	GBIF	11
Museum of Southwestern Biology, Division of Arthropods	10.15468/jtovgy	GBIF	11
New species of subterranean and endogean terrestrial isopods (Crustacea, Oniscidea) from Tuscany (central Italy)	10.5252/zoosystema2018v40a11	GBIF	11
Porcupine Marine Natural History Society Records	10.15468/c8jqsr	GBIF	11
St Andrews BioBlitz 2015	10.15468/xtrbvy	GBIF	11
Staffordshire Wildlife Trust Nature Reserves Inventory	10.15468/vhdows	GBIF	11
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure Ville de Paris provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/0obof8	GBIF	11
Three new species of the genus Ischnomesus (Isopoda: Asellota: Ischnomesidae) from Brazilian deep sea	10.11646/zootaxa.4312.2.3	GBIF	11
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Jean-Michel LEMAIRE	10.15468/tr1xmq	GBIF	10
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Pierre NOEL (pnoel)	10.15468/ziwt1p	GBIF	10
Collections of Bioclass, school #179, Moscow	10.15468/4f0bmt	GBIF	10
Marine Benthic Fauna List, Island of Læsø, Denmark	10.15468/ty0smg	GBIF	10
Merseyside BioBank (verified)	10.15468/ar0p6s	GBIF	10
National indicator data for river condition in New Zealand	10.15468/rcmdre	GBIF	10
SNH Invertebrate Site Condition Monitoring 2015-16: Methven Woods SSSI	10.15468/dv1u0r	GBIF	10

Table B1. Continued

Dataset name	DOI	Source	# Records
Terrestrial and limnic invertebrates systematic collection, NTNU University Museum	10.15468/fsreqb	GBIF	10
Urban Roots Malls Mire Invertebrate Records 2014	10.15468/kpl3zg	GBIF	10
ZooplanktonBeaufortSeaNOGAP2	10.15468/jchc6q	GBIF	10
(Table 2a) Abundance of macrobenthos species in samples from Cruise AMK54 stations in the Novaya Zemlya Trough	10.1594/pangaea.769660	GBIF	9
Anymals+plants - Citizen Science Data	10.15468/ee6ps6	GBIF	9
Artenvielfalt der Nordsee - Helgoland	10.15468/omx28y	GBIF	9
Base BOMBINA du Parc Naturel régional Lorraine - Données bibliographique de la base BOMBINA	10.15468/jrd2ir	GBIF	9
Bioblitz 2014 Kalvebod Fælled, Denmark	10.15468/4entqy	GBIF	9
Carrière calcaire de Chateaupanne (Mauges-sur-Loire) - modification des conditions d'exploitation - renouvellement de l’autorisation - inventaires 2004-2014 pour étude d'impact carrière	10.15468/a9f7rb	GBIF	9
Consultancy Contract Surveys - Marine Species Records	10.15468/hs4wyn	GBIF	9
Continuous Plankton Recorder Dataset (SAHFOS)	10.15468/ygwilu	GBIF	9
Dorset SSSI Species Records 1952 - 2004 (Natural England)	10.15468/vcjzts	GBIF	9
INSDC Host Organism Sequences	10.15468/e97kmy	GBIF	9
Macrobenthos monitoring in function of aggregate extraction activities in the Belgian part of the North Sea	10.14284/199	GBIF	9
Montgomeryshire Wildlife Trust records held by BIS	10.15468/vozyfp	GBIF	9
Morphology and Taxonomy of Isopoda Anthuroidea (Crustacea) from Sulawesi with description of six new species	10.5852/ejt.2021.768.1501	GBIF	9
Plan de gestion multi-sites à Chiroptères mené par le CEN Aquitaine depuis 2008 - Observations faunistiques fortuites réalisées par le CEN Aquitaine	10.15468/egq9qc	GBIF	9
Projet de raccordement au réseau public de transport d'électricité du parc éolien en mer de Dieppe - Le Tréport - Campagne Benthos Benne et Drague 2015-2016 - Projet de raccordement du DLT	10.15468/m5rswu	GBIF	9
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de l'association RENARD provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/izjsyw	GBIF	9

Table B1. Continued

Dataset name	DOI	Source	# Records
Taxonomy of Paraplatyarthrus Javidkar and King (Isopoda: Oniscidea: Paraplatyarthridae) with description of five new species from Western Australia, and comments on Australian Trichorhina Budde-Lunde, 1908 (Platyarthridae)	10.11646/zootaxa.4243.3.1	GBIF	9
Woodmeadow Invertebrate Survey 2015	10.15468/zyqxsa	GBIF	9
A Review Of Bopyrids (Crustacea: Isopoda: Bopyridae) Parasitic On Caridean Shrimps (Crustacea: Decapoda: Caridea) From China	10.1206/amnb-921-00-01.1	GBIF	8
Alterra (NL) - Entomofauna inventory in edges of arable fields province of Groningen	10.15468/bu8fea	GBIF	8
Base de datos de la Sala de Colecciones Biológicas de la Universidad Católica del Norte (SCBUCN)	10.15468/d3auf9	GBIF	8
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Cyril EPICOCO	10.15468/th8va6	GBIF	8
Colecciones Zoológicas de la Universidad de León, Colección de Malacostráceos	10.15468/yyaxl7	GBIF	8
Crustáceos de la Colección de Referencia de Biología Marina de la Universidad del Valle (CERBMcr-UV)	10.15472/uofnlo	GBIF	8
Deep-sea megabenthos of the Eurasian Central Arctic based on image analysis.	10.15468/u2vs8v	GBIF	8
Lizard Island Research Station	10.15468/5bsrkm	GBIF	8
MACROBENTHOS COMPOSITION FROM THE RIO LAGARTOS HYPERHALINE COASTAL LAGOON SYSTEM, YUCATAN, MEXICO	10.15468/yvggw4	GBIF	8
Macrobenthos monitoring at long-term monitoring stations in the Belgian part of the North Sea from 2001 on	10.14284/202	GBIF	8
Rådgivende Biologer	10.15468/tqxvcg	GBIF	8
SNH Invertebrate Site Condition Monitoring 2015-16: Rum SSSI	10.15468/sgiez0	GBIF	8
Survey data of tidal flats on the Monitoring sites 1000 project, BDCJ	10.15468/bywe7w	GBIF	8
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Danten Benoît provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/ceht86	GBIF	8
The CPR Survey	10.17031/1629	GBIF	8

Table B1. Continued

Dataset name	DOI	Source	# Records
The first record of Gnathostenetroididea Kussakin, 1967 from Australian waters with description of four new species of Gnathostenetroides Amar, 1957 (Crustacea: Isopoda: Asellota) from the Great Barrier Reef	10.11646/zootaxa.4554.2.1	GBIF	8
Universidad de San Carlos de Guatemala - Colección de Crustáceos	10.15468/pm2gef	GBIF	8
Woodmeadow Invertebrate Survey 2016	10.15468/1epq4f	GBIF	8
2019_nsmk_ms_20200324	10.15468/hnojw7	GBIF	7
Analysis of the macrobenthic community near Nieuwpoort (1970-1971)	10.14284/206	GBIF	7
Benthic Epifauna Biomass and Abundance Data, Arctic Marine Biodiversity Observing Network (AMBON) research cruise, August 2017	10.15468/yg2y7v	GBIF	7
Colección del Departamento de Biología Animal (Zoología) de la Universidad de La Laguna	10.15468/yevjxm	GBIF	7
COMARGIS: Information System on Continental Margin Ecosystems	10.15468/0djslr	GBIF	7
Data on the biodiversity of macrophyte communities and associated aquatic organisms in lakes of the Vologda Region (North-Western Russia): algae and invertebrates	10.15468/yy3dx5	GBIF	7
Diveboard - Scuba diving citizen science observations	10.15468/tnjrgy	GBIF	7
HBRG Highland Seashore Project Dataset	10.15468/sau7qh	GBIF	7
iNaturalist records from Northern Ireland	10.15468/ctgb63	GBIF	7
Löydös Open Finnish Observation Database	10.15468/8fzv2j	GBIF	7
Macroinvertebrados bentónicos de playas de arena (Monte Hermoso-Pehuen Có, Buenos Aires, Argentina)	10.15468/sfn1ql	GBIF	7
Northern Ireland Environmental Recorders - Marine Species Records	10.15468/y25jdr	GBIF	7
Saisie naturaliste opportuniste dans SICEN Occitanie - Données opportunistes du CEN Midi-Pyrénées	10.15468/jqsgs8	GBIF	7
Seasearch Marine Surveys in the Channel Islands	10.15468/0ppp4p	GBIF	7
Sheffield and Rotherham Wildlife Trust - Records	10.15468/x3wdpp	GBIF	7
Six new epigean species of Caecidotea (Isopoda: Asellidae) distributed along the Trans-Mexican Volcanic Belt in Central Mexico	10.11646/zootaxa.4965.1.2	GBIF	7
Species list recorded by baited cameras at deep sea area in Japan	10.15468/vgfyzy	GBIF	7
St Andrews BioBlitz 2014	10.15468/erweal	GBIF	7
Ty Canol National Nature Reserve (NNR) Species Inventory	10.15468/shxquu	GBIF	7

Table B1. Continued

Dataset name	DOI	Source	# Records
UAM Insect Collection (Arctos)	10.15468/qs8slz	GBIF	7
A review of the genus Apocepon Nierstrasz & Brender à Brandis (Isopoda: Epicaridea: Bopyridae) parasitic on purse crabs (Decapoda: Brachyura: Leucosiidae) from Chinese waters, with description of a new species	10.5281/zenodo.2646359	GBIF	6
Artenvielfalt am Eich-Gimbsheimer Altrhein	10.15468/3hek17	GBIF	6
Artenvielfalt auf der Weide - GEO-Hauptveranstaltung in Crawinkel	10.15468/tzcuw3	GBIF	6
BRERC Notable Species records within the last 10 years	10.15468/vntgox	GBIF	6
BUND - Dassower See (Lübeck/Dassow)	10.15468/qvd4dz	GBIF	6
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de BLOND Cyrille	10.15468/whsrzt	GBIF	6
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de LEPAREUR Fanny	10.15468/7vuca9	GBIF	6
Community Foundation for Ireland Records	10.15468/vpbxgk	GBIF	6
Dalbekschlucht	10.15468/kezgzc	GBIF	6
Estonian Museum of Natural History Department of Zoology	10.15468/98cxtc	GBIF	6
First record of the genus Agnara (Isopoda: Oniscidea) from Iran with descriptions of two new species	10.1080/00222933.2016.1193645	GBIF	6
GEO-Hauptveranstaltung (Insel Vilm)	10.15468/zgyujv	GBIF	6
Macrozoobenthos composition, abundance and biomass in the Arctic Ocean along a transect between Svalbard and the Makarov Basin 1991	10.15468/iaaimu	GBIF	6
Malacostracans Specimens of Akita Prefectural Museum	10.15468/uftxpn	GBIF	6
MoJ BioBlitz August 2018	10.15468/gbyber	GBIF	6
Observations of three Idotea species (I. balthica, I. chelipes and I. granulosa) in Northern Europe, including the Baltic Sea - field data	10.14284/8	GBIF	6
SILENE-FAUNE-PACA - Ville_de_Digne_les_Bains_2017_12_18	10.15468/cyqhjs	GBIF	6
SNH Invertebrate Site Condition Monitoring 2013/14: Black Wood of Rannoch SSSI	10.15468/vue3gs	GBIF	6
Species found in the NOMS estate 2005 - Present	10.15468/wcx4is	GBIF	6

Table B1. Continued

Dataset name	DOI	Source	# Records
Subtidal macrobenthos monitoring in function of a foreshore suppletion at the Belgian coast, period 2013-2016	10.14284/342	GBIF	6
Swiss Occurrence Records of Native Species of Various Faunal Groups	10.15468/sj6q9x	GBIF	6
Texas Tech University - Invertebrate Zoology	10.15468/ga4bmd	GBIF	6
University of Tartu Natural History Museum and Botanical Garden Zooloogical Collections	10.15468/6hfnux	GBIF	6
Base de données faune - DREAL Centre-Val de Loire. Polygones	10.15468/iwm38f	GBIF	5
BD de la plateforme partagée pour la diffusion des données naturalistes de Haute-Normandie - Inventaires faunistiques et floristiques de la ville du Havre	10.15468/3uduna	GBIF	5
Biodiversidad de crustáceos dulceacuícolas del centro de Nuevo León y noroeste de Tamaulipas (R53, Río San Juan y Río Pesqueria)	10.15468/gsafre	GBIF	5
data.mnhn.lu observation data	10.15468/n4k9j5	GBIF	5
Distribution of haploniscids (Isopoda, Asellota, Haploniscidae) in Icelandic waters, with description of Haploniscus astraphes n. sp. from the Iceland basin and the Southeast Atlantic Ocean	10.11646/zootaxa.4231.3.1	GBIF	5
Haplomunnidae (Crustacea: Isopoda) reviewed, with a description of an intact specimen of Thylakogaster Wilson & Hessler, 1974	10.11646/zootaxa.326.1.1	GBIF	5
IBSA Surveys from Bennelongia Environmental Consultants	10.15468/thwwv5	GBIF	5
Macrobenthos Chukchi Sea, 1986	10.15468/owfiui	GBIF	5
Nivå Bay species list, Zealand, Denmark	10.15468/97nj93	GBIF	5
Observations faune et flore du Parc national des Écrins	10.15468/xckykf	GBIF	5
Ohio Wesleyan University Parasite Specimens (Arctos)	10.15468/8njvkv	GBIF	5
PondNet data 2012-2014	10.15468/qv8ped	GBIF	5
Réserves Naturelles de France (RNF) - RNF - LPO Rhône-Alpes	10.15468/bzahe2	GBIF	5
Seasearch Marine Surveys in Wales	10.15468/4us2hk	GBIF	5
SNH Invertebrate Site Condition Monitoring 2013/14: Earlshall Muir SSSI	10.15468/9dk3dk	GBIF	5
SNH Invertebrate Site Condition Monitoring 2015-16: Morrich More SSSI	10.15468/tofgk4	GBIF	5
St Andrews BioBlitz 2016	10.15468/146yiz	GBIF	5

Table B1. Continued

Dataset name	DOI	Source	# Records
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure Naturessonne provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/pfgk5j	GBIF	5
Temporal data series of Benthic macrofauna abundance and composition from the Patos Lagoon estuary	10.15468/lsoc2v	GBIF	5
Three new species of Ischioscia Verhoeff, 1928 (Isopoda, Oniscidea, Philosciidae) from Serranía de Perijá, Andean Cordillera, Colombian Caribbean	10.5252/zoosystema2020v42a8	GBIF	5
Two new species of idoteid isopods (Crustacea, Isopoda, Idoteidae) from Korea	10.11646/zootaxa.4858.2.2	GBIF	5
Water Framework Directive AGE, Recorder-Lux database	10.15468/mhcb8w	GBIF	5
Abundance of macrobenthos organisms in the northern Wadden Sea in 2007	10.1594/pangaea.755036	GBIF	4
Abundance of macrobenthos organisms in the northern Wadden Sea in 2008	10.1594/pangaea.755037	GBIF	4
Abundance of macrobenthos organisms in the northern Wadden Sea in 2009	10.1594/pangaea.755038	GBIF	4
Abundance of macrobenthos organisms in the northern Wadden Sea in 2010	10.1594/pangaea.755039	GBIF	4
Alterra (NL) - Entomofauna inventory in cattle grazed dune grassland	10.15468/zp5oif	GBIF	4
Alterra (NL) - Entomofauna inventory in Speulder forest	10.15468/bzy3j3	GBIF	4
Antarctic Marine Species Sequence Data	10.15468/q2xdwg	GBIF	4
Artenvielfalt auf dem Schulgelände	10.15468/0ioibr	GBIF	4
Atlas of Life in the Coastal Wilderness	10.15468/rtxjkt	GBIF	4
Biological Reference Collections ICM-CSIC	10.15470/qlqqdx	GBIF	4
Biotope in Rheine - Aktion 350	10.15468/gpphjn	GBIF	4
Cancrion khanhensis sp. nov. (Crustacea: Isopoda: Entoniscidae) infesting Monomia haanii (Stimpson, 1858) (Crustacea: Brachyura: Portunidae) from Nha Trang Bay, Khanh Hoa, Vietnam, with remarks on larval stages of entoniscids and description of a new family, genus and two new species of hyperparasites	10.11646/zootaxa.4894.3.4	GBIF	4
Casual records for Scottish Wildlife Trust reserves - Verified data	10.15468/zeay1d	GBIF	4
Colección Carcinológica de Yucatán	10.15468/qaecsh	GBIF	4
Crustacea Collection of Natural History Museum and Institute, Chiba	10.15468/jkxmar	GBIF	4

Table B1. Continued

Dataset name	DOI	Source	# Records
Crustacea specimens of Kuroshio Biological Research Foundation	10.15468/al2by0	GBIF	4
Description of four new species of the Cirolana ‘ parva group’ (Crustacea: Isopoda: Cirolanidae) from Thailand, with supporting molecular (COI) data	10.1080/00222933.2016.1180718	GBIF	4
Dipterists Forum: Field Week 2017 (Snowdonia)	10.15468/u77728	GBIF	4
Données d'observations des plongeurs de la FFESSM - Données DORIS de la FFESSM	10.15468/vguvbq	GBIF	4
Données faune, flore et fonge du Conservatoire des espaces naturels du Nord Pas-de-Calais	10.15468/afs3ve	GBIF	4
Further investigations of the effects of the Nella Dan oil spill 1988/94	10.4225/15/54adb63de539d	GBIF	4
GEO-Hauptveranstaltung Bodden (Vilm)	10.15468/ismecy	GBIF	4
Greenland macrobenthos 2006	10.15468/u7ulpu	GBIF	4
Hatikka.fi observations	10.15468/te1t6l	GBIF	4
Inventaire de la Réserve naturelle de l'Etang noir - Données de présence récoltées	10.15468/uzxvll	GBIF	4
Inventaire entomologique standardisé des ZNIEFF de Martinique - Observations diverses réalisées à l'occasion de l'inventaire entomologique des ZNIEFF de Martinique	10.15468/wa7eed	GBIF	4
Inventario computarizado de la colección de parásitos de peces del noroeste de México	10.15468/vehk6o	GBIF	4
Inventory and BioBlitz Records from rare Charitable Research Reserve	10.5886/hh6td9jn	GBIF	4
Kiesbagger (Mittelhausen)	10.15468/kxby8i	GBIF	4
Klutensee	10.15468/bb4gdi	GBIF	4
Langes Tannen (Uetersen)	10.15468/1beryy	GBIF	4
Liaison autoroutière concédée entre Machilly et Thonon les Bains et suppression des passages à niveau N° 65 et 66 à Perrignier Haute-Savoie - Crustacés - Inventaire non standardisé - FRAPNA74 - 24HNAT	10.15468/dbk2cb	GBIF	4
Local BioBlitz Challenge 2013	10.15468/gcyq62	GBIF	4
Museu Paraense Emílio Goeldi - Carcinológica Collection	10.15468/tmqcgv	GBIF	4
Naturschutzgebiet Bausenberg (Niederzissen)	10.15468/xjeiwv	GBIF	4
Nottinghamshire Wildlife Trust - Records	10.15468/p2vf4u	GBIF	4
Numérisation des données faune contenues dans les Bulletins de la Société Linnéenne de Bordeaux - Bulletin de la Société Linnéenne de Bordeaux, Tome 144 (N.S) n° 37 (3), 2009 - Données faune	10.15468/9fanwu	GBIF	4

Table B1. Continued

Dataset name	DOI	Source	# Records
Numérisation des données faune contenues dans les Bulletins de la Société Linnéenne de Bordeaux - Bulletin de la Société Linnéenne de Bordeaux, Tome 148 (N.S) n° 41 (3), 2013 - Données faune	10.15468/8z2hrc	GBIF	4
Ontario BioBlitz Species Records	10.5886/mc7h1q	GBIF	4
Plankton&BenthosResearch	10.15468/f55mxn	GBIF	4
Port Phillip Bay Environmental Study Data 1992-1996 - infauna records	10.15468/tdbqff	GBIF	4
Programme CarNET B (Cartographie Nationale des Enjeux Territorialisés de Biodiversité Remarquable) - Carnet B Lorraine	10.15468/snvauq	GBIF	4
RACCORDEMENT ELECTRIQUE DE LA FERME EOLIENNE FLOTTANTE DE GROIX ET BELLE-ILE - Création de la liaison sous-marine et souterraine à 63 000 volts - Campagne benthos Benne2015-2017-Raccordement du parc éolien Groix/Belle-île	10.15468/uufgb2	GBIF	4
Réserves Naturelles de France (RNF) - RNF - Données de l'Association GEREPI (GEstion de la REserve naturelle nationale du PInail)	10.15468/vpufca	GBIF	4
SILENE-FAUNE-PACA - CEN_PACA_2017_12_18	10.15468/3kmwvz	GBIF	4
SNH Invertebrate Site Condition Monitoring 2015-16: Den of Airlie SSSI	10.15468/iorelw	GBIF	4
Study of epibenthos and demersal fish in and around the thiocarbamate discharge area of the Belgian Continental Shelf (1977-1981)	10.14284/204	GBIF	4
Suivi et inventaire de la réserve naturelle de Saucats - La Brède - Inventaire et suivi Entomologiques de 2012-2013	10.15468/pee75t	GBIF	4
Two new species and a new record of Metacirolana Kussakin, 1979 (Crustacea: Isopoda: Cirolanidae) from Indonesia	10.11646/zootaxa.4370.5.4	GBIF	4
UAM Insect Observations (Arctos)	10.15468/8nv0mp	GBIF	4
(Table 2) Megafauna density 2002, 2004 and 2007 in the deep-sea observatory AWI-HAUSGARTEN, Fram Strait	10.1594/pangaea.807446	GBIF	3
(Table 3a and b) Median biomass of macrobenthos in surface sediments	10.15468/dffsmm	GBIF	3
Alterra (NL) - Entomofauna inventory in Amerongen forest	10.15468/qli3ts	GBIF	3
Artenerfassung für Jedermann in der Grundschule Kirchboitzen	10.15468/fldsvx	GBIF	3
Artenvielfalt der Nordsee - Sylt	10.15468/nvhjkx	GBIF	3
Artenvielfalt rund um die Dalbek-Schule	10.15468/sstxbs	GBIF	3

Table B1. Continued

Dataset name	DOI	Source	# Records
Auburn University Museum of Natural History Invertebrates	10.15468/kpb05k	GBIF	3
BioBlitz Barcelona 2010-14	10.15470/ssy7h3	GBIF	3
Biodiversidad selecta de los humedales de Laguna de Términos - Pantanos de Centla (Crustáceos)	10.15468/4hcqs2	GBIF	3
Biotope auf dem Gelände der Eggerstedt- Kaserne in der Nachbarschaft der Theodor-Heuss-Schule	10.15468/k10mi2	GBIF	3
Brucerolis gen. n., and Acutiserolis Brandt, 1988, deep-water southern genera of isopods (Crustacea, Isopoda, Serolidae)	10.3897/zookeys.18.129	GBIF	3
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de MICHALKE Friedrich	10.15468/m88gaf	GBIF	3
Coastal and Marine Species Database	10.15468/oynwkx	GBIF	3
Collection Crustacea NHCY	10.15468/7zrnia	GBIF	3
Crustacea Collection - Instituto Nacional de Pesquisas da Amazônia (INPA)	10.15468/dihynq	GBIF	3
Cylindroniscus platoi (Isopoda: Oniscidea: Styloniscidae), a new cave-dwelling species from Lagoa Santa Karst, Southeastern Brazil	10.11646/zootaxa.4461.3.6	GBIF	3
Dalbek-Schlucht	10.15468/bztwdl	GBIF	3
Danisco-Wiese	10.15468/pkqdoq	GBIF	3
Données de l'association Indre Nature - Données faune Indre Nature 2016-2017	10.15468/vaso6t	GBIF	3
Données naturalistes du CEN Auvergne concernant la Faune, la Flore et la Fonge - Données naturalistes faune du Conservatoire des espaces naturels Auvergne saisies avant le 18 février 2019.	10.15468/igo6zd	GBIF	3
Données ONF faune-flore-fonge	10.15468/ykstli	GBIF	3
Dorset Sites of Nature Conservation Interest (SNCI) species records pre 2000	10.15468/qyg29v	GBIF	3
EDIT - ATBI in Mercantour/Alpi Marittime (France/Italy)	10.15468/4z4hto	GBIF	3
Fauna and flora of Sumskyi district in Sumy region of Ukraine	10.15468/tndu8x	GBIF	3
Freigelände Naturschutzscheune Reinheimer Teich (Kreis Darmstadt-Dieburg)	10.15468/uxdjq1	GBIF	3
GEO-Hauptveranstaltung im Nationalpark Bayerischer Wald	10.15468/bxxbmj	GBIF	3
Gestion de sites - Données invertébrés sites CEN MP	10.15468/8upqwk	GBIF	3
Gulf of Gdansk	10.14284/262	GBIF	3

Table B1. Continued

Dataset name	DOI	Source	# Records
Homoplasy and morphological stasis revealed through multilocus phylogeny of new myrmecophilous species in Armadillidiidae (Isopoda: Oniscidea)	10.15468/ugz5xb	GBIF	3
Illinois Natural History Survey Insect Collection	10.15468/eol0pe	GBIF	3
Inventaires menés dans le cadre du Groupe Invertébrés de Midi-Pyrénées (GIMP) - Données invertébrés en Midi-Pyrénées	10.15468/5bqura	GBIF	3
kerkyra	10.15468/jdyl94	GBIF	3
La Planète revisitée - Corse 2019-2022 - La Planète Revisitée Corse: observations lors des phases de reconnaissance et d'installation	10.15468/2ecziz	GBIF	3
Langes Tannen	10.15468/ggcyus	GBIF	3
Lebensraum Walram	10.15468/ykmjru	GBIF	3
Lothian Wildlife Information Centre Secret Garden Survey	10.15468/k8goct	GBIF	3
Macrobenthos and Phytoplankton monitoring in the Belgian coastal zone in the context of the EU Water Framework Directive (WFD)	10.15468/6rk9c3	GBIF	3
Marine Invertebrate from Argentina, Uruguay and Chile	10.15468/xntwha	GBIF	3
Maritimes 4VSW Research Vessel Surveys	10.15468/gw9n44	GBIF	3
Naturpark Kottenforst-Ville 15.6.09	10.15468/m9jzql	GBIF	3
Naturpark Kottenforst-Ville 17.6.09	10.15468/y4sjuo	GBIF	3
Naturpark Kottenforst-Ville Sammelaktion 15.-19.6.	10.15468/xt011t	GBIF	3
Naturschutzgebiet Bausenberg	10.15468/md7w7n	GBIF	3
Northeast Area Monitoring and Assessment Program Near Shore Trawl Survey (NEAMAP)	10.15468/vyglei	GBIF	3
NSW BioNet Atlas	10.15468/14jd9g	GBIF	3
Numérisation des données faune contenues dans les Bulletins de la Société Linnéenne de Bordeaux - Bulletin de la Société Linnéenne de Bordeaux, Tome 136 (N.S) n° 29 (1), 2001 - Données faune	10.15468/7q25t7	GBIF	3
Out on a limb: novel morphology and position on appendages of two new genera and three new species of ectoparasitic isopods (Epicaridea: Dajidae) infesting isopod and decapod hosts	10.5252/zoosystema2021v43a4	GBIF	3
Programme national de science participative sur la Biodiversité Littorale (BioLit)	10.15468/xmv4ik	GBIF	3
Réserves Naturelles de France (RNF) - RNF - Données du Conservatoire d'espaces naturel Centre	10.15468/cuj0gy	GBIF	3

Table B1. Continued

Dataset name	DOI	Source	# Records
Revision of the genera Cyphonethes Verhoeff, 1926 and Titanethes Schioedte, 1849 (Isopoda: Oniscoidea: Trichoniscidae) with a description of a new genus and three new taxa	10.11646/zootaxa.4459.2.3	GBIF	3
RMT Trawl catch from the 1984/85 V5 SIBEX2 voyage	10.15468/qklire	GBIF	3
Schulhof der Astrid-Lindgren-Schule (Elmshorn)	10.15468/ecoaxi	GBIF	3
Seasearch Marine Surveys in Ireland	10.15468/pyugge	GBIF	3
Senckenberg - CeDAMar Resource	10.15468/oc9tsb	GBIF	3
SILENE-FAUNE-PACA - Tour_du_Valat_2017_12_18	10.15468/nnw2xy	GBIF	3
SNH Invertebrate Site Condition Monitoring 2013/14: Cadder Wilderness SSSI	10.15468/ag88wc	GBIF	3
SNH Invertebrate Site Condition Monitoring 2013/14: Cairngorms SSSI	10.15468/v69w8a	GBIF	3
Study on plankton at the port of Ostend in 1965	10.14284/72	GBIF	3
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure AVEN du Grand Voyeux provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/gjji7u	GBIF	3
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste ducourneau philippe provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/kqktar	GBIF	3
Système d'Information sur la Nature et les Paysages d'Ile de France - Données naturalistes hors Île-de-France saisies dans Cettia-idf	10.15468/topfnj	GBIF	3
Three new species of abdominal shrimp parasites (Crustacea: Isopoda: Bopyridae Hemiarthrinae) from the Indo-West Pacific	10.11646/zootaxa.4845.2.7	GBIF	3
Three new species of Tridentella Richardson, 1905 (Isopoda: Cymothoida: Tridentellidae) from New Caledonia	10.11646/zootaxa.4399.1.6	GBIF	3
Two new species of the marine isopod genus Cirolana Leach, 1818 (Crustacea Isopoda: Cirolanidae) from the coast of the western Gulf of Thailand	10.11646/zootaxa.4950.3.3	GBIF	3
Unser kleines Rasenstück/Dürer-Gymnasium Nürnberg	10.15468/onaexb	GBIF	3
Yorkshire Wildlife Trust Shoresearch	10.15468/1nw3ch	GBIF	3
Zukünftiges NSG Höftland/Bockholmwik	10.15468/licmym	GBIF	3
(Table 2) Species density and composition of an inshore and offshore station in Kongsfjord, Svalbard	10.1594/pangaea.807423	GBIF	2
20 Jahre Naturschutzgebiet Dreienberg	10.15468/eujkfo	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
5. Tag der Artenvielfalt: Thema Stadtbiotope	10.15468/rws1pw	GBIF	2
A new species of Aphantolana Moore & Brusca, 2003 (Crustacea: Isopoda Cirolanidae) from the Andaman Islands, northern Indian Ocean	10.11646/zootaxa.4860.4.4	GBIF	2
A new species of Benthana (Crustacea: Isopoda: Philosciidae) from southern Brazil	10.1590/s0101-81752008000200019	GBIF	2
A new species of Pseudione Kossmann, 1881 (Isopoda, Cymothoida, Bopyridae) parasitizing the lobster Nephropsis aculeata Smith, 1881 (Decapoda, Astacidea, Nephropidae) in the Southwestern Atlantic	10.11646/zootaxa.4461.2.5	GBIF	2
Abundance of benthos infauna at station GIK23011-2	10.1594/pangaea.98692	GBIF	2
Abundance of benthos infauna at station POS128/2_281	10.1594/pangaea.98723	GBIF	2
Activité générale du bureau d'étude BIOTOPE (1994 - 2013) - Données acquises dans le cadre de l'activité du bureau d'étude BIOTOPE (1994 - 2013)	10.15468/d3mzxf	GBIF	2
Alter Nördlicher Friedhof (München)	10.15468/mwncvk	GBIF	2
Análisis taxonómicos de macrofauna bentónica para el Plan de Vigilancia Ambiental de las instalaciones de prueba de la sección 3 de Navantia Cartagena	10.15470/cyai9c	GBIF	2
Analysis of macrobenthos in the Southern Bight of the North Sea (1971-1972)	10.14284/208	GBIF	2
Arachnida and Myriapoda (Luomus)	10.15468/sjz32u	GBIF	2
Arcturina serrulatus sp. nov. and a new record of Limnoria rhombipunctata (Crustacea: Malacostraca: Isopoda) from South Korea	10.11646/zootaxa.4286.3.8	GBIF	2
Artenvielfalt der Nordsee - Bremerhaven (Dorum-Neufeld)	10.15468/1nx8rw	GBIF	2
Artenvielfalt im Umfeld der Burgwegschule	10.15468/irih8t	GBIF	2
Artenvielfalt Kreis Gießen	10.15468/3dqrtw	GBIF	2
Avon Baselining - Invertebrates	10.15468/fejuuu	GBIF	2
Bayerische Donau - Blindheim Donaubrücke	10.15468/xyxkih	GBIF	2
BB-Biodiversität b	10.15468/y5jdy2	GBIF	2
Biological observations from the Discovery Investigations 1925-1952	10.15468/qoqbu7	GBIF	2
Biotope entdecken im Kockmecker Siepen (Sauerland)	10.15468/phafd6	GBIF	2
BoBO - Botanic Garden and Botanical Museum Berlin Observations	10.15468/9ll2gz	GBIF	2
Bodentiere rund um die Schule	10.15468/8omfbl	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
British Antarctic (Terra Nova) Expedition, 1910-1913	10.15468/0gsnmz	GBIF	2
Canadian Museum of Nature Parasite Collection	10.15468/khwbzb	GBIF	2
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Monsieur JOURDE Rémi	10.15468/4my9db	GBIF	2
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Monsieur Pratz Jean-Louis	10.15468/gnzy46	GBIF	2
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Pierre DUFRENE	10.15468/fz9vti	GBIF	2
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Zicrona - Parcs nationaux	10.15468/nurqv6	GBIF	2
Cirolana bambang, a distinctive new species of Cirolana Leach, 1818 (Crustacea: Isopoda: Cirolanidae) from Bitung, Indonesia	10.11646/zootaxa.4375.3.10	GBIF	2
CLICNAT- Base de données naturaliste picarde - Données de terrains coproduites par ADEP et CPIE Pays de l'Aisne	10.15468/9c7nsw	GBIF	2
Commissioned surveys and staff surveys and reports for Scottish Wildlife Trust reserves - Unassessed data	10.15468/dfwjgc	GBIF	2
Community analysis and feeding ecology of the ichthyofauna in Gazi Bay sampled in August 1993	10.14284/146	GBIF	2
Deponie Klausdorf	10.15468/ypv7ft	GBIF	2
Die Teiche im Britzer Garten	10.15468/sjwh3n	GBIF	2
Données du Parc national des Pyrénées - Observations occasionnelles Parc national des Pyrénées	10.15468/g3k5au	GBIF	2
Données Faune Base SIRFF - FNE Centre-Val de Loire - Système d’Information Régional sur la Faune et la Flore - FNE Centre-Val de Loire - Données 2015	10.15468/8isgcf	GBIF	2
DONNÉES NATURALISTES ATBI MERCANTOUR HORS PÉRIMÈTRE PARC	10.15468/ps878a	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
Données transmises au PNR Périgord-Limousin n'étant pas commanditées par le parc - Inventaire Faune récolté lors du stage des Curieux de Nature	10.15468/237h7e	GBIF	2
Einen Tag lang Forscher sein - Die 5c der Erich-Kästner-Schule erforscht das Bachemer Wiesental	10.15468/v7vhxz	GBIF	2
Entdeckertour am Muldestausee	10.15468/1sziel	GBIF	2
Fauna inventories from the intertidal zone in Wimereux, France	10.15468/ktfmzh	GBIF	2
Feuchtbiotop Otto-Hahn-Gymnasium	10.15468/cw0rjr	GBIF	2
Feuchtbiotop, Wildtier- und Artenschutzstation Sachsenhagen, Sielmanns Natur-Ranger	10.15468/wkdxvr	GBIF	2
Fortalecimiento de las colecciones de ECOSUR. Primera fase (Zooplancton Chetumal)	10.15468/evh3kd	GBIF	2
Frost Entomological Museum	10.15468/epw1ws	GBIF	2
Garten J. Scherrer (Lachen-Speyerdorf)	10.15468/lalzoo	GBIF	2
Gelände des Schulzentrums am Himmelsbarg	10.15468/z6fkc3	GBIF	2
GEO-Tag der Artenvielfalt auf dem Bausenberg mit den 4. Klassen der Brohltaler Grundschulen	10.15468/camiet	GBIF	2
Gewässer des Wartbergparks Stuttgart (beim Naturlabor der Umweltakademie Baden-Württemberg)	10.15468/nta3gn	GBIF	2
Grünes Germersheim	10.15468/dqxy5g	GBIF	2
Hainhoop - Tonkuhle - Bullenmoor (Arpke)	10.15468/wa8zjl	GBIF	2
Herrensee-Gebiet (Fischbachtal im Odenwald)	10.15468/tay7dt	GBIF	2
Himmelmoor	10.15468/wvbe61	GBIF	2
Hyperbenthos community in the salt marsh of Saeftinghe in 1990 and 1991	10.14284/225	GBIF	2
Insektenvielfalt Ahe/Weichelsee	10.15468/vyg2fw	GBIF	2
Invertebrados Bentónicos de la II y III Expedición Colombia a la Antártica (CCO)	10.15472/jd4g0x	GBIF	2
Kinderbauernhof Pinke-Panke	10.15468/vsng2c	GBIF	2
Königsdorfer Wald	10.15468/efof1h	GBIF	2
KUO Crustacea collections (KUO)	10.15468/sq4vxv	GBIF	2
Landschaftsschutzgebiet Buchhorst 3	10.15468/vjdfov	GBIF	2
Landschaftsschutzgebiet Schmutterwald	10.15468/yxap8q	GBIF	2
Langenberger Forst am Ochsenweg/Niebüll-Leck	10.15468/9gh3xn	GBIF	2
Lebensraum Fluß/Zwickauer Mulde in Wolkenburg	10.15468/wfe2yw	GBIF	2
LK 11 im Mönchspark	10.15468/a7aeqk	GBIF	2
Lüner Holz (Lüneburg)	10.15468/ofocgz	GBIF	2
Macroinvertebrados bentónicos del muro de San Carlos- Zulia- Venezuela	10.15468/878pvc	GBIF	2
Nationalpark Jasmund	10.15468/kaibbk	GBIF	2
Natur aus zweiter Hand am Muldestausee	10.15468/xre7uv	GBIF	2
Naturpark Kottenforst-Ville 16.6.09	10.15468/znhob4	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
New species of Gnathiidae (Crustacea, Isopoda, Cymothoida) from seamounts off northern New Zealand	10.5281/zenodo.2645742	GBIF	2
New species of Xiphoniscus and new record of Androdeloscia escalonai (Isopoda, Scutocoxifera, Oniscidea, Philosciidae) from Brazilian Amazon	10.11646/zootaxa.4350.2.11	GBIF	2
NSG Berschau - Auengebiet (Neustadt/Wied)	10.15468/xzzggm	GBIF	2
Numérisation des données faune contenues dans les Bulletins de la Société Linnéenne de Bordeaux - Bulletin de la Société Linnéenne de Bordeaux, Tome 138 (N.S) n° 31 (4), 2003 - Données faune	10.15468/s3tv5t	GBIF	2
Observaciones de Crustáceos de la Universidad del Valle (CERBMcr-UV)	10.15472/h3uxrl	GBIF	2
Observations naturalistes indépendantes d'origine privée partagées sur la base de données Kollect Nouvelle-Aquitaine - Observations faunistiques de Olivier VANNUCCI	10.15468/duyc5s	GBIF	2
Park Schönfeld (Kassel)	10.15468/bprupn	GBIF	2
Plan de gestion 2015 - 2022 du site de Castelmerle (47) mené par le CEN Nouvelle-Aquitaine - Observations faunistiques (inventaire) réalisées par le CEN Aquitaine	10.15468/t9cqfc	GBIF	2
RACCORDEMENT ELECTRIQUE DE LA FERME EOLIENNE FLOTTANTE DE GROIX ET BELLE-ILE - Création de la liaison sous-marine et souterraine à 63 000 volts - Campagne benthos Rocheux2015-Raccordement du parc éolien Groix/Belle-île	10.15468/hyduh5	GBIF	2
Redescription of Ryukyua circularis (Pillai, 1954) (Isopoda, Cymothoidae), parasite of the Bleeker smoothbelly sardinella Amblygaster clupeoides Bleeker, 1849 from India	10.11646/zootaxa.4526.2.5	GBIF	2
Redescription of the monotypic micro-predatory isopod genera Alitropus H. Milne Edwards, 1840 and Barybrotes Schioedte & Meinert, 1879 (Isopoda, Cymothoida), with a taxonomic key to the Cymothooidea Leach, 1814 from India	10.15468/fmgmuz	GBIF	2
Rohrmeistereiplateau und angrenzendes Gebiet	10.15468/pycurc	GBIF	2
Rückkehr der Biber in Rheinland-Pfalz - Biber in der Primmerbach	10.15468/d0q6t1	GBIF	2
Rund um den Eichwald, Schulhof Friedrich Fröbel Gymnasium - Bad Blankenburg	10.15468/w7nc8k	GBIF	2
Schanzenanlage Bergham	10.15468/ra8276	GBIF	2
Schatzinsel Norderney	10.15468/sfzmol	GBIF	2
Schloß Türnich (Kerpen)	10.15468/kagw2b	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
Schule Sulzbach (Oberegg)	10.15468/2kjl39	GBIF	2
Schulgarten Janusz-Korczak-Realschule	10.15468/mzoije	GBIF	2
Schulgarten Zinnowwald-Grundschule	10.15468/i9i8nt	GBIF	2
Schulgelände Kranich-Gymnasium (Salzgitter)	10.15468/o5blyt	GBIF	2
Schulgelände SGD/Viersen	10.15468/1bojag	GBIF	2
Schulhof A.-Lindgren-Schule (Elmshorn)	10.15468/m4gzrl	GBIF	2
Schulhof der Astrid-Lindgren-Schule Elmshorn	10.15468/oojg7s	GBIF	2
Schulhof der Astrid-Lindgren-Schule und Umgebung (Elmshorn)	10.15468/si3wr9	GBIF	2
Schulwald Sprendlingen	10.15468/pgrmsy	GBIF	2
Schwanner Warte/Kinderhaus St. Elisabeth Waldplatz	10.15468/i9sr9m	GBIF	2
SNH Invertebrate Site Condition Monitoring 2013/14: Ben Lomond SSSI	10.15468/srj4pt	GBIF	2
Southern Maine Community College Gulf of Maine Invertebrate Data	10.15468/v2eq9j	GBIF	2
Species boundaries and phylogeographic patterns in new species of Nannoniscus (Janiroidea: Nannoniscidae) from the equatorial Pacific nodule province inferred from mtDNA and morphology	10.1093/zoolinnean/zlaa174	GBIF	2
Stable isotope ratios of C and N in benthic macrofauna from Mediterranean seagrass litter accumulations from Calvi Bay in 2011-2012	10.14284/454	GBIF	2
Stadt Königs Wusterhausen	10.15468/pwznow	GBIF	2
Stadtpark Herzberg (Elster)	10.15468/bd6ih1	GBIF	2
Steinbruch Haas Stuttgart-Münster	10.15468/mlwzh3	GBIF	2
Streuobstwiese RSG (Cham)	10.15468/xouaoi	GBIF	2
Study of epibenthos and demersal fish in and around the dredging areas of the Belgian Continental Shelf (1977-1981)	10.14284/192	GBIF	2
Study of the biotic environment in the Sluice Dock in relation to oyster farming between 1960 and 1964	10.14284/135	GBIF	2
Sudeniederung (Amt Neuhaus)	10.15468/0jv6c4	GBIF	2
Südpark (Bochum-Wattenscheid)	10.15468/o78meq	GBIF	2
Sukzession Industriebrache	10.15468/hj1piq	GBIF	2
Système d'Information sur la Nature et les Paysages d'Ile de France - Données d'observateurs divers (observateurs transmettant un nombre de données peu élevé) saisies dans Cettia-idf	10.15468/uurom6	GBIF	2
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Maxime Zucca provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/iuwwhy	GBIF	2

Table B1. Continued

Dataset name	DOI	Source	# Records
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du/de la naturaliste Vindras Laurent provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/ofdd4a	GBIF	2
Système d'Information sur la Nature et les Paysages d'Ile de France - Inventaire éclair de Natureparif - Année 2015	10.15468/ongruz	GBIF	2
The Ecology and Biogeography of Heard Island Marine Benthos 1987/88	10.26179/5b62a18cb394e	GBIF	2
The first record of the genus Desertoniscus Verhoeff, 1930 (Isopoda, Oniscidea, Agnaridae) from Europe, with the description of a new species	10.11646/zootaxa.4347.3.10	GBIF	2
Two new species of Atlantoscia Ferrara & Taiti, 1981 (Isopoda: Oniscidea: Philosciidae) from southern Brazil described in the light of integrative taxonomy	10.11646/zootaxa.4482.3.7	GBIF	2
Type material housed in the Carcinological Collection of the Museo de La Plata, Argentina	10.11646/zootaxa.4303.1.5	GBIF	2
Überschwemmungsgebiet der Wied	10.15468/asoob4	GBIF	2
Umgebung der Gesamtschule Hamburg-Winterhude	10.15468/0f5kib	GBIF	2
Umgebung des Spalatin Gymnasium Altenburg	10.15468/u6tjek	GBIF	2
Victorian Biodiversity Atlas	10.15468/khlfs3	GBIF	2
Von Elf bis Elf" Der Botanische Garten Wuppertal	10.15468/ay8iie	GBIF	2
Wald und Wiese am Buchwald	10.15468/szv8az	GBIF	2
Walldorf-Wiesloch: "Natur über den Gleisen"	10.15468/yl8grx	GBIF	2
Woodmeadow Invertebrate Survey 2014	10.15468/iqu35i	GBIF	2
Woodmeadow Invertebrate Survey 2017	10.15468/14uuhu	GBIF	2
Wulfsmuehle/Pinnau	10.15468/qyqlqw	GBIF	2
ZUEC-CRU - Coleção de Crustacea do Museu de Zoologia da UNICAMP	10.15468/ovnu86	GBIF	2
"Biodiversidad en el valle de Cuatro Ciénegas". (Peces)	10.15468/rcpvx0	GBIF	1
"Schule am Inselsee" Güstrow	10.15468/krmiia	GBIF	1
"Schwarzes Teich" (Waldpark Radebeul)	10.15468/9oyqky	GBIF	1
2015_nsmk_smpl_ms	10.15468/szqjn2	GBIF	1
4ème phase d'animation (2018 - 2020) du Document d'objectifs du site Natura 2000 FR7200733 "Coteaux du Boudouyssou et plateau de Lascrozes" et du site Natura 2000 FR7200732 "Coteaux de Thézac et de Montayral" menée par le CEN Aquitaine - Observations faunistiques (inventaire) réalisées par le CEN Aquitaine	10.15468/rbmv44	GBIF	1
6. Tag der Artenvielfalt Hockenheim Thema: Wald 12.-14.6.2009	10.15468/d1ryop	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
A New Abdominally Parasitizing Bopyrid, Anisarthrus okunoi sp. nov. (Crustacea: Isopoda), Infesting the Hinge-Beak Shrimp Rhynchocinetes uritai Kubo, 1942 (Crustacea: Decapoda: Rhynchocinetidae)	10.12782/sd.20.1.037	GBIF	1
A new amphibious troglobitic styloniscid from Brazil (Isopoda, Oniscidea, Synocheta)	10.11646/zootaxa.4294.2.11	GBIF	1
A new species of Bragasellus (Isopoda, Asellidae) from NW Spain, with a key to the known species of the genus	10.11646/zootaxa.4861.2.6	GBIF	1
A new species of Lucasioides Kwon (Isopoda: Oniscidea: Agnaridae) from China	10.5281/zenodo.242379	GBIF	1
A new species of Pseudione Kossmann, 1881 (Crustacea, Isopoda, Bopyridae) parasitizing the squat lobster Munida microphthalma A. Milne-Edwards, 1880 in the Southwestern Atlantic	10.11646/zootaxa.4377.3.7	GBIF	1
A new species of seagrass-boring Limnoria (Limnoriidae, Isopoda, Crustacea) from Japan	10.11646/zootaxa.4232.2.8	GBIF	1
A new species of Syscenus Harger, 1880 (Crustacea: Isopoda: Aegidae) from eastern Australia, with a revised diagnosis of the genus	10.3853/j.0067-1975.49.1997.1261	GBIF	1
A new stygobiotic Stenasellus Dollfus, 1897 (Asellota: Stenasellidae) from Socotra Island, Yemen	10.11646/zootaxa.4683.4.5	GBIF	1
A third species of Aatolana Bruce, 1993 (Crustacea: Isopoda: Cirolanidae)	10.3853/j.0067-1975.50.1998.1272	GBIF	1
Abundance of benthos infauna at station GIK23006-3	10.1594/pangaea.98691	GBIF	1
Abundance of benthos infauna at station GIK23017-1	10.1594/pangaea.98694	GBIF	1
Abundance of benthos infauna at station GIK23040-1	10.1594/pangaea.98700	GBIF	1
Abundance of benthos infauna at station POS128/2_267	10.1594/pangaea.98721	GBIF	1
Abundance of benthos infauna at station POS128/2_276	10.1594/pangaea.98722	GBIF	1
Abundance of benthos infauna at station PS1240-1	10.1594/pangaea.98728	GBIF	1
AKG-Gelände (Bensheim)	10.15468/7tiexg	GBIF	1
AKG-Gelände in Bensheim	10.15468/jm2mld	GBIF	1
Aktion - Friedensburg Oberschule	10.15468/rkx1za	GBIF	1
Alter Kreidebruch Saßnitz/Rügen	10.15468/hnlv1w	GBIF	1
Alter Lagerplatz und Gartenteich (Dortmund/Hörde)	10.15468/pr04pj	GBIF	1
Altholzparzelle Eilenriede Hannover	10.15468/w6gllt	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Análisis taxonómicos de macrofauna bentónica para el Plan de Vigilancia Ambiental de Navantia Cartagena	10.15470/5vopsk	GBIF	1
Animation du Document d’objectifs du site Natura 2000 FR7200799 - Carrières de Castelculier (47) menée par le CEN Nouvelle-Aquitaine - Observations faunistiques (inventaire) réalisées par le CEN Aquitaine	10.15468/ybkf2x	GBIF	1
Animation du Document d’objectifs du site Natura 2000 FR7200799 - Carrières de Castelculier (47) menée par le CEN Nouvelle-Aquitaine - Observations faunistiques (inventaire) réalisées par le CEN Nouvelle-Aquitaine	10.15468/3mkm6m	GBIF	1
Artenvielfalt auf der Wiese	10.15468/rf5hbw	GBIF	1
Artenvielfalt auf Zollverein	10.15468/rdyn0n	GBIF	1
Artenvielfalt des "Grünen Klassenzimmers"	10.15468/7qnqm2	GBIF	1
Artenvielfalt im Beckerbruch (Dessau)	10.15468/i2dupd	GBIF	1
Artenvielfalt in der Kinderakademie im RFZ	10.15468/4n30hf	GBIF	1
Artenvielfalt in der Quälingsbachaue Gladbeck	10.15468/8xapir	GBIF	1
Artenvielfalt in der Stadt: Botanischer Garten Wuppertal und Hardt	10.15468/d3i3px	GBIF	1
Assistance technique à la gestion écologique des espaces naturels propriétés de ValOrizon sur la commune de Damazan (47) menée par le CEN Aquitaine entre 2017 et 2023 - Observations faunistiques (inventaire) réalisées par le CEN Aquitaine	10.15468/ykdh3j	GBIF	1
Assistance technique à la gestion écologique des espaces naturels propriétés de ValOrizon sur la commune de Damazan (47) menée par le CEN Aquitaine entre 2017 et 2023 - Observations faunistiques (inventaire) réalisées par le CEN Nouvelle-Aquitaine	10.15468/pnayz7	GBIF	1
Atlas écologique régional des papillons de jour et zygènes (Lépidoptères) de Midi-Pyrénées - Données de l'Atlas écologique régional des papillons de jour et zygènes	10.15468/yw2sr2	GBIF	1
Australian River Assessment System	10.15468/fwoc93	GBIF	1
Bach	10.15468/dstwtf	GBIF	1
Bachabschnitt der Nette (Osnabrück)	10.15468/hquolv	GBIF	1
Bäche im Mooswald (Vörstetten)	10.15468/jz9xdg	GBIF	1
Baggerseen bei Krauchenwies	10.15468/ltf0ce	GBIF	1
Balkon (Norderstedt)	10.15468/skvmrq	GBIF	1
Bannwald Burghauser Forst	10.15468/rqowlx	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Base BOMBINA du Parc Naturel régional Lorraine - Modernisation des ZNIEFF du PnrL	10.15468/2idi70	GBIF	1
Base de datos de fauna batial, abisopelágica y abisal del Golfo de México	10.15468/gyejjx	GBIF	1
Bayerische Donau - Riedlingen	10.15468/zucgkx	GBIF	1
Bayerische Donau - Tapfheim	10.15468/1zh13s	GBIF	1
Ben Lui NNR invertebrate records compiled from SNH files	10.15468/3sxbdc	GBIF	1
BenthosChukchiFN762_1976_Falk5	10.15468/pdbroz	GBIF	1
Besonderer Ort - besondere Natur: Die Mainzer Zitadelle	10.15468/bybmve	GBIF	1
Binsenwiesen	10.15468/qrp2lf	GBIF	1
Biodiv-Camp Sandmagerrasen in Nürnberg/Langwasser	10.15468/ofndep	GBIF	1
Biodiversitätsdatenbank Nationalpark Hohe Tauern	10.15468/k4qyyw	GBIF	1
Biodiverskripsi: Biodiversity Theses Database	10.15468/cocfqh	GBIF	1
Biologische Station im Kreis Wesel	10.15468/ykb5iv	GBIF	1
Biosphäre Bliesgau	10.15468/ycdxmi	GBIF	1
Biosphärenpark Wienerwald - Pfaffstätten	10.15468/nvvtyn	GBIF	1
Biosphärenpark Wienerwald - Wiener Steinhofgründe	10.15468/xnbfxv	GBIF	1
Biotop Binsenwiesen und Ernst-Reiter-Wiese (Wehrheim/Taunus)	10.15468/fclugs	GBIF	1
Bonner Schülerinnen am Rodder Maar	10.15468/0yzvmr	GBIF	1
Botanischer Garten (Saarbrücken)	10.15468/tcgxfe	GBIF	1
Brander Wald (Stolberg)	10.15468/wzubu0	GBIF	1
Brenz (Heidenheim)	10.15468/nblzxn	GBIF	1
Bulau	10.15468/jmsv5z	GBIF	1
BUND Naturschutzzentrum St. Julian	10.15468/fkjwn2	GBIF	1
Butterberg, Dardesheim	10.15468/uyap2i	GBIF	1
Caecidotea camaxtli (Isopoda: Asellidae) a new species from the Tlaxcala valley, Mexico	10.11646/zootaxa.4624.3.6	GBIF	1
Canberra Nature Map	10.15468/uv6p4z	GBIF	1
Caracterización bionómica de zonas de dragado y de afección de obras en la Ría de Ferrol	10.15470/wlbvov	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de ALONSO Florian	10.15468/bk3kj1	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Maxime ESNAULT	10.15468/dy3tjr	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de NOËL Rémi	10.15468/roynoa	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Pierre NOEL (M2MNHN)	10.15468/lld6x4	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Pierre NOEL (Tatihou)	10.15468/c0lq9u	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Ségolène FAUSTEN	10.15468/zm7huu	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de SWIFT Olivier	10.15468/wsyd2b	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes de Thibault RAMAGE	10.15468/ypmslg	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes d'Océane ROQUINARC'H	10.15468/pxx2hv	GBIF	1
CardObs: Observations naturalistes issues de l'outil de saisie et gestion CardObs mis en place par le Service du Patrimoine Naturel (MNHN)/UMS PatriNat (OFB - CNRS - MNHN) - Données naturalistes du Comité départemental de l'Essonne (CODEP91) de la Fédération Française d'Etudes et de Sports Sous-Marins	10.15468/awzzoh	GBIF	1
Centralisation des données d'études sur le territoire de la Communauté de Communes de la Côte d'Albâtre	10.15468/ttkfwr	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Centre for Environmental Data and Recording (CEDaR) Marine Species Data	10.15468/reat6p	GBIF	1
Churchyards for London	10.15468/iwpzxk	GBIF	1
Citacions biodiversitat Espais Naturals Protecció Especial	10.15470/m5ic1c	GBIF	1
Clare Biological Records Centre Dataset 2004-2007	10.15468/fbb6kb	GBIF	1
CLICNAT- Base de données naturaliste picarde - Données terrain de l'Association des Entomologistes Picards	10.15468/y379s4	GBIF	1
CLICNAT- Base de données naturaliste picarde - Inventaire de terrain sur les ZNIEFF	10.15468/bct7hq	GBIF	1
Colección de Crustáceos Decápodos y Estomatópodos del Centro Oceanográfico de Cádiz (CCDE-IEOCD)	10.15468/anztjy	GBIF	1
Comparative study of the organismic assemblages associated with the demosponge Sarcotragus foetidus Schmidt, 1862 in the coasts of Cyprus and Greece	10.15468/tmvw8g	GBIF	1
Conchological Society of Great Britain & Ireland: marine mollusc records	10.15468/aurwcz	GBIF	1
Convention Fédération française de golf - MNHN 2016-2019	10.15468/zsxuss	GBIF	1
DASSH Data Archive Centre volunteer sightings records	10.15468/xwiw3h	GBIF	1
Description of new species of algal-boring Limnoria (Crustacea, Isopoda, Limnoriidae) from Japan and redescription of Limnoria segnoides Menzies, 1957 and L. nagatai Nunomura, 2012	10.11646/zootaxa.4550.2.5	GBIF	1
Données d'occurrences issues des Formulaires standards de données des sites Natura 2000 - Données d'occurrence Espèces issues de la base Natura 2000: espèces d'intérêt communautaire et autres espèces remarquables renseignées dans les FSD	10.15468/g2ptuw	GBIF	1
Données Faune de l'Agence des Espaces Verts (AEV) d'Ile de France	10.15468/o3ukgd	GBIF	1
Données sur les Invertébrés aquatiques de la Réserve Naturelle des Marais de Bruges - Inventaire Entomologiques	10.15468/qysjd3	GBIF	1
Düne am Ulvenberg (Darmstadt)	10.15468/c3bkkm	GBIF	1
Düpenauwiesen	10.15468/5nvyjt	GBIF	1
Early succession in benthic hard bottom communities in Kongsfjorden, Svalbard - abundance	10.1594/pangaea.351152	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Ecological study of the plankton in the port of Ostend in 1965	10.14284/194	GBIF	1
Epibenthos and demersal fish monitoring in function of dredge disposal monitoring in the Belgian part of the North Sea	10.14284/198	GBIF	1
Epifauna community at Waarde and Saeftinghe (Westerschelde) in 1991	10.14284/224	GBIF	1
Erft in Selikum (Neuss)	10.15468/ylvphz	GBIF	1
Erlengraben/Lipp-Tal (Östringen)	10.15468/95qyg8	GBIF	1
Estudio de la comunidad zooplanctónica y fitoplanctónica en los ecosistemas de arrecifes coralinos mesofóticos del Parque Nacional Corales de Profundidad	10.15472/p70rj0	GBIF	1
Estudio de la fauna edáfica en una selva baja inundable de la Reserva de la biósfera de Sian Ka'an Quintana Roo	10.15468/t2wsln	GBIF	1
Evaluation of the effect of disposal of dredging material on macrobenthos communities in the Maas plain (1988)	10.14284/196	GBIF	1
Expedition "Schulgelände"	10.15468/ewklow	GBIF	1
Faberpark (Nürnberg/Stein)	10.15468/ciwsd1	GBIF	1
Fauna and flora inventories (terrestrial and limnetic) from the South of Belgium	10.15468/dwlarm	GBIF	1
FBIP: Actinopterygii and Elasmobranchii occurrence record throughout South Africa	10.15468/zvvx7d	GBIF	1
Feriendorf des Kreises Gedern (Ober-Seemen)	10.15468/iqhlfj	GBIF	1
Feriendorf Ober-Seemen	10.15468/s7neas	GBIF	1
Feuchtwiese in Langes Tannen (LMS), Klasse 5c	10.15468/ytvuil	GBIF	1
Feuerlöschteich, Wald und Dünen in den Holmer Sandbergen	10.15468/vn9uhn	GBIF	1
FFH-Gebiet Ahrbachtal	10.15468/joupjm	GBIF	1
FFH-Gebiet Klosterwasser/Burkau	10.15468/vpfqb1	GBIF	1
Fife Nature Records Centre combined dataset 2018/19	10.15468/qbg6jf	GBIF	1
Fledermaus	10.15468/ltou6r	GBIF	1
Fluss - Vielfalt	10.15468/ucrjle	GBIF	1
Förderzentrum Schmölln	10.15468/ezpgvd	GBIF	1
Freiburger Tag der Artenvielfalt	10.15468/os5bjq	GBIF	1
Freiheitsring (Frechen)	10.15468/mn7p0n	GBIF	1
Fuldaaue (Stadtgebiet Fulda)	10.15468/veh6ha	GBIF	1
Fürstenberger Ralley Teil 3	10.15468/yv2p5b	GBIF	1
Garten Hamburg Uhlenhorst	10.15468/d60qys	GBIF	1
Gelände der Lahntalschule Biedenkopf und Lahnauen	10.15468/u3sabf	GBIF	1
GEO Hauptveranstaltung Tirol (Innsbruck)	10.15468/n3uph3	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Geo-Tag der Artenvielfalt Süßen Hornwiesen-Grundschule	10.15468/5jnyjo	GBIF	1
Gewann Krampf (Heilbronn)	10.15468/crg9hw	GBIF	1
Goethe-Hauptschule/Projekt I-10	10.15468/xaikey	GBIF	1
Gronau - auf der Suche nach dem Neunauge	10.15468/70foai	GBIF	1
Grundschüler erkunden Schulumgebung	10.15468/go2d9k	GBIF	1
Grundwasserlebensraum im Englischen Garten (München)	10.15468/yso2gm	GBIF	1
Gunma Museum of Natural History, Crustacea Specimens	10.15468/is8pqb	GBIF	1
Gurgltal (Tarrenz)	10.15468/tjcduh	GBIF	1
Gymnicher Mühle	10.15468/2r1rhj	GBIF	1
Haarbach Höfe	10.15468/pd4wxp	GBIF	1
Hache im Ellernbruch (Sudweyhe/Weyhe)	10.15468/qd8niy	GBIF	1
Hainbachtal in Oelsnitz/V.	10.15468/o6svf3	GBIF	1
Hamberger Brücke/Würmtal (Pforzheim)	10.15468/wubqak	GBIF	1
Heider Bergsee (Brühl)	10.15468/pochpz	GBIF	1
Hintere Halde	10.15468/kqbuzy	GBIF	1
Hüttenseepark (Meißendorf)	10.15468/wxmbeu	GBIF	1
Im Bauerngarten	10.15468/favyae	GBIF	1
Informe "Control de Organismos" en la Ría de Ferrol 2019	10.15470/m58paq	GBIF	1
Innenstadt Göttingen - Natur Zuhause	10.15468/m4edff	GBIF	1
Integrierte Gesamtschule Flensburg Wiemoosgraben	10.15468/s7xzec	GBIF	1
Inventaire biodiversité du site Pierre Fabre d'Aignan	10.15468/483gdr	GBIF	1
Inventaire de la Réserve Naturelle de l'étang de Cousseau - Inventaire Entomologiques	10.15468/6zsxe5	GBIF	1
Inventaire de la Réserve Naturelle des Dunes et Marais d'Hourtin - Inventaire entomologique	10.15468/4geman	GBIF	1
Inventaire de la réserve naturelle géologique de Saucats - La Brède - Etude sur les protocoles I2M2	10.15468/7c8jvm	GBIF	1
Inventaires naturalistes du Service du Patrimoine naturel/UMS PatriNat - Inventaire de la Forêt de la Commanderie, Fontainebleau	10.15468/sut6xp	GBIF	1
Inventaires naturalistes du Service du Patrimoine naturel/UMS PatriNat - Inventaire de l'îlot du Lédénez Vraz	10.15468/us69wq	GBIF	1
Invertebrate Paleontology Division, Yale Peabody Museum	10.15468/nqheui	GBIF	1
Invertebrates compiled by W.Block	10.15468/5kbwve	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Jeu de données convention Saint-Gobain Distribution Bâtiment France -SPN-MNHN - Données Saint-Gobain Distribution Bâtiment France	10.15468/lqbbsv	GBIF	1
Kabelskebach (Kabelsketal, Saalkreis)	10.15468/ldebcg	GBIF	1
Kaniswall/Gosener Wiesen an der Spree	10.15468/xim1hm	GBIF	1
Kaulsdorf	10.15468/gzxkhu	GBIF	1
Kenai National Wildlife Refuge, Alaska (KNWR) Insect specimens (Arctos)	10.15468/xwl4xi	GBIF	1
Kiesgruben Wemb	10.15468/gdfc3l	GBIF	1
Kinder- und Jugendferiendorf des Kreises Groß-Gerau - Gedern/Ober-Seemen	10.15468/bzyrco	GBIF	1
Kindergarten	10.15468/kt0jmy	GBIF	1
Kindervilla Aussengelände/Hiltroper Park	10.15468/ro3ihb	GBIF	1
Kinderwald Hannover	10.15468/axqxjx	GBIF	1
Kita-Wäldchen Fuchsturmweg Jena	10.15468/t5kqlo	GBIF	1
Klasse 3a	10.15468/khe2si	GBIF	1
Klassenfahrt Usedom/Wald und Küste in Zinnowitz	10.15468/4zpveo	GBIF	1
Knechtweide (Kohlfurth)	10.15468/s3qrze	GBIF	1
Kochertgraben II	10.15468/atgawc	GBIF	1
Kohlbach (Sulzfeld)	10.15468/y1slhp	GBIF	1
Königstetten	10.15468/vkomyx	GBIF	1
Kremmer Luch	10.15468/qme6rj	GBIF	1
Kühnauer See (Dessau)	10.15468/buaolb	GBIF	1
Kurler Busch (Scharnhorst)	10.15468/byhdai	GBIF	1
Küste Wismar-Wendorf bis Hoben	10.15468/yptgzr	GBIF	1
LACM Rancho La Brea	10.15468/zdn495	GBIF	1
Landschaftspark St.Leonhard-Deisendorf	10.15468/etporu	GBIF	1
Landschaftsschutzgebiet Buchhorst 4	10.15468/hlq8fu	GBIF	1
Laubenheimer Bodenheimer Ried - von Stromtalwiesen und Flutrasen	10.15468/xnfqki	GBIF	1
Laubwald Dreiländereck (Aachen/Vaals[NL])	10.15468/tyi8lf	GBIF	1
Lebensraum Stadt und Park	10.15468/xink53	GBIF	1
Leipanthura casuarina, new genus and species of anthurid isopod from Australian coral reefs without a " five-petalled " tail (Isopoda, Cymothoida, Anthuroidea)	10.3897/zookeys.18.198	GBIF	1
Liether Park (LMS), 5a	10.15468/m0bl0n	GBIF	1
Liether Park (LMS), 6c	10.15468/ncwafn	GBIF	1
Liether Park 2 (LMS), Klasse 6c	10.15468/dpjmis	GBIF	1
LifeWatch observatory data: reference collection of unique observations in the Belgian Part of the North Sea	10.14284/267	GBIF	1
Lillachtal mit Kalktuffquelle bei Weißenohe	10.15468/vehehm	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Lindau im Bodensee	10.15468/vimum3	GBIF	1
Listhof und Umgebung	10.15468/wfxej0	GBIF	1
Luch Niederlehme, Schüler der Klasse 7	10.15468/y6scjf	GBIF	1
Lustadter Wald	10.15468/vep1yx	GBIF	1
Macrobenthos monitoring at long-term monitoring stations in the Belgian part of the North Sea between 1979 and 1999	10.14284/201	GBIF	1
Macrobenthos monitoring in function of dredge disposal monitoring in the Belgian part of the North Sea	10.14284/200	GBIF	1
Macrobenthos of the Western Scheldt estuary in September 1978	10.14284/131	GBIF	1
Macrobenthos: temporal patterns for stations 115b and 330 in the Belgian Part of the North Sea	10.14284/523	GBIF	1
Macrobentos de cuatro playas de alta energía ubicadas en la Península de La Guajira, noroeste del Golfo de Venezuela	10.15468/c73cdd	GBIF	1
Macrozoobenthos, Joint Open Sea Surveys August 2017, EMBLAS-II	10.15468/pt6cvw	GBIF	1
Mangfalltal	10.15468/fdbcji	GBIF	1
Marine Data from The Wildlife Trusts (TWT) Dive Team; 2014-2018	10.15468/aqr7zv	GBIF	1
Marine Invertebrate Diversity Initiative (OBIS Canada)	10.15468/jr2dvh	GBIF	1
Marine Non Native Species records from Natural Resources Wales (NRW) Monitoring Research and Ad-hoc Sightings	10.15468/jc9uj9	GBIF	1
Mit allen Sinnen durch den Wald/Schmücke (ev. Kita Heldrungen)	10.15468/a3kvgq	GBIF	1
Mittelriede Höhe Gliesmarode-Braunschweig	10.15468/afyoe9	GBIF	1
Mühlenbach bei Buxtehude	10.15468/0gbfbz	GBIF	1
NABU Naturschutzhof Netttetal (Sassenfeld) e.V.	10.15468/78wbnu	GBIF	1
Natur-Erlebnisgebiet der Naturschutz-Akademie Hessen und Umgebung	10.15468/1nwayh	GBIF	1
NatureShare	10.15468/4cqg2v	GBIF	1
Naturnachmittag 'Artenvielfalt an der Ecke' (Wäldchen an der Wegegabelung)	10.15468/5niduz	GBIF	1
Naturpark Drömling	10.15468/9b8ujb	GBIF	1
Naturpark Kottenforst-Ville 18.6.09	10.15468/agfdpb	GBIF	1
Naturpark Kottenforst-Ville 19.6.09	10.15468/qozlib	GBIF	1
Naturschutzgebiet Börstig bei Hallstadt	10.15468/uptda2	GBIF	1
Naturschutzgebiet Lippeaue (Marl) - Pfadis in Sickingmühle	10.15468/spzlfb	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
New and little-known species of isopods (Crustacea, Isopoda) from the eastern Mediterranean	10.11646/zootaxa.4311.2.1	GBIF	1
New species of Sargassum-boring Limnoria Leach, 1814 (Crustacea, Isopoda Limnoriidae) from Japan	10.11646/zootaxa.4970.1.4	GBIF	1
NHMD Entomology Collection	10.15468/nnobcm	GBIF	1
Nottekanal, Klasse 7 - 10	10.15468/dwwqx8	GBIF	1
NSG Dellwiger Wald, Dortmund	10.15468/c5itv4	GBIF	1
NW-Innenhof Gesamtschule Herten 7.6.2001	10.15468/xq2ygh	GBIF	1
Ober-Olmer Wald	10.15468/6zsivw	GBIF	1
Ober-Olmer Wald 09	10.15468/zag8aq	GBIF	1
Observations naturalistes hors étude réalisées par les organismes utilisant la base de données Kollect Nouvelle-Aquitaine - Observations faunistiques hors étude réalisées par le CEN Aquitaine	10.15468/qa3kq2	GBIF	1
Observations naturalistes hors étude réalisées par les organismes utilisant la base de données Kollect Nouvelle-Aquitaine - Observations faunistiques hors étude réalisées par le CEN Nouvelle-Aquitaine	10.15468/fs23ut	GBIF	1
Occurrences de vecteurs de maladies recensées à l'Hôpital de Mènontin	10.15468/wpqlgi	GBIF	1
Olympiapark (München)	10.15468/hlrd2v	GBIF	1
Örtzemündung (Stedden)	10.15468/rjrhsu	GBIF	1
Ostfriesland	10.15468/1uvbst	GBIF	1
Participation aux politiques publiques départementales de l'environnement - Données CEN M-P départements	10.15468/agctst	GBIF	1
Paul-Gerhardt-Schule Dassel	10.15468/bza0nc	GBIF	1
Plan de gestion 2015 - 2019 du site du Coteau de Casserouge (47) mené par le CEN Aquitaine - Observations faunistiques (inventaire) réalisées par le CEN Aquitaine	10.15468/2shkx5	GBIF	1
Plan de gestion 2016 - 2020 du site du domaine de Rodié (47) mené par le CEN Nouvelle-Aquitaine - Observations faunistiques fortuites réalisées par le CEN Aquitaine	10.15468/7qryh4	GBIF	1
Plan régional d'actions en faveur des odonates (PRAO): 3ème phase (2017 - 2018) menée par le CEN Aquitaine - Observations faunistiques fortuites réalisées par le CEN Aquitaine (Étude Agrion de Mercure)	10.15468/ndeyf6	GBIF	1
Plymouth sound dataset. Soft sediment macrobenthos from the Plymouth Sound from 1995	10.14284/297	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Pottundkopp	10.15468/0tuomy	GBIF	1
Priest Pot species list, Cumbria, Britain	10.15468/lih6qc	GBIF	1
Programa Poseidon - Citizen Science Project Results	10.14284/470	GBIF	1
Quellgebiet Flossach - Klassen 4 a und 4 b VS Tussenhausen	10.15468/getpvg	GBIF	1
RACCORDEMENT ELECTRIQUE DE LA FERME EOLIENNE FLOTTANTE DE GROIX ET BELLE-ILE - Création de la liaison sous-marine et souterraine à 63 000 volts - Campagne benthos Intertidal 2017-Raccordement du parc éolien Groix/Belle-île	10.15468/txjdnf	GBIF	1
RACCORDEMENT ELECTRIQUE DE LA FERME EOLIENNE FLOTTANTE DE GROIX ET BELLE-ILE - Création de la liaison sous-marine et souterraine à 63 000 volts - Campagne benthos subtidal Drague2015-Raccordement du parc éolien Groix/Belle-île	10.15468/ypabzj	GBIF	1
Réalisation du dossier d'avant-projet pour l'extension de la Réserve Naturelle Nationale des Marais d'Yves (17) - Inventaires endofaune benthique	10.15468/fcbqub	GBIF	1
Regionalpark(Hattersheim)	10.15468/whyljk	GBIF	1
Reifrocknarzissenwiese Löcknitz	10.15468/ofnwua	GBIF	1
Renaturierung Werse (Innenbereich Beckum)	10.15468/35acb1	GBIF	1
Reusaer Wald	10.15468/cvdzbg	GBIF	1
Revision of Pleuroprion zur Strassen, 1903 (Holidoteidae) and re-evaluation of Spectrarcturus Schultz, 1981 (Arcturidae) (Crustacea, Isopoda, Valvifera)	10.11646/zootaxa.4894.1.1	GBIF	1
Rhopalione kali sp. nov., first known epicaridean parasite on the Malaysian pinnotherid crab, Serenotheres besutensis (Serène, 1967)	10.11646/zootaxa.4590.2.5	GBIF	1
Riedensee	10.15468/keh8mk	GBIF	1
Riedkanal Bötzingen	10.15468/hqacia	GBIF	1
Riekdahler Wiesen	10.15468/bh4yyq	GBIF	1
Rössewiesen am Krumbholz	10.15468/wc0v5b	GBIF	1
Roter Berg Werdau (Leubnitz)	10.15468/ql4gzj	GBIF	1
Rotes Steigle (Panzerübungplatz Böblingen)	10.15468/8umtlw	GBIF	1
Rund um das LUGY	10.15468/7y7miq	GBIF	1
Rund um den Hainbergsee	10.15468/ybncmd	GBIF	1
Rund ums Schulgelände	10.15468/m7hcfz	GBIF	1
Rur	10.15468/wjl9jr	GBIF	1
Sahrbachtal Kreis Ahrweiler	10.15468/g5yljv	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Saisie de données naturalistes d'observateurs indépendants sur la plateforme de l'Observatoire FAUNA - Données naturalistes de Annie JUGLAS	10.15468/fwjyyu	GBIF	1
Saisie de données naturalistes d'observateurs indépendants sur la plateforme de l'Observatoire FAUNA - Données naturalistes de Patrice ROBISSON	10.15468/ez7gdr	GBIF	1
sarce_rockyshores	10.15468/1rdkla	GBIF	1
Schriesheimer Steinbruch	10.15468/vslar7	GBIF	1
schulgarten	10.15468/hjgkc0	GBIF	1
Schulgarten der Volksschule	10.15468/jszlxx	GBIF	1
Schulgarten Huttenheim (Philippsburg/Baden)	10.15468/n4izks	GBIF	1
Schulgarten Zinnowwald-GS	10.15468/nceu99	GBIF	1
Schulgarten-St.-Georg-Gymnasium	10.15468/snlxbk	GBIF	1
Schulgelände Ceciliengymnasium	10.15468/f5ebe2	GBIF	1
Schulgelände des Schulzentrums am Himmelsbarg	10.15468/wcc5cm	GBIF	1
Schulgelände Dientzenhofer-Gymnasium (Bamberg)	10.15468/n8ra9q	GBIF	1
Schulgelände Gebrüder-Grimm-Schuleund Umgebung (Lingen)	10.15468/oymewb	GBIF	1
Schulgelände Grolland	10.15468/gl3obj	GBIF	1
Schulgelände IGS-Frosch (Thaleischweiler-Fröschen)	10.15468/thdaca	GBIF	1
Schulgelände Paul-Gerhardt-Schule-Dassel	10.15468/jqoapo	GBIF	1
Schulgelände Regelschule Gorndorf/Umgebung Jugend- und Stadtteilzentrum Gorndorf	10.15468/xou4co	GBIF	1
Schulgelände Schule auf der Aue, Münster	10.15468/dft3sz	GBIF	1
Schulhof (Bad Waldsee)	10.15468/ehvsnk	GBIF	1
Schulhof Bühlschule Giengen	10.15468/eqx5od	GBIF	1
Schulhof und Anlagensee in Nellingen	10.15468/rsrpkj	GBIF	1
Schulhofuntersuchung Thomas-Mann-OS	10.15468/pyokcv	GBIF	1
Schulteich Heinrich-Mann-Schule	10.15468/geh9u3	GBIF	1
Schulumgebung Grüterschule, Rheine	10.15468/wixcfp	GBIF	1
Schulwald Grundschule Brügge	10.15468/6h3vcs	GBIF	1
Schulwald Marksuhl	10.15468/whsh4e	GBIF	1
Selz (Ingelheim am Rhein)	10.15468/vxrf7h	GBIF	1
Shellfish (MNHM-MS)	10.15468/6vocgk	GBIF	1
SILENE-FAUNE-PACA - Parc_National_des_Ecrins_2017_12_18	10.15468/0ojuzb	GBIF	1
South Caribbean Diversity	10.15468/xeray1	GBIF	1
Stadtpark Schmölln	10.15468/pxinrx	GBIF	1
Stadtpark Sulzbach-Rosenberg	10.15468/09rvrb	GBIF	1
Standing water cartography, Recorder-Lux database	10.15468/fa8neg	GBIF	1
Stausee (Oberdigisheim/Meßstetten)	10.15468/bt6ibb	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Steinbachtal (Würzburg)	10.15468/yjxrvo	GBIF	1
Steinbruch Pluwig	10.15468/l1bddk	GBIF	1
Sternwiese Mülheim-Broich	10.15468/9bbtnf	GBIF	1
Streuobstwiese	10.15468/jfn5os	GBIF	1
Streuobstwiese Kattenhund (Schleswig)	10.15468/qacgll	GBIF	1
Streuobstwiese Kugelberg (Ulm)	10.15468/ylhovj	GBIF	1
Study of epibenthos and demersal fish of the titanium dioxide discharge area in the Dutch Continental Shelf (1976-1981)	10.14284/241	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la naturaliste Chloé Chabert	10.15468/yf9vqs	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure ANCA (les Amis Naturalistes des Coteaux d'Avron) provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/ufcrtq	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure Département 77 provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/vezows	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure Seine-et-Marne Environnement provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/nlto1y	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données de la structure SFO - Société Française d'Odonatologie provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/agfbob	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Grégoire Loïs	10.15468/wzuyl3	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste JAPIOT Xavier provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/u0lu0h	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Larregle Guillaume provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/mlqyne	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du naturaliste Thierry Roy provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/1wyctb	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du Parc Naturel Régional du Vexin provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/5uol10	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Données du/de la naturaliste D'HINZELIN MARCEL provenant de la base de donnée du SINP Île-de-France CETTIA	10.15468/pjmdcu	GBIF	1
Système d'Information sur la Nature et les Paysages d'Ile de France - Inventaire éclair de Natureparif 2014	10.15468/mqmy8i	GBIF	1
Tag der Artenvielfalt am Bruckenwasen	10.15468/a7evqr	GBIF	1
Tag der Artenvielfalt im Taubental	10.15468/ge5em2	GBIF	1
Tag der Artenvielfalt mit SchülerInnen des Europa-Gymnasiums in Wörth am Rhein	10.15468/bfate1	GBIF	1
Tauchaktion	10.15468/rmsvxw	GBIF	1
Tauchen und Meer	10.15468/ve7eov	GBIF	1
Tauchen und Meer 02	10.15468/utfmvr	GBIF	1
Taxonomic revision of Brasiloniscus (Oniscidea, Pudeoniscidae) with description of a new species	10.5852/ejt.2018.434	GBIF	1
TBW-Schafberg	10.15468/swvmoa	GBIF	1
The first Turcolana Argano & Pesce, 1980 (Isopoda: Cirolanidae) from the Greek mainland	10.11646/zootaxa.4170.1.6	GBIF	1
The fishery ground near Alexandria. XXI. Tanaidacea and Isopoda by H.J. Larwood (1940).Notes and Memoirs No35.	10.15468/1w5yeb	GBIF	1
Three new species of Scyracepon Tattersall, 1905 (Isopoda: Bopyridae) from Pacific islands, with comments on the rarity of bopyrids parasitizing brachyurans	10.11646/zootaxa.4851.1.6	GBIF	1
Tiere und Pflanzen um uns herum!	10.15468/nbirph	GBIF	1
Tiergarten Straubing	10.15468/zpadpe	GBIF	1
Triebesbach (Zeulenroda-Triebes)	10.15468/3zqivb	GBIF	1
Uferzone Wipper (Biesenrode)	10.15468/ceibjl	GBIF	1
Umgebung der Gesamtschule Winterhude (Hamburg)	10.15468/dmw3vl	GBIF	1
Umgebung der Grundschule Oderberg	10.15468/k1ap02	GBIF	1
University of Alberta Freshwater Invertebrate Collection (UAFIC)	10.18165/ryex9i	GBIF	1
University of Texas, Biodiversity Center, Entomology Collection (UTIC)	10.15468/sanyq7	GBIF	1
Unser Schulhof	10.15468/cyeusg	GBIF	1
Unter hellen Zinnen und finsteren Grotten	10.15468/mzruxi	GBIF	1
Urwald 2 (Bad Waldsee)	10.15468/xo6205	GBIF	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Vergleich der Fauna eines naturbelassenen mit einem wasserwirtschaftlich veränderten Gewässer	10.15468/ud5tqe	GBIF	1
Waldränder der Frankenhöhe (Rothenburg ob der Tauber)	10.15468/nmjchu	GBIF	1
Waldstück am Schullandheim Bad Bederkesa	10.15468/sdiz8h	GBIF	1
Waldwandel in Monschau-Mützenich-Boverei	10.15468/t2pfbb	GBIF	1
Wassermann	10.15468/mwqkjg	GBIF	1
Weidenhüttendorf an der Würm (München)	10.15468/tkpq49	GBIF	1
Weinberge und angrenzende Felsflächen (Drieschen) in Hatzenport/Terrassenmosel	10.15468/xqwqxm	GBIF	1
Werl macht sich auf die Suche	10.15468/eoamwt	GBIF	1
Werremündung im Schwarzatal	10.15468/ihzazs	GBIF	1
Wildes Bremer Leben im Park	10.15468/52td5o	GBIF	1
Wirbach	10.15468/mipz3i	GBIF	1
Wupperaue bei Kemna (Wuppertal)	10.15468/eu9jpa	GBIF	1
Zielbach (Töll)	10.15468/q2gkol	GBIF	1
Zoo Frankfurt	10.15468/phakpd	GBIF	1
RSMP Baseline Dataset	10.14466/CefasDataHub.34	OBIS	6,858
SHARK - National zoobenthos monitoring in Sweden since 1971	10.15468/fggzdr	OBIS	3,599
Marine Recorder Snapshot extract of surveys entered by JNCC	10.17031/mehqrq	OBIS	3,016
SHARK - Regional monitoring, recipient control and monitoring projects of zoobenthos in Sweden since 1972	10.15468/cesssx	OBIS	2,797
Marine Recorder Snapshot extract of surveys entered by Natural England	10.17031/thn0xd	OBIS	1,550
Marine Recorder Snapshot extract of surveys entered by NRW	10.17031/b3efts	OBIS	1,497
Bigood	-	OBIS	1,212
Marine Recorder Snapshot extract of surveys entered by National Museums Northern Ireland (NMNI)	10.17031/frdvov	OBIS	1,009
SHARK_Epibenthos_2015_DEEP_Asko_version_2017-04-26.zip	-	OBIS	948
Marine Recorder Snapshot extract of surveys entered by The archive for marine species and habitats data (DASSH)	10.17031/myrqac	OBIS	921
POHJE database	-	OBIS	913
NIWA Invertebrate Collection	-	OBIS	663
Marine Recorder Snapshot extract of surveys entered by NatureScot	10.17031/pqhlyg	OBIS	657
SHARK_Epibenthos_2010_AQBI_Sodermanland_version_2017-04-26.zip	-	OBIS	478

Table B1. Continued

Dataset name	DOI	Source	# Records
Macrobenthos from the eastern English Channel in 1999 and 2001	-	OBIS	387
North Atlantic and Arctic Isopoda sampled during the IceAGE project	-	OBIS	384
DFO Central and Arctic Multi-species Stock Assessment Surveys	-	OBIS	362
COLEÇÃO DE CARCINO DO MUSEU NACIONAL (CARCINO:MNRJ)	-	OBIS	329
Discovery Expedition Biological Reports	-	OBIS	252
North Atlantic and Arctic Isopoda sampled during the BIOICE project	-	OBIS	252
NOAA National Benthic Inventory	-	OBIS	227
Atlantic Reference Centre	-	OBIS	190
Marine Recorder Snapshot extract of surveys entered by SeaSearch	10.17031/yq0gbg	OBIS	182
Biodiversity of benthic assemblages on the Arctic continental shelf: historical data from Canada (1955 to 1977)	-	OBIS	173
DFO Quebec Region MLI museum collection	-	OBIS	169
Irish Benthos monitoring as part of the Water framework directive since 2012	-	OBIS	168
Benthic Haploniscidae (Isopoda) collected around Iceland during the BIOICE, IceAGE, IceAGE2,IceAGE_RR and IceAGE3 expedtions in 1992-2020	10.14284/551	OBIS	157
Dutch long term monitoring of macrobenthos in the Dutch Continental Economical Zone of the North Sea	-	OBIS	155
1778-1998 Ivor Rees North Wales Marine Fauna Ad-hoc sightings shore and ship-based surveys	10.17031/35prlf	OBIS	128
Royal Belgian Institute of Natural Sciences Crustacea Collection	-	OBIS	118
University of Florida Museum of Natural History Invertebrate Zoology	-	OBIS	113
MEDITS-Spain: Demersal and mega-benthic species from the MEDITS (Mediterranean International Trawl Survey) project on the Spanish continental shelf between 1994 and 2010	-	OBIS	112
QUADRIGE - Coastal monitoring database and products, 1974 onwards. (6064)	-	OBIS	111
British Antarctic (Terra Nova) Expedition Zoology	-	OBIS	101
Marine Recorder Snapshot extract of surveys entered by Kent Wildlife Trust	10.17031/rkwbds	OBIS	101
HELCOM/OSPAR Ballast water observations	-	OBIS	99
Bishop Museum Marine Invertebrates Specimens	-	OBIS	96

Table B1. Continued

Dataset name	DOI	Source	# Records
Marine Invertebrata specimen database of Osaka Museum of Natutal History	10.15468/zhubgk	OBIS	83
DFO Zoobenthos data from upper Frobisher Bay, 1967 to 1973	-	OBIS	67
Bay of Puck dataset	-	OBIS	52
MBIS Marine Fauna and Flora observations around New Zealand	-	OBIS	51
iNaturalist research-grade observations	-	OBIS	49
Macrobenthos monitoring in function of aggregate extraction activities in the Belgian part of the North Sea	10.14284/199	OBIS	49
Roscoff inventories: marine fauna and flora since 1800	10.21411/qhtc-a855	OBIS	49
Sizing ocean giants: patterns of intraspecific size variation in marine megafauna	-	OBIS	49
Abundance and biomass of infaunal species as part of Essential fish habitat surveys, Co. Down Coast (Northern Ireland) 2012-2013	10.17031/wykk77	OBIS	47
2014 Centre for the Environment, Fisheries and Aquaculture Science (Cefas) Farnes East recommended Marine Conservation Zone (rMCZ) Seabed survey Update	-	OBIS	43
Biomôr 1 dataset. Benthic data from the Southern Irish Sea from 1989-1991	-	OBIS	38
2012-2015 Orkney Islands Council Marine Non-Native Species Monitoring Programme	10.17031/jnfcip	OBIS	33
Bay of Fundy Species List	-	OBIS	33
SEFSC CAGES Alabama Fish length Data with CPUE	-	OBIS	32
2012-2013 University of Plymouth Falmouth maerl bed infauna and sediment survey using diver cores	10.17031/1710	OBIS	30
Soviet Antarctic Expedition 1956-1958	-	OBIS	29
Explore Your Shore	10.14284/563	OBIS	28
Ocean Genome Legacy Collection	-	OBIS	27
Colección de Artropodos del Museo de Historia Natural Marina de Colombia - Makuriwa	10.15472/eateut	OBIS	26
Macrobenthos samples collected in the Scottish waters in 2001	-	OBIS	25
Infaunal abundances from mud samples taken from the Outer Ards penninsula (Northern Ireland) in 2014 and 2016 as part of an assessment of the Modiolus modiolus reefs	-	OBIS	24
Mytilini	-	OBIS	24
Species list recorded by baited cameras at deep sea area in Japan	10.48518/00007	OBIS	24

Table B1. Continued

Dataset name	DOI	Source	# Records
Survey data of tidal flats on the Monitoring sites 1000 project, BDCJ	-	OBIS	24
Benthic fauna collected in the Arrábida Marine Protected Area (SW Portugal) from 2007 to 2009	10.14284/461	OBIS	21
FRB: Bottom fauna of Saint John Harbour and estuary as surveyed in 1959 and 1961	-	OBIS	21
Plankton&BenthosResearch	-	OBIS	20
Macrobenthos Data from Shoreham, the Tyne and the Thames Estuaries, UK, 2000 to 2006	10.14466/CefasDataHub.45	OBIS	19
Macrozoobenthos_sand_Coconet_IBER-BAS	-	OBIS	19
Cobscook Bay Inventory: A Historical Checklist of Marine Invertebrates Spanning 162 Years	-	OBIS	18
IOW Macrozoobenthos monitoring Baltic Sea (1980-2005)	-	OBIS	18
Subtidal hyperbenthos monitoring in function of a foreshore suppletion at the Belgian coast, period 2013-2016	10.14284/344	OBIS	18
Benthos Gironde Estuary	-	OBIS	17
Macrobenthos in the Dutch Sector of the North Sea 1991-2001	-	OBIS	17
Benthic fauna of the Southwest Alentejo and Vicentine Coast Natural Park (SW Portugal) collected in August 2011	10.14284/464	OBIS	15
CEMIEO_GS_RL_UNAM	-	OBIS	15
Littoral Monitoring Network of Cantabria (Invertebrates)	-	OBIS	15
Macrobenthos data from the Norwegian Skagerrak coast	-	OBIS	14
Macrobenthos from the Norwegian waters	-	OBIS	14
North Sea Benthos Survey	-	OBIS	13
Macrobenthos monitoring at long-term monitoring locations, period 2001-ongoing	10.14284/202	OBIS	12
Zooplankton data from central and northern Strait of Georgia	-	OBIS	12
Abundance of intertidal algae and invertebrates on the Atlantic coast of Nova Scotia	-	OBIS	11
LBMRev	-	OBIS	11
Macrozoobenthos of marine waters in mainland Portugal collected in March and September 2010	10.14284/463	OBIS	11
Specific diversity data of macrobenthic communities in the "Pierre Noire" study site in the English Channel from 1977 on	10.21411/kfms-pq29	OBIS	11
Macrobenthos of the Western Scheldt (Ossenisse, Valkenisse, Terneuzen and Vlissingen) on 27 and 28 September 1978	10.14284/231	OBIS	9

Table B1. Continued

Dataset name	DOI	Source	# Records
ACER: Marine Resource Inventory of the Seaside Adjunct, Kejimkujik National Park	-	OBIS	8
Asia-Pacific Dataset	-	OBIS	7
Laspibay-Black Sea	-	OBIS	7
Macrozoobenthos data collected from the Constanta East profile in the Romanian marine waters between 1977 and 1999	-	OBIS	7
Specific diversity data of macrobenthic communities in the "Rivière de Morlaix" study site in the English Channel from 1977 to 1996	10.21411/qxef-sr30	OBIS	7
Acadia University: Invertebrates from mudflats in the Minas Basin (Bay of Fundy), collected for the NaGISA project July 2008	-	OBIS	6
Benthic macrofauna of the Ericeira coast (central Portugal) collected in May 2001	10.14284/462	OBIS	6
Macrobenthos monitoring in the Mar Piccolo of Taranto in June 2013 and April 2014	10.6092/2bqm-v560	OBIS	6
Macrozoobenthos_seagrass_Perseus_IBER-BAS	-	OBIS	6
PELD-ELPA Temporal data series of Benthic macrofauna abundance and composition from the Patos Lagoon estuary	-	OBIS	6
Subtidal macrobenthos monitoring in function of a foreshore suppletion at the Belgian coast, period 2013-2016	10.14284/342	OBIS	6
UNBSJ: Long-term monitoring of benthic infaunal invertebrates at sites in Saint John Harbour, New Brunswick	-	OBIS	6
CAISN: Abundance and biomass of benthic invertebrates collected in four ports of the Canadian Arctic during summers of 2011 and 2012	-	OBIS	5
Danube Mouths Zoobenthos data from 1977 to 1999	-	OBIS	5
Eastern Channel dataset	-	OBIS	5
Epifauna community at Waarde and Saeftinghe (Westerschelde) in 1991	10.14284/224	OBIS	5
Macrozoobenthos_sand_Perseus_IBER-BAS	-	OBIS	5
Marine Biological Sample Database, JAMSTEC	10.48518/00001	OBIS	5
MARITIMES SUMMER RESEARCH VESSEL SURVEY	-	OBIS	5
Strelbay	-	OBIS	5
Abundance & Biomass of benthic infauna from grab samples taken as part of an ecosystem assessment of Belfast Lough in 2012	10.17031/5ydwq8	OBIS	4
Arctic soft-sediment macrobenthos	-	OBIS	4

Table B1. Continued

Dataset name	DOI	Source	# Records
Benthic infaunal abundance and biomass from Belfast Lough dredge disposal monitoring operations 2017 and 2018	10.17031/tyhfos	OBIS	4
Royal Belgian Institute of Natural Sciences marine Chelicerata collection	-	OBIS	4
Subtidal epibenthos and demersal fish monitoring in function of a foreshore suppletion at the Belgian coast, period 2013-2016	10.14284/343	OBIS	4
Crustacea of the environs of Saint John, New Brunswick, Canada as observed in 1967	-	OBIS	3
DFO SABS: Wildish collection of sublittoral macro-infauna collected in the Bay of Fundy and its estuaries	-	OBIS	3
Epibenthos and demersal fish monitoring data in function of wind energy development in the Belgian part of the North Sea	10.14284/53	OBIS	3
Hyperbenthos community in the salt marsh of Saeftinghe in 1990 and 1991	10.14284/225	OBIS	3
Infaunal abundance and biomass data from surveys of the East Antrim Maerl bed in 2004	10.17031/qp1mem	OBIS	3
Kalamitsi	-	OBIS	3
Kongsfjorden/Spitsbergen - soft bottom fauna	10.14284/263	OBIS	3
SARONIKOS	-	OBIS	3
SeaWatch-B: citizens monitoring the Belgian North Sea from the beach (2014-2018)	10.14284/401	OBIS	3
Stable isotope ratios of C and N in benthic macrofauna from Mediterranean seagrass litter accumulations from Calvi Bay in 2011-2012	10.14284/454	OBIS	3
The south coast survey of Hatakejima Islands from 1969	10.48518/00012	OBIS	3
VIMS NorthEast Area Monitoring and Assessment Program	-	OBIS	3
Abundance and Biomass of infaunal species from grab samples from a benthic assessment for Fair Head tidal energy development site (Fair Head, Co. Antrim), 2014	10.17031/vlnar9	OBIS	2
Abundances of benthic infauna from grab sediment samples as part of the INIS Hyrdo project, Co. Down (Northern Ireland), 2011	10.17031/ktr5zl	OBIS	2
Analysis of macrobenthos in the Southern Bight of the North Sea (1971-1972)	10.14284/208	OBIS	2
Finnish Baltic Sea zooplankton monitoring	-	OBIS	2
Macrobenthos monitoring in the Trieste harbour, North Adriatic Sea (Port Authority) in June 2013 and March 2015	10.6092/04wy-4b44	OBIS	2

Table B1. Continued

Dataset name	DOI	Source	# Records
MARITIMES SPRING RESEARCH VESSEL SURVEY	-	OBIS	2
MegFeod-Black Sea	-	OBIS	2
Scientific Results of the New Zealand Government Trawling Expedition, 1907	-	OBIS	2
Spatial distribution of the macrozoobenthos on the 'Slikken van Vianen' (Oosterschelde) in 1979	10.14284/227	OBIS	2
Study of epibenthos and demersal fish in and around the dredging areas of the Belgian Continental Shelf (1977-1981)	10.14284/192	OBIS	2
Study on plankton at the port of Ostend in 1965	10.14284/72	OBIS	2
Tidal and diurnal rhythms of the hyperbenthos at the 'Vlakte van de Raan' on 14 October 1996	10.14284/218	OBIS	2
Universidad CES	-	OBIS	2
Abundance and biomass of benthic infauna as part of the North Channel habitat mapping project, 2017	10.17031/ijdo70	OBIS	1
Analysis of the macrobenthic community near Nieuwpoort (1970-1971)	10.14284/206	OBIS	1
DFO Quebec Region Coastal biodiversity of the benthic epifauna of the St. Lawrence Estuary (2018-2019)	10.26071/ogsl-c2a02113-e69c	OBIS	1
Ecological study of the plankton in the port of Ostend, 1965	10.14284/194	OBIS	1
Epibenthos and demersal fish monitoring at long-term monitoring stations in the Belgian part of the North Sea	10.14284/54	OBIS	1
Feeding rhythms of the common goby Pomatoschistus microps at the brackish tidal marsh 'Het verdronken land van Saeftinge' in 1994	10.14284/228	OBIS	1
Groundfish Survey Invertebrate Data	-	OBIS	1
HELCOM/OSPAR Estonia ports water sampling	-	OBIS	1
HELCOM/OSPAR Netherlands ports water sampling	-	OBIS	1
Jalta-Black Sea	-	OBIS	1
MacroBenthos collected at Issungnak Artificial Island in Southern Beaufort Sea, 1981-1982	-	OBIS	1
Macrobenthos from English waters between 2000-2002	-	OBIS	1
Macrobenthos of the Western Scheldt estuary in September 1978	10.14284/131	OBIS	1
Macrobentos de cuatro playas de alta energía ubicadas en la península de la Guajira, noroeste del Golfo de Venezuela	-	OBIS	1

Table B1. Continued

Dataset name	DOI	Source	# Records
Macrozoobenthos data collected in the Northern part of the Romanian littoral (Danube mouths) between 2000 and 2010	-	OBIS	1
Macrozoobenthos data from the southeastern North Sea in 2000	-	OBIS	1
Macrozoobenthos, National Pilot Monitoring Studies Phyllophora April 2017, EMBLAS-II	-	OBIS	1
Macrozoobenthos, National Pilot Monitoring Studies Phyllophora August 2017, EMBLAS-II	-	OBIS	1
Macrozoobenthos, National Pilot Monitoring Studies Phyllophora July 2017, EMBLAS-II	-	OBIS	1
Marine Recorder Snapshot extract of surveys entered by Wildlife Trusts	10.17031/zwrsjz	OBIS	1
MARITIMES 4VSW RESEARCH VESSEL SURVEY	-	OBIS	1
Mesopelagic Crustaceans of the North Western Portuguese Coast between 1998 and 2000	10.14284/466	OBIS	1
NZ research trawl surveys since 2008	-	OBIS	1
Polish Arctic Marine Programme	10.14284/183	OBIS	1
Programa Poseidon - Citizen Science Project Results	10.14284/470	OBIS	1
Rhodolith Beds in Northern New Zealand: Characterisation of Associated Biodiversity	-	OBIS	1
SHARK_Epibenthos_2012_SVVAEK_Fjallsviksviken_version_2017-04-26.zip	-	OBIS	1
ZooPlankton_159498	-	OBIS	1
ZooPlankton_161107	-	OBIS	1
ZooPlankton_9980	-	OBIS	1
For 253 dataset no name or citation was provided		OBIS	44,838

Appendix C – Chapter 5

Table C1. Bioregions of shallow-water marine Isopoda occurring at depths from 0 to 200 m. For each region the most common species is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	12,119	315	25	79	*Edotia triloba*	*Ancinus depressus, Paranthura floridensis, Machatrium spathulicarpus, Schizobopyrina urocaridis, Excorallana mexicana*
2	31,274	170	42	74	*Saduria entomon*	*Lekanesphaera rugicauda, Natatolana gallica, Idotea neglecta, Pseudarachna hirsuta, Pleurocrypta galateae*
3	3,576	306	8	88	*Bullowanthura pambula*	*Amakusanthura olearia, Ianiropsis alanmillari, Apanthura styphelia, Neastacilla macilenta, Crabyzos longicaudatus*
4	2,330	151	12	76	*Caecognathia crenulatifrons*	*Exosphaeroma amplicauda, Gnathia trilobata, Idotea fewkesi, Exosphaeroma rhomburum, Califanthura squamosissima*
5	1,279	179	12	88	*Acanthoserolis schythei*	*Chaetarcturus aculeatus, Acanthomunna spinipes, Leptoserolis orbiculata, Ianiropsis longipes, Munna gallardoi*
6	1,274	183	5	86	*Leptanthura laevigata*	*Austroarcturus laevis, Joeropsis beuroisi, Ianiropsis palpalis, Iathrippa capensis, Natatolana pilula*
7	709	108	5	80	*Isocladus armatus*	*Cassidina typa, Natatolana aotearoa, Dynamenoides decima, Macrochiridothea uncinata, Natatolana narica*

Table C1. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
8	604	137	3	83	*Onychatrium forceps*	*Gnathia masca, Metacirolana serrata, Gnathia wistari, Gnathia variobranchia, Accalathura avena*
9	204	57	2	84	*Expanathura macronesia*	*Apanthuroides calculosa, Joeropsis dimorpha, Apanthuroides aldabrae, Paracassidinopsis perlata, Mesanthura quadrata*
10	125	43	2	72	*Cassidias africana*	*Elaphognathia ramosa, Oxinasphaera furcata, Baharilana lira, Metacirolana chemola, Cirolana undata*
11	174	32	1	63	*Lyidotea nodata*	*Haliophasma beaufortia, Paracassidina incompta, Agostodina shara, Neastacilla soelae, Austrarcturella pictila*
12	201	19	1	53	*Neonaesa rugosa*	*Mesanthura hieroglyphica, Paranthura bellicauda, Joeropsis hawaiiensis, Creniola breviceps, Colidotea edmondsoni*
13	61	16	1	63	*Acanthoserolis schythei*	*Leptoserolis sheppardae, Munnogonium quequensis, Macrochiridothea robusta, Pentaceration pleonarietis, Cassidias argentinea*
14	50	10	1	60	*Cymodoce brasiliensis*	*Cymodoce brasiliensis, Excorallana oculata, Aporobopyrus calypso, Sphaeromopsis mourei, Cymodoce meridionalis*

Table C2. Bioregions of marine Isopoda occurring at intermediate depths of more than 200 m down to 500 m. For each region the most common species is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	10,717	118	18	88	*Ilyarachna longicornis*	*Leptanthura tenuis, Eurydice truncata, Echinopleura aculeata, Rocinela danmoniensis, Caecognathia abyssorum*
2	259	82	4	93	*Ceratoserolis trilobitoides*	*Serolella pagenstecheri, Desmosoma modestum, Dolichiscus ferrazi, Lionectes humicephalotus, Chaetarcturus longispinosus*
3	139	61	2	100	*Tasmarcturus simplicissimus*	*Tasmarcturus simplicissimus, Joeropsis bicarinata, Acanthomunna lagorchestes, Notopais minya, Pentaceration spinosissima*
4	127	24	2	50	*Politolana polita*	*Ptilanthura tenuis, Hyssura vimsae, Politolana polita, Politolana impressa, Rocinela americana*
5	117	22	1	82	*Notopais spicatus*	*Califanthura pingouin, Munna neglecta, Paranthura costana, Bathygnathia porca, Meridiosignum kerguelensis*
6	76	12	1	67	*Cirolana mclaughlinae*	*Cirolana mclaughlinae, Dolichiscus spinosetosus, Edotia tangaroa, Accalathura gigantissima, Leptanthura glacialis*
7	55	8	1	88	*Acanthamunnopsis milleri*	*Acanthamunnopsis milleri, Zeuxokoma setosa, Paramunna quadratifrons, Munnogonium erratum, Munna stephenseni*
8	50	5	1	80	*Brucerolis hurleyi*	*Brucerolis hurleyi, Aega semicarinata, Rocinela satagia, Brucerolis bromleyana, Natatolana nitida*

Table C3. Bioregions of deep-sea Isopoda occurring deeper than 500 m. For each region the most common species is listed, as well as the five most indicative species for the region. The percentage of endemic species in each bioregion is given. A cell equals a 4° latitudinal-longitudinal grid cell.

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
1	1,831	148	13	67	*Ilyarachna longicornis*	*Echinozone arctica, Caecognathia abyssorum, Pleurogonium rubicundum, Dendrotion spinosum, Astacilla intermedia*
2	1,630	120	5	59	*Macrostylis magnifica*	*Macrostylis magnifica, Leptanthura chardyi, Cornuamesus biscayensis, Munnopsurus atlanticus, Acanthocope carinata*
3	895	77	4	52	*Chelator insignis*	*Astacilla caeca, Bathygnathia curvirostris, Storthyngura truncata, Caecognathia multispinis, Rapaniscus dewdneyi*
4	322	80	3	90	*Brucerolis victoriensis*	*Stylomesus sarsi, Haplodendron buzwilsoni, Tasmarcturus simplicissimus, Austrarcturella brychia, Xenosella coxospinosa*
5	129	71	2	87	*Ceratoserolis meridionalis*	*Holodentata caeca, Frontoserolis abyssalis, Desmosoma australis, Antarcturus hempeli, Macrostylis cerritus*
6	156	51	2	88	*Haploniscus bruuni*	*Eurycope manifesta, Desmosoma dolosus, Gnathia lacunacapitalis, Nannoniscus perunis, Nannoniscus coalescum*
7	235	40	4	83	*Brucerolis brandtae*	*Haploniscus miccus, Acanthomunna proteus, Prochelator tupuhi, Notopais zealandica, Aegiochus nohinohi*
8	121	25	2	60	*Bathynomus giganteus*	*Zeuxokoma elongata, Politolana concharum, Politolana wickstenae, Prochelator incomitatus, Hapsidohedra ochlera*

Table C3. Continued

Bioregion	# Records	# Species	# Cells	% Endemism	Most common species	Top 5 most indicative species
9	58	24	1	92	*Stylomesus natalensis*	*Stylomesus natalensis, Bathynatalia gilchristi, Apanthura insignifica, Notoxenoides acalama, Brianaudea louwae*
10	56	24	1	100	*Chaetarcturus crosnieri*	*Leptanthura segonzaci, Pseudione clevai, Eragia profunda, Stegidotea carinata, Gigantione elconaxii*
11	53	18	1	83	*Natatolana pellucida*	*Bullowanthura crebrui, Quantanthura pacifica, Albanthura stenodactyla, Paranthura longa, Bathygnathia tapinoma*
12	52	18	1	72	*Nannoniscus menziesi*	*Nannoniscus menziesi, Haploniscus intermedius, Janirella ornata, Microthambema tenuis, Micromesus nannoniscoides*
13	59	13	1	92	*Eugerdella kurabyssalis*	*Dendromunna kurilensis, Austroniscus acutus, Fortimesus trispiculum, Parvochelus serricaudis, Microcope stenopigus*
14	53	9	1	67	*Haploniscus silus*	*Haploniscus saphos, Haploniscus silus, Hydroniscus lobocephalus, Bathybadistes andrewsi, Chauliodoniscus tasmanaeus*
15	60	7	1	100	*Metacirolana arnaudi*	*Metacirolana arnaudi, Bathylana apalpalis, Ianiropsis palpalis, Iathrippa capensis, Natatolana anophthalma*
16	53	5	1	20	*Munneurycope murrayi*	*Vanhoeffenura pulchra, Munneurycope murrayi, Limnoria hicksi, Munnopsis abyssalis, Hemiarthrus abdominalis*

Appendix D – List of subterranean isopods

Table D1. Alphabetical list of subterranean isopod species.

Species
Abebaioscia troglodytes
Acanthastenasellus forficuloides
Acteoniscus petrochilosi
Aegonethes antilocapra
Aegonethes cervinus
Afrocerberus letabai
Alboscia jotajota
Alistratia beroni
Alloschizidium cavernicolum
Alpioniscus balthasari
Alpioniscus busljetai
Alpioniscus christiani
Alpioniscus drazinai
Alpioniscus epigani
Alpioniscus fragilis
Alpioniscus giurensis
Alpioniscus haasi
Alpioniscus henroti
Alpioniscus heroldii
Alpioniscus herzegowinensis
Alpioniscus hirci
Alpioniscus iapodicus
Alpioniscus kratochvili
Alpioniscus kuehni
Alpioniscus lossinii
Alpioniscus magnus
Alpioniscus mandalinae
Alpioniscus matsakisi
Alpioniscus onnisi
Alpioniscus sideralis
Alpioniscus stochi
Alpioniscus strasseri
Alpioniscus thracicus
Alpioniscus trogirensis
Alpioniscus vardarensis

Species
Alpioniscus vejdovskyi
Alpioniscus velebiticus
Alpioniscus verhoeffi
Amakusanthura botosaneanui
Amakusanthura lathridia
Amazoniscus eleonorae
Amazoniscus leistikowi
Amazoniscus spica
Amerigoniscus centralis
Amerigoniscus henroti
Amerigoniscus nicholasi
Andhracoides shabuddin
Androniscus brentanus
Androniscus degener
Androniscus dentiger
Androniscus noduliger
Androniscus paolettii
Androniscus stygius
Androniscus subterraneus
Angeliera cosettae
Angeliera dubitans
Angeliera gracilis
Angeliera ischiensis
Angeliera phreaticola
Angeliera psamathus
Angeliera racovitzai
Angeliera rivularis
Angeliera xarifae
Annina fustis
Annina lacustris
Antrolana lira
Armadillidium tabacarui
Arubolana aruboides
Arubolana imula
Arubolana parvioculata

Table D1. Continued

Species
Arubolana rotunditelson
Asellus (Asellus) hilgendorfii
Asellus (Asellus) ismailsezarii
Asellus (Asellus) musashiensis
Asellus (Asellus) primoryensis
Atlantasellus cavernicolus
Atlantasellus dominicanus
Baeticoniscus bullonorum
Bahalana abacoana
Bahalana bowmani
Bahalana caicosana
Bahalana cardiopus
Bahalana exumina
Bahalana geracei
Bahalana yagerae
Balkanoniscus corniculatus
Balkanoniscus minimus
Balkanostenasellus rumelicus
Balkanostenasellus skopljensis
Bamaoniscus lobatus
Banatoniscus karbani
Benthana iporangensis
Benthana xiquinhoi
Beroniscus capreolus
Beroniscus marcelli
Biharoniscus fericeus
Biharoniscus racovitzai
Bilistra cavernicola
Bilistra mollicopulans
Borutzkyella ravesi
Botolana leptura
Brackenphiloscia vandeli
Brackenridgia acostai
Brackenridgia ashleyi
Brackenridgia bridgesi
Brackenridgia cavernarum
Brackenridgia palmitensis

Species
Brackenridgia reddelli
Brackenridgia sphinxensis
Brackenridgia villalobosi
Bragasellus afonsoae
Bragasellus aireyi
Bragasellus bragai
Bragasellus comasi
Bragasellus comasioides
Bragasellus conimbricensis
Bragasellus escolai
Bragasellus frontellum
Bragasellus incurvatus
Bragasellus lagari
Bragasellus lagarioides
Bragasellus meijersae
Bragasellus molinai
Bragasellus notenboomi
Bragasellus pauloae
Bragasellus rouchi
Bragasellus seabrai
Bragasellus stocki
Brasileirinho cavaticus
Buddelundia eberhardi
Bulgarocerberus phreaticus
Bulgaronethes haplophthalmoides
Bunderanthura bundera
Bureschia bulgarica
Bureschia serbica
Burmoniscus coecus
Caecianiropsis birsteini
Caecianiropsis ectiformis
Caecianiropsis goseongensis
Caecianiropsis psammophila
Caecidotea acuticarpa
Caecidotea adenta
Caecidotea alabamensis
Caecidotea alleghenyensis

Table D1. Continued

Species
Caecidotea ancyla
Caecidotea antricola
Caecidotea barri
Caecidotea beattyi
Caecidotea bicrenata
Caecidotea bilineata
Caecidotea cannula
Caecidotea carolinensis
Caecidotea catachaetus
Caecidotea chiapas
Caecidotea circulus
Caecidotea cumberlandensis
Caecidotea cyrtorhynchus
Caecidotea dauphina
Caecidotea dimorpha
Caecidotea extensolinguala
Caecidotea filicispeluncae
Caecidotea fonticulus
Caecidotea franzi
Caecidotea fustis
Caecidotea henroti
Caecidotea hobbsi
Caecidotea holsingeri
Caecidotea incurva
Caecidotea insula
Caecidotea jordani
Caecidotea kendeighi
Caecidotea kenki
Caecidotea lesliei
Caecidotea mackini
Caecidotea macropropoda
Caecidotea metcalfi
Caecidotea mitchelli
Caecidotea nickajackensis
Caecidotea nordeni
Caecidotea nortoni
Caecidotea packardi

Species
Caecidotea pasquinii
Caecidotea paurotrigona
Caecidotea phreatica
Caecidotea pricei
Caecidotea recurvata
Caecidotea reddelli
Caecidotea richardsonae
Caecidotea rotunda
Caecidotea salemensis
Caecidotea scrupulosa
Caecidotea scypha
Caecidotea sequoiae
Caecidotea serrata
Caecidotea simonini
Caecidotea simulator
Caecidotea sinuncus
Caecidotea spatulata
Caecidotea steevesi
Caecidotea stiladactyla
Caecidotea stygia
Caecidotea teresae
Caecidotea tridentata
Caecidotea vandeli
Caecidotea vomeri
Caecidotea zullini
Caecosphaeroma (Caecosphaeroma) virei
Caecosphaeroma (Vireia) burgundum
Caecostenetroides ascensionis
Caecostenetroides ischitanum
Caecostenetroides leptosoma
Caecostenetroides nipponicum
Caecostenetroides ruderalis
Calabozoa pellucida
Calasellus californicus
Calasellus longus
Calycuoniscus spinosus
Castellanethes ighousi

Table D1. Continued

Species
Castellanethes ougougensis
Castellanethes sanfilippoi
Castellanethes soloisensis
Castellanethes velox
Catailana whitteni
Caucasocyphoniscus cavaticus
Caucasonethes borutzkyi
Caucasonethes vandeli
Cetinjella monasterii
Chaimowiczia obybytyra
Chaimowiczia tatus
Chaimowiczia uai
Circoniscus buckupi
Circoniscus carajasensis
Cirolana acanthura
Cirolana adriani
Cirolana conditoria
Cirolana crenata
Cirolana cubensis
Cirolana lingua
Cirolana magna
Cirolana marosina
Cirolana pleoscissa
Cirolana poissoni
Cirolana radicicola
Cirolana yucatana
Cirolana yunca
Cirolanides texensis
Cirolanides wassenichae
Colchidoniscus kutaissianus
Columbasellus acheron
Cordioniscus africanus
Cordioniscus andreevi
Cordioniscus antiparosi
Cordioniscus beroni
Cordioniscus bulgaricus
Cordioniscus graecus

Species
Cordioniscus graevei
Cordioniscus kalimnosi
Cordioniscus kithnosi
Cordioniscus lusitanicus
Cordioniscus paragamiani
Cordioniscus patrizii
Cordioniscus schmalfussi
Cordioniscus stebbingi
Cordioniscus vandeli
Coxicerberus abbotti
Coxicerberus adriaticus
Coxicerberus andamanensis
Coxicerberus anfindicus
Coxicerberus arenicola
Coxicerberus boninensis
Coxicerberus brasiliensis
Coxicerberus delamarei
Coxicerberus enckelli
Coxicerberus fukudai
Coxicerberus insularis
Coxicerberus interstitialis
Coxicerberus kiiensis
Coxicerberus littoralis
Coxicerberus machadoi
Coxicerberus magnus
Coxicerberus mexicanus
Coxicerberus minutus
Coxicerberus mirabilis
Coxicerberus nunezi
Coxicerberus parvulus
Coxicerberus pauliani
Coxicerberus predatoris
Coxicerberus ramosae
Coxicerberus redangensis
Coxicerberus remanei
Coxicerberus renaudi
Coxicerberus rossii

Table D1. Continued

Species
Coxicerberus ruffoi
Coxicerberus simplex
Coxicerberus singhalensis
Coxicerberus syrticus
Coxicerberus tabai
Creaseriella anops
Cruregens fontanus
Cubaris mirandai
Curassanthura bermudensis
Curassanthura canariensis
Curassanthura halma
Curassanthura jamaicensis
Curassanthura yucatanensis
Cyathura tridentata
Cylindroniscus cavicola
Cylindroniscus flaviae
Cylindroniscus maya
Cylindroniscus platoi
Cylindroniscus vallesensis
Cyphonethes biseriatus
Cyphonethes herzegowinensis
Cyphonethes tajanus
Cyphoniscellus gueorguievi
Cyphoniscellus herzegowinensis
Etlastenasellus confinis
Etlastenasellus mixtecus
Eurydice dollfusi
Exalloniscus convexus
Exumalana reptans
Faucheria faucheri
Gabunillo aridicola
Gallasellus heilyi
Graeconiscus caecus
Graeconiscus gevi
Graeconiscus guanophilus
Graeconiscus kournasensis
Graeconiscus liebegotti

Species
Graeconiscus paxi
Graeconiscus strinatii
Graeconiscus strouhali
Haloniscus anophthalmus
Haloniscus longiantennatus
Haloniscus stilifer
Haloniscus tomentosus
Haplophthalmus caecus
Haplophthalmus movilae
Haplophthalmus siculus
Haplophthalmus tismanicus
Haptolana belizana
Haptolana bowmani
Haptolana pholeta
Haptolana somala
Haptolana trichostoma
Haptolana yarraloola
Hawaiioscia microphthalma
Hawaiioscia paeninsulae
Hawaiioscia parvituberculata
Hawaiioscia rapui
Hawaiioscia rotundata
Hoctunus vespertilio
Hondoniscus kitakamiensis
Hondoniscus mogamiensis
Hondoniscus ureirensis
Hyloniscus flammula
Hypsimetopus intrusor
Iansaoniscus georginae
Iansaoniscus iraquara
Iansaoniscus leilae
Iansaoniscus paulae
Isoyvesia striata
Iuiuniscus iuiuensis
Janinella brasiliensis
Janinella renaudae
Johannella purpurea

Table D1. Continued

Species
Kagalana tonde
Kensleylana briani
Kimberleydillo waldockae
Kithironiscus dobrogicus
Kithironiscus paragamiani
Leonardoscia hassalli
Leucocyphoniscus pisanus
Libanonethes novus
Libanonethes probosciferus
Ligidium (Stygoligidium) cavaticum
Lirceolus bisetus
Lirceolus cocytus
Lirceolus hardeni
Lirceolus nidulus
Lirceolus pilus
Lirceolus smithii
Lirceus culveri
Lirceus usdagalun
Lucayalana troglexuma
Macedonethes castellonensis
Macedonethes skopjensis
Macedonethes stankoi
Mackinia birsteini
Mackinia continentalis
Mackinia coreana
Mackinia japonica
Mackinia troglodytes
Magniezia africana
Magniezia gardei
Magniezia guinensis
Magniezia laticarpa
Magniezia studiosorum
Marocolana delamarei
Merozoon vestigatum
Mesoniscus alpicola
Mesoniscus graniger
Metacirolana mayana

Species
Metacirolana ponsi
Metaprosekia igatuensis
Metastenasellus boutini
Metastenasellus camerounensis
Metastenasellus congolensis
Metastenasellus dartevellei
Metastenasellus leleupi
Metastenasellus leysi
Metastenasellus powelli
Metastenasellus tarrissei
Metastenasellus wikkiensis
Metatrichoniscoides salirensis
Mexicerberus troglodytes
Mexiconiscus laevis
Mexilana saluposi
Mexistenasellus atotonoztok
Mexistenasellus coahuila
Mexistenasellus colei
Mexistenasellus floridensis
Mexistenasellus magniezi
Mexistenasellus nulemex
Mexistenasellus parzefalli
Mexistenasellus wilkensi
Microcerberus appolliniacus
Microcerberus caroliniensis
Microcerberus monodi
Microcerberus plesai
Microcerberus remyi
Microcerberus stygius
Microcerberus thracicus
Microcharon acherontis
Microcharon agripensis
Microcharon alamiae
Microcharon anatolicus
Microcharon angelicae
Microcharon angelieri
Microcharon antonellae

Table D1. Continued

Species
Microcharon apolloniacus
Microcharon arganoi
Microcharon ariegensis
Microcharon boui
Microcharon boulanouari
Microcharon boutini
Microcharon bureschi
Microcharon coineanae
Microcharon comasi
Microcharon doueti
Microcharon eurydices
Microcharon galapagoensis
Microcharon halophilus
Microcharon harrisi
Microcharon heimi
Microcharon hellenae
Microcharon hercegovinensis
Microcharon herrerai
Microcharon hispanicus
Microcharon juberthiei
Microcharon karamani
Microcharon kirghisicus
Microcharon latus
Microcharon letiziae
Microcharon longistylus
Microcharon luciae
Microcharon lydicus
Microcharon major
Microcharon margalefi
Microcharon marinus
Microcharon meijersae
Microcharon motasi
Microcharon notenboomi
Microcharon novariensis
Microcharon nuragicus
Microcharon oltenicus
Microcharon orghidani

Species
Microcharon orphei
Microcharon othrys
Microcharon oubrahimae
Microcharon ourikensis
Microcharon phlegethonis
Microcharon phreaticus
Microcharon profundalis
Microcharon quilli
Microcharon raffaellae
Microcharon reginae
Microcharon rouchi
Microcharon sabulum
Microcharon salvati
Microcharon silverii
Microcharon sisyphus
Microcharon stygius
Microcharon tanakai
Microcharon tantalus
Microcharon teissieri
Microcharon thracicus
Microcharon ullae
Microcharon zibani
Microjaera anisopoda
Microjaera morii
Microjanira dentifrons
Microparasellus aloufi
Microparasellus hellenicus
Microparasellus libanicus
Microparasellus puteanus
Miktoniscus longispina
Miktoniscus racovitzai
Mingrelloniscus inchhuricus
Mladenoniscus belavodae
Monolistra (Microlistra) bolei
Monolistra (Microlistra) calopyge
Monolistra (Microlistra) fongi
Monolistra (Microlistra) jalzici

Table D1. Continued

Species
Monolistra (Microlistra) pretneri
Monolistra (Microlistra) schottlaenderi
Monolistra (Microlistra) sketi
Monolistra (Microlistra) spinosa
Monolistra (Microlistra) spinosissima
Monolistra (Monolistra) caeca
Monolistra (Monolistra) monstruosa
Monolistra (Monolistrella) velkovrhi
Monolistra (Pseudomonolistra) bosnica
Monolistra (Pseudomonolistra) hercegovinensis
Monolistra (Pseudomonolistra) radjai
Monolistra (Typhlosphaeroma) bericum
Monolistra (Typhlosphaeroma) boldorii
Monolistra (Typhlosphaeroma) lavalensis
Monolistra (Typhlosphaeroma) matjasici
Monolistra (Typhlosphaeroma) pavani
Monolistra (Typhlosphaeroma) racovitzai
Moserius elbanus
Moserius gruberae
Moserius inexpectatus
Moserius percoi
Moserius talamonensis
Namibianira aigamasensis
Namibianira aikabensis
Namibianira arnhemensis
Namibianira dracohalitus
Neophreatoicus assimilis
Neostenetroides magniezi
Neostenetroides schotteae
Neostenetroides stocki
Niambia botswanaensis
Niambia ghaubensis
Niambia namibiaensis
Nichollsia kashiense
Nichollsia menoni
Nipponasellus hubrichti

Species
Nipponasellus kagaensis
Papuaphiloscia insulana
Papuaphiloscia laevis
Papuaphiloscia parkeri
Paractenoscia cavernicola
Paraplatyarthrus crebesconiscus
Paraplatyarthrus cunyuensis
Paraplatyarthrus occidentoniscus
Paraplatyarthrus pallidus
Paraplatyarthrus subterraneus
Parastenasellus chappuisi
Pectenoniscus carinhanhensis
Pectenoniscus fervens
Pectenoniscus iuiuensis
Pectenoniscus juveniliensis
Pectenoniscus liliae
Pectenoniscus montalvaniensis
Pectenoniscus morrensis
Pectenoniscus pankaru
Pectenoniscus santanensis
Phreatoasellus akyioshiensis
Phreatoasellus higoensis
Phreatoasellus iriei
Phreatoasellus joianus
Phreatoasellus kawamurai
Phreatoasellus miurai
Phreatoasellus uenoi
Phreatoasellus yoshinoensis
Phreatoicoides gracilis
Phreatoicoides longicollis
Phreatoicus orarii
Phreatoicus typicus
Pilbarophreatoicus platyarthricus
Platanosphaera ariadnae
Platanosphaera cavernarum
Platanosphaera kournasensis
Platyarthrus hoffmannseggii

Table D1. Continued

Species
Pongycarcinia xiphidiourus
Porcellio cavernicolus
Porcellionides cavernarum
Porcellionides habanensis
Porcellionides minutissimus
Proasellus acutianus
Proasellus adriaticus
Proasellus alavensis
Proasellus albigensis
Proasellus ambracicus
Proasellus amiterninus
Proasellus anophtalmus
Proasellus aquaecalidae
Proasellus aragonensis
Proasellus arnautovici
Proasellus arthrodilus
Proasellus bagradicus
Proasellus barduanii
Proasellus basnosanui
Proasellus bellesi
Proasellus beroni
Proasellus beticus
Proasellus boui
Proasellus cantabricus
Proasellus cavaticus
Proasellus chappuisi
Proasellus chauvini
Proasellus claudei
Proasellus coiffaiti
Proasellus collignoni
Proasellus comasi
Proasellus cretensis
Proasellus danubialis
Proasellus delhezi
Proasellus deminutus
Proasellus dianae
Proasellus ebrensis

Species
Proasellus elegans
Proasellus escolai
Proasellus espanoli
Proasellus exiguus
Proasellus ezzu
Proasellus faesulanus
Proasellus franciscoloi
Proasellus gardinii
Proasellus gauthieri
Proasellus gineti
Proasellus gjorgjevici
Proasellus gourbaultae
Proasellus grafi
Proasellus granadensis
Proasellus guipuzcoensis
Proasellus henseni
Proasellus hercegovinensis
Proasellus hermallensis
Proasellus hurki
Proasellus hypogeus
Proasellus intermedius
Proasellus jaloniacus
Proasellus karamani
Proasellus lagari
Proasellus lescherae
Proasellus leysi
Proasellus ligusticus
Proasellus linearis
Proasellus ljovuschkini
Proasellus lusitanicus
Proasellus lykaonicus
Proasellus malagensis
Proasellus maleri
Proasellus margalefi
Proasellus mateusorum
Proasellus meijersae
Proasellus micropectinatus

Table D1. Continued

Species
Proasellus minoicus
Proasellus monodi
Proasellus monsferratus
Proasellus montenigrinus
Proasellus navarrensis
Proasellus nolli
Proasellus notenboomi
Proasellus orientalis
Proasellus ortizi
Proasellus oviedensis
Proasellus pamphylicus
Proasellus parvulus
Proasellus patrizii
Proasellus pavani
Proasellus phreaticus
Proasellus pisidicus
Proasellus polychaetus
Proasellus racovitzai
Proasellus rectangulatus
Proasellus rectus
Proasellus rouchi
Proasellus similis
Proasellus sketi
Proasellus slavus
Proasellus slovenicus
Proasellus solanasi
Proasellus soriensis
Proasellus spelaeus
Proasellus spinipes
Proasellus stocki
Proasellus strouhali
Proasellus synaselloides
Proasellus thermonyctophilus
Proasellus valdensis
Proasellus vandeli
Proasellus variegatus
Proasellus vignai

Species
Proasellus vizcayensis
Proasellus vulgaris
Proasellus walteri
Proasellus winteri
Protelsonia bureschi
Protelsonia gjorgjevici
Protelsonia hungarica
Protelsonia lakatnikensis
Protocerberus schminkei
Protocharon arenicola
Protocharon stocki
Protojanira leleupi
Protojanira prenticei
Protojaniroides ficki
Protojaniroides perbrincki
Protonethes ocellatus
Protracheoniscus gakalicus
Pseudobuddelundiella hostensis
Pseudobuddelundiella ljovuschkini
Pygolabis eberhardi
Pygolabis gascoyne
Pygolabis humphreysi
Pygolabis paraburdoo
Pygolabis weeliwolli
Quatuordillo caecus
Remasellus parvus
Rhodopioniscus beroni
Saharolana seurati
Salmasellus howarthi
Salmasellus steganothrix
Schizidium beroni
Schizidium perplexum
Sibirasellus parpurae
Skotobaena monodi
Skotobaena mortoni
Spelaeonethes brixiensis
Spelaeonethes mancinii

Table D1. Continued

Species
Spelaeonethes medius
Spelaeonethes nodulosus
Spelaeoniscus ragonesei
Spelunconiscus castroi
Spelunconiscus septemlacuum
Speocirolana bolivari
Speocirolana disparicornis
Speocirolana endeca
Speocirolana fustiura
Speocirolana guerrei
Speocirolana hardeni
Speocirolana lapenita
Speocirolana pelaezi
Speocirolana prima
Speocirolana pubens
Speocirolana thermydronis
Speocirolana xilitla
Speocirolana zumbadora
Sphaerolana affinis
Sphaerolana interstitialis
Sphaerolana karenae
Sphaeromides bureschi
Sphaeromides polateni
Sphaeromides raymondi
Sphaeromides virei
Spherarmadillo cavernicola
Stenasellus agiuranicus
Stenasellus asiaticus
Stenasellus assorgiai
Stenasellus bedosae
Stenasellus boutini
Stenasellus bragai
Stenasellus breuili
Stenasellus brignolii
Stenasellus buili
Stenasellus cambodianus
Stenasellus chapmani

Species
Stenasellus costai
Stenasellus covillae
Stenasellus deharvengi
Stenasellus escolai
Stenasellus foresti
Stenasellus galhanoae
Stenasellus grafi
Stenasellus guinensis
Stenasellus henryi
Stenasellus javanicus
Stenasellus kenyensis
Stenasellus magniezi
Stenasellus messanai
Stenasellus migiurtinicus
Stenasellus mongnatei
Stenasellus monodi
Stenasellus nuragicus
Stenasellus pardii
Stenasellus racovitzai
Stenasellus rigali
Stenasellus ruffoi
Stenasellus simonsi
Stenasellus stocki
Stenasellus strinatii
Stenasellus taitii
Stenasellus tashanensis
Stenasellus tashanicus
Stenasellus vermeuleni
Stenasellus virei
Stenobermuda iliffei
Stenobermuda mergens
Strouhaloniscellus anophthalmus
Strouhaloniscellus biokovoensis
Strouhaloniscellus gordani
Stygasellus phreaticus
Stygocyathura beroni
Stygocyathura broodbakkeri

Table D1. Continued

Species
Stygocyathura chapmani
Stygocyathura cuborientalis
Stygocyathura curassavica
Stygocyathura fijiensis
Stygocyathura filipinica
Stygocyathura hummelincki
Stygocyathura mexidos
Stygocyathura milloti
Stygocyathura motasi
Stygocyathura munae
Stygocyathura numeae
Stygocyathura orghidani
Stygocyathura papuae
Stygocyathura parapotamica
Stygocyathura rapanuia
Stygocyathura salpiscinalis
Stygocyathura sbordonii
Stygocyathura specus
Stygocyathura taitii
Stygocyathura univam
Stygocyathura wadincola
Synasellus albicastrensis
Synasellus barcelensis
Synasellus bragai
Synasellus bragaianus
Synasellus brigantinus
Synasellus capitatus
Synasellus dissimilis
Synasellus exiguus
Synasellus favaiensis
Synasellus flaviensis
Synasellus fragilis
Synasellus henrii
Synasellus hurki
Synasellus insignis
Synasellus intermedius
Synasellus lafonensis

Species
Synasellus leysi
Synasellus longicauda
Synasellus longicornis
Synasellus mariae
Synasellus mateusi
Synasellus meijersae
Synasellus meirelesi
Synasellus minutus
Synasellus nobrei
Synasellus notenboomi
Synasellus pireslimai
Synasellus pombalensis
Synasellus robusticornis
Synasellus serranus
Synasellus tirsensis
Synasellus transmontanus
Synasellus valpacensis
Synasellus vidaguensis
Synasellus vilacondensis
Tainisopus fontinalis
Tainisopus napierensis
Tauroligidium stygium
Tauronethes lebedinskyi
Thailandoniscus brehieri
Thailandoniscus whitteni
Thaumatoniscellus speluncae
Titanethes albus
Trachelipus cavaticus
Trachelipus troglobius
Trichonethes kosswigi
Trichoniscoides arlanza
Trichoniscoides bellesi
Trichoniscoides broteroi
Trichoniscoides cantabricus
Trichoniscoides galiana
Trichoniscoides govillari
Trichoniscoides jonfernandezi

Table D1. Continued

Species
Trichoniscoides machadoi
Trichoniscoides marinae
Trichoniscoides meridionalis
Trichoniscoides ouremensis
Trichoniscoides serrai
Trichoniscoides sicoensis
Trichoniscoides subterraneus
Trichoniscoides viejoi
Trichoniscus anopthalmus
Trichoniscus aphonicus
Trichoniscus baschierii
Trichoniscus bassoti
Trichoniscus beroni
Trichoniscus beschkovi
Trichoniscus bononiensis
Trichoniscus bulgaricus
Trichoniscus bureschi
Trichoniscus cavernicola
Trichoniscus dancaui
Trichoniscus garevi
Trichoniscus gudauticus
Trichoniscus hoctuni
Trichoniscus jeanneli
Trichoniscus lindbergi
Trichoniscus petrovi
Trichoniscus racovitzai
Trichoniscus rhodopiensis
Trichoniscus stoevi
Trichoniscus tenebrarum
Trichoniscus tranteevi
Trichoniscus tuberculatus
Trichoniscus valkanovi
Trichoniscus vandeli
Trichorhina anophthalma
Trichorhina atoyacensis
Trichorhina bequaerti
Trichorhina boneti

Species
Trichorhina cipoensis
Trichorhina guanophila
Trichorhina pataxosi
Trichorhina pearsei
Trichorhina vandeli
Tricyphoniscus bureschi
Troglarmadillidium stygium
Troglarmadillo cavernae
Trogleluma machadoi
Troglocyphoniscus absoloni
Troglocyphoniscus osellai
Trogloianiropsis lloberai
Troglonethes arrabidaensis
Troglonethes aurouxi
Troglonethes fonsocalvoi
Troglonethes olissipoensis
Trogloniscus cavernicolus
Trogloniscus clarkei
Trogloniscus deharvengi
Trogloniscus hengliensis
Trogloniscus trilobatus
Troglopactes botosaneanui
Troglophiloscia belizensis
Troglophiloscia laevis
Troglophiloscia silvestrii
Turcolana adaliae
Turcolana cariae
Turcolana detecta
Turcolana lepturoides
Turcolana pamphyliae
Turcolana reichi
Turcolana rhodica
Turcolana smyrnae
Turcolana steinitzi
Typhlarmadillidium occidentale
Typhlocirolana buxtoni
Typhlocirolana fontis

Table D1. Continued

Species
Typhlocirolana gurneyi
Typhlocirolana haouzensis
Typhlocirolana ichkeuli
Typhlocirolana longimera
Typhlocirolana margalefi
Typhlocirolana moraguesi
Typhlocirolana rifana
Typhlocirolana tiznitensis
Typhlocirolana troglobia
Typhloligidium coecum
Typhloligidium karabijajlae
Typhloligidium kovali
Typhloligidium lithophagum
Typhlotricholigioides aquaticus
Uenasellus iyoensis
Vandeloniscellus bulgaricus
Venezillo articulatus
Venezillo boneti
Venezillo cacahuampilensis
Venezillo chiapensis
Venezillo llamasi
Venezillo osorioi
Venezillo pleogoniophorus
Venezillo tenerifensis
Wiyufiloides osornoensis
Xangoniscus aganju
Xangoniscus ceci
Xangoniscus dagua
Xangoniscus ibiracatuensis
Xangoniscus itacarambiensis
Xangoniscus lapaensis
Xangoniscus loboi
Xangoniscus lundi
Xangoniscus odara
Xangoniscus santinhoi
Yucatalana robustispina
Zulialana coalescens

Appendix E – List of parasitic isopods

Table E1. Annotated species list of parasitic isopods. Species are listed alphabetically. Information is provided on the type of parasitism (ecto- or endoparasitic), the site of attachment to the host, and the taxonomic placement of the host species. However, for some species the hosts and/or site of attachment is not known. This is especially the case for gnathiids, where only the larval stages are parasitic and species are commonly described and identified based on the adult male. (?) indicates that the consulted literature did not specifically mention the given information or was inconclusive. In these cases the typical site of attachment for the genus or family is provided.

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Achelion occidentalis	endoparasitic	brood cavity	Decapoda – Mithracidae & Inachoididae
Acrobelione anisopoda	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Acrobelione halimedae	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Acrobelione langi	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Acrobelione reverberii	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Aegathoa elongata	ectoparasitic	unknown	Teleostei – Carangidae
Aegathoa oculata	ectoparasitic	unknown	Teleostei – Sparidae (?)
Aegoniscus gigas	ectoparasitic	marsupium	Isopoda – Aegidae
Aegophila cappa	ectoparasitic	thorax	Isopoda – Aegidae
Aegophila socialis	ectoparasitic	legs; marsupium	Isopoda – Aegidae
Afrignathia multicavea	ectoparasitic	unknown	unknown
Agarna bengalensis	ectoparasitic	unknown	unknown
Agarna cumulus	ectoparasitic	branchial cavity	Teleostei – Acanthuridae
Agarna malayi	ectoparasitic	branchial cavity	Teleostei – Dorosomatidae & Mugilidae
Agarna pustulosa	ectoparasitic	branchial cavity	unknown
Akrophryxus acinaces	ectoparasitic	antennules	Decapoda – Goneplacidae
Akrophryxus milvus	ectoparasitic	antennules	Decapoda – Ethusidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Akrophryxus pallipalicus	ectoparasitic	antennules	Decapoda – Palicidae
Albunione australiana	ectoparasitic	branchial cavity (?)	Decapoda – Albuneidae
Albunione indecora	ectoparasitic	branchial cavity (?)	Decapoda – Albuneidae
Albunione yoda	ectoparasitic	branchial cavity	Decapoda – Albuneidae
Allathelges alisonae	ectoparasitic	abdomen	Decapoda – Diogenidae
Allathelges pakistanensis	ectoparasitic	abdomen	Decapoda – Diogenidae
Allobopyrus rumphiusi	ectoparasitic	unknown	Decapoda – Palaemonidae
Allocancrion yunnu	endoparasitic	visceral cavity	Decapoda – Plagusiidae
Allodiplophryxus floridanus	ectoparasitic	abdomen	Decapoda – Palaemonidae
Allodiplophryxus unilateralis	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Allokepon hendersoni	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae
Allokepon longicauda	ectoparasitic	unknown	Decapoda – Portunidae
Allokepon monodi	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae & Inachoididae
Allokepon sinensis	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae
Allokepon tiariniae	ectoparasitic	branchial cavity	Decapoda – Epialtidae
Allophryxus ruber	ectoparasitic	unknown	unknown
Allorbimorphus australiensis	ectoparasitic	unknown	Decapoda – Porcellanidae
Allorbimorphus haigae	ectoparasitic	unknown	Decapoda – Porcellanidae
Allorbimorphus lamellosus	ectoparasitic	unknown	Decapoda – Porcellanidae
Allorbimorphus scabriculi	ectoparasitic	unknown	Decapoda – Porcellanidae
Allorbimorphus tuberculus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Amblycephalon indicus	ectoparasitic	body surface	Teleostei – Sphyraenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Amblycephalon schadleri	ectoparasitic	body surface (?)	unknown
Anacepon sibogae	ectoparasitic	unknown	Decapoda – Parthenopidae
Anathelges foliosus	ectoparasitic	abdomen (?)	unknown
Anathelges hyphalus	ectoparasitic	abdomen	Decapoda – Paguridae
Anathelges hyptius	ectoparasitic	abdomen	Decapoda – Paguridae
Anathelges resupinatus	ectoparasitic	abdomen (?)	unknown
Anchiarthrus derelictus	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Ancyroniscus bonnieri	ectoparasitic	marsupium	Isopoda – Sphaeromatidae
Ancyroniscus orientalis	ectoparasitic	marsupium (?)	Isopoda – Aegidae
Anilocra abudefdufi	ectoparasitic	beneath eye	Teleostei – Pomacentridae
Anilocra acanthuri	ectoparasitic	fins	Teleostei – Acanthuridae
Anilocra acuminata	ectoparasitic	unknown	unknown
Anilocra acuta	ectoparasitic	base of fins	Holostei – Lepisosteidae; Teleostei – Sciaenidae & Esocidae
Anilocra alloceraea	ectoparasitic	unknown	Teleostei (?)
Anilocra amboinensis	ectoparasitic	unknown	Teleostei – Acanthuridae & Balistidae
Anilocra angeladaviesae	ectoparasitic	unknown	unknown
Anilocra ankistra	ectoparasitic	unknown	Teleostei (?)
Anilocra apogonae	ectoparasitic	head	Teleostei – Apogonidae
Anilocra atlantica	ectoparasitic	unknown	Teleostei (?)
Anilocra australis	ectoparasitic	unknown	unknown
Anilocra brillae	ectoparasitic	beneath eye	Teleostei – Serranidae
Anilocra bunkleywilliamsae	ectoparasitic	unknown	Teleostei – Sparidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Anilocra capensis	ectoparasitic	below dorsal fin	Teleostei – Sparidae
Anilocra caudata	ectoparasitic	unknown	Teleostei (?)
Anilocra cavicauda	ectoparasitic	unknown	Teleostei (?)
Anilocra chaetodontis	ectoparasitic	beneath eye	Teleostei – Chaetodontidae
Anilocra chromis	ectoparasitic	beneath eye	Teleostei – Pomacentridae
Anilocra clupei	ectoparasitic	head	Teleostei – Dorosomatidae & Pempheridae
Anilocra coxalis	ectoparasitic	unknown	unknown
Anilocra dimidiata	ectoparasitic	body surface	Teleostei – Dorosomatidae & Leiognathidae
Anilocra elviae	ectoparasitic	claspers	Elasmobranchii (sharks) – Lamnidae
Anilocra frontalis	ectoparasitic	body surface	Teleostei – Sparidae, Mullidae, Sciaenidae, Soleidae & Labridae
Anilocra gigantea	ectoparasitic	unknown	Teleostei – Lutjanidae
Anilocra grandmaae	ectoparasitic	body surface	Teleostei – Dorosomatidae
Anilocra guinensis	ectoparasitic	unknown	unknown
Anilocra hadfieldae	ectoparasitic	unknown	unknown
Anilocra haemuli	ectoparasitic	body surface	Teleostei – Haemulidae
Anilocra hedenborgi	ectoparasitic	unknown	unknown
Anilocra holacanthi	ectoparasitic	beneath eye	Teleostei – Pomacanthidae
Anilocra holocentri	ectoparasitic	interorbital region of head	Teleostei – Holocentridae
Anilocra huacho	ectoparasitic	unknown	unknown
Anilocra ianhudsoni	ectoparasitic	unknown	unknown
Anilocra jovanasi	ectoparasitic	unknown	unknown
Anilocra koolanae	ectoparasitic	body surface	Teleostei – Caesionidae & Carangidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Anilocra laevis	ectoparasitic	unknown	unknown
Anilocra leptosoma	ectoparasitic	body surface; branchial cavity	Teleostei – Dorosomatidae & Sciaenidae
Anilocra longicauda	ectoparasitic	caudal peduncle	Teleostei – Polynemidae
Anilocra marginata	ectoparasitic	unknown	unknown
Anilocra meridionalis	ectoparasitic	beneath eye; operculum	Teleostei – Dorosomatidae
Anilocra monoma	ectoparasitic	unknown	Teleostei – Sparidae, Dorosomatidae, Clupeidae & Sciaenidae
Anilocra montti	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Salmonidae
Anilocra morsicata	ectoparasitic	anterior to dorsal fin	Teleostei – Stomiidae
Anilocra myripristis	ectoparasitic	unknown	Teleostei – Holocentridae
Anilocra nemipteri	ectoparasitic	posterodorsally to eye	Teleostei – Nemipteridae
Anilocra occidentalis	ectoparasitic	unknown	Teleostei (?)
Anilocra partiti	ectoparasitic	beneath eye	Teleostei – Pomacentridae
Anilocra paulsikkeli	ectoparasitic	unknown	unknown
Anilocra physodes	ectoparasitic	body surface	Teleostei – Sparidae, Sphyraenidae & Mugilidae
Anilocra pilchardi	ectoparasitic	body surface	Teleostei – Alosidae, Dorosomatidae, Engraulidae & Sparidae
Anilocra plebeia	ectoparasitic	unknown	unknown
Anilocra pomacentri	ectoparasitic	posterodorsally to eye	Teleostei – Pomacentridae
Anilocra prionuri	ectoparasitic	below nostril	Teleostei – Acanthuridae
Anilocra recta	ectoparasitic	unknown	unknown
Anilocra rhodotaenia	ectoparasitic	unknown	unknown
Anilocra rissoniana	ectoparasitic	unknown	unknown
Anilocra soelae	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Anilocra tropica	ectoparasitic	unknown	unknown
Anisarthrus okunoi	ectoparasitic	abdomen	Decapoda – Rhynchocinetidae
Anisarthrus pelseneeri	ectoparasitic	abdomen	Decapoda – Alpheidae
Anisorbione curva	ectoparasitic	unknown	Decapoda – Penaeidae
Anomophryxus deformatus	ectoparasitic	abdomen	Decapoda – Pandalidae
Anphira branchialis	ectoparasitic	branchial cavity	Teleostei – Serrasalmidae
Anphira guianensis	ectoparasitic	branchial cavity	Teleostei – Serrasalmidae
Anphira junki	ectoparasitic	branchial cavity	Teleostei – Triportheidae
Anphira xinguensis	ectoparasitic	branchial cavity	Teleostei – Serrasalmidae
Antephrya limacis	ectoparasitic	unknown	unknown
Anuropodione amphiandra	ectoparasitic	branchial cavity	Decapoda – Munididae
Anuropodione carolinensis	ectoparasitic	branchial cavity	Decapoda – Munididae
Anuropodione dubius	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Anuropodione megacephalon	ectoparasitic	branchial cavity	Decapoda – Munididae
Anuropodione senegalensis	ectoparasitic	branchial cavity	Decapoda – Munididae
Aparapenaeon brevicoxalis	ectoparasitic	branchial cavity (?)	Decapoda – Solenoceridae & Penaeidae
Aparapenaeon calculosa	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Aparapenaeon japonica	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Aparapenaeon takii	ectoparasitic	branchial cavity (?)	unknown
Apocepon digitatum	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Apocepon leucosiae	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Apocepon pulcher	ectoparasitic	branchial cavity	Decapoda – Leucosiidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Apocumoechus paranebaliae	ectoparasitic	unknown	Leptostraca – Paranebaliidae
Apophrixus constrictus	ectoparasitic	abdomen	Decapoda – Alpheidae
Apophrixus philippinensis	ectoparasitic	abdomen	Decapoda
Aporobopyrina amboinae	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Aporobopyrina anomala	ectoparasitic	branchial cavity	Decapoda – Munididae
Aporobopyrina javaensis	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Aporobopyrina lamellata	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyroides upogebiae	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Aporobopyrus aduliticus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus bonairensis	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus bourdonis	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus calypso	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus collardi	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus curtatus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus dollfusi	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus enosteoidis	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus galleonus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus gracilis	ectoparasitic	branchial cavity	Decapoda
Aporobopyrus megacephalon	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus muguensis	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Aporobopyrus orientalis	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Aporobopyrus oviformis	ectoparasitic	branchial cavity	Decapoda – Porcellanidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Aporobopyrus parvulus	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus parvus	ectoparasitic	branchial cavity	Decapoda
Aporobopyrus pleopodatus	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus retrorsa	ectoparasitic	branchial cavity	Decapoda – Munididae
Aporobopyrus ryukyuensis	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Aporobopyrus trilobatus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Arcturocheres gaussicola	ectoparasitic	marsupium	Isopoda – Austrarcturellidae
Arcturocheres pulchripes	ectoparasitic	marsupium	Isopoda – Arcturidae
Argeia atlantica	ectoparasitic	branchial cavity (?)	Decapoda – Crangonidae
Argeia pugettensis	ectoparasitic	branchial cavity	Decapoda – Crangonidae
Argeiopsis guamensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Argeiopsis inhacae	ectoparasitic	branchial cavity	Decapoda – Stenopodidae
Argeiopsis kensleyi	ectoparasitic	branchial cavity	Decapoda – Spongicolidae
Arthrophryxus beringanus	ectoparasitic	unknown	Lophogastrida – Eucopiidae
Artystone bolivianensis	endoparasitic	visceral cavity	Teleostei – Loricariidae
Artystone minima	endoparasitic	visceral cavity	Teleostei – Lebiasinidae & Characidae
Artystone trysibia	endoparasitic	visceral cavity	Teleostei – Loricariidae, Cichlidae & Callichthyidae
Asconiscus simplex	ectoparasitic	marsupium	Mysida –Mysidae
Asotana formosa	ectoparasitic	buccal cavity	Teleostei – Serrasalmidae
Asotana magnifica	ectoparasitic	buccal cavity	Teleostei – Serrasalmidae
Asotana splendida	ectoparasitic	buccal cavity (?)	Teleostei – Loricariidae
Aspidophryxus discoformis	ectoparasitic	cephalothorax	Mysida – Mysidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Aspidophryxus frontalis	ectoparasitic	cephalon	Mysida – Mysidae
Aspidophryxus izuensis	ectoparasitic	carapace	Mysida – Mysidae
Aspidophryxus japonicus	ectoparasitic	thorax	Mysida – Mysidae
Aspidophryxus peltatus	ectoparasitic	antennules	Mysida – Mysidae
Astalione cruciaria	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Asymmetrione aequalis	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Asymmetrione ambodistorta	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Asymmetrione asymmetrica	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Asymmetrione clibanarii	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Asymmetrione dardani	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Asymmetrione desultor	ectoparasitic	branchial cavity (?)	Decapoda – Paguridae
Asymmetrione foresti	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Asymmetrione globifera	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae & Paguridae
Asymmetrione harmoniae	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Asymmetrione nossibensis	ectoparasitic	branchial cavity	Decapoda
Asymmetrione sallyae	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Asymmetrione shiinoi	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Asymmetrione tuxtlaensis	ectoparasitic	branchial cavity	Decapoda – Diogenidae & Calcinidae
Asymmetrorbione drepanopleon	ectoparasitic	branchial cavity	Decapoda – Sicyoniidae
Asymmetrorbione kempi	ectoparasitic	branchial cavity	Decapoda – Sicyoniidae
Athelges aegyptius	ectoparasitic	abdomen	Decapoda – Diogenidae
Athelges ankistron	ectoparasitic	abdomen	Decapoda – Diogenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Athelges bilobus	ectoparasitic	abdomen (?)	Decapoda – Paguridae
Athelges caudalis	ectoparasitic	abdomen	Decapoda – Calcinidae & Diogenidae
Athelges cladophorus	ectoparasitic	abdomen	Decapoda – Paguridae
Athelges guitarra	ectoparasitic	abdomen (?)	Decapoda – Paguridae
Athelges intermedia	ectoparasitic	abdomen (?)	Decapoda – Paguridae
Athelges lacertosi	ectoparasitic	abdomen (?)	Decapoda – Paguridae
Athelges paguri	ectoparasitic	abdomen	Decapoda – Paguridae
Athelges pelagosae	ectoparasitic	abdomen	Decapoda – Paguridae
Athelges prideauxii	ectoparasitic	abdomen (?)	Decapoda – Paguridae
Athelges takanoshimensis	ectoparasitic	abdomen	Decapoda – Paguridae & Diogenidae
Athelges tenuicaudis	ectoparasitic	abdomen	Decapoda – Paguridae
Atypocepon intermedium	ectoparasitic	branchial cavity (?)	unknown
Avada eldredgei	ectoparasitic	unknown	Decapoda – Porcellanidae
Avada kedavra	ectoparasitic	unknown	Decapoda – Epialtidae
Avada porcellanae	ectoparasitic	unknown	Decapoda – Porcellanidae
Axiophilus mirabiledictu	endoparasitic	unknown	Decapoda – Callianassidae
Azygopleon schmitti	ectoparasitic	abdomen	Decapoda – Alpheidae
Balanopleon tortuganus	ectoparasitic	branchial cavity	Decapoda – Munididae
Bambalocra intwala	ectoparasitic	unknown	Teleostei – Pomacanthidae
Bathione humboldtensis	ectoparasitic	unknown	Decapoda – Munididae
Bathione magnafolia	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Bathygnathia adlerzia	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Bathygnathia affinis	ectoparasitic	unknown	unknown
Bathygnathia bathybia	ectoparasitic	unknown	unknown
Bathygnathia cardiocondyla	ectoparasitic	unknown	Teleostei (?)
Bathygnathia curvirostris	ectoparasitic	unknown	Teleostei (?)
Bathygnathia depaolorosae	ectoparasitic	unknown	unknown
Bathygnathia japonica	ectoparasitic	unknown	unknown
Bathygnathia magnifica	ectoparasitic	unknown	unknown
Bathygnathia monodi	ectoparasitic	unknown	unknown
Bathygnathia oedipus	ectoparasitic	unknown	unknown
Bathygnathia opisthopsis	ectoparasitic	unknown	Teleostei (?)
Bathygnathia porca	ectoparasitic	unknown	Teleostei (?)
Bathygnathia segonzaci	ectoparasitic	unknown	unknown
Bathygnathia tapinoma	ectoparasitic	unknown	Teleostei (?)
Bathygnathia vollenhovia	ectoparasitic	unknown	Teleostei (?)
Bathygyge grandis	ectoparasitic	branchial cavity	Decapoda – Glyphocrangonidae
Bopyrella articulata	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrella calmani	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Bopyrella harmopleon	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Bopyrella malensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrella moluccensis	ectoparasitic	unknown	Decapoda – Alpheidae
Bopyrella tanytelson	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrella thomsoni	ectoparasitic	branchial cavity	Decapoda – Alpheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Bopyrina abbreviata	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Bopyrina choprai	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Bopyrina gigas	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae
Bopyrina ocellata	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Bopyrina sewelli	ectoparasitic	branchial cavity (?)	Decapoda – Chlorotocellidae
Bopyrinella albida	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrinella hadrocoxalis	ectoparasitic	branchial cavity	Decapoda – Thoridae
Bopyrinella nipponica	ectoparasitic	branchial cavity	Decapoda – Thoridae
Bopyrinella parameces	ectoparasitic	branchial cavity (?)	Decapoda – Ogyrididae
Bopyrinella stricticauda	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrinella thorii	ectoparasitic	branchial cavity	Decapoda – Thoridae
Bopyrinina dorsimaculata	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Bopyrinina paucimaculata	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Bopyrione longicapitata	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrione multifeminae	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Bopyrione synalphei	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Bopyrione toloensis	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Bopyrione woodmasoni	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Bopyrissa dawydoffi	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Bopyrissa diogeni	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Bopyrissa distorta	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Bopyrissa fraissei	ectoparasitic	branchial cavity	Decapoda – Diogenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Bopyrissa guamensis	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Bopyrissa kensleyi	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Bopyrissa liberorum	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Bopyrissa magellanica	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Bopyrissa marami	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Bopyrissa novaeguineensis	ectoparasitic	branchial cavity (?)	Decapoda
Bopyrissa oceania	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Bopyrissa pyriforma	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Bopyrissa wolffi	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Bopyrissa xiphidiostega	ectoparasitic	branchial cavity	Decapoda – Calcinidae & Diogenidae
Bopyroides cluthae	ectoparasitic	branchial cavity	Decapoda – Pandalidae
Bopyroides hippolytes	ectoparasitic	branchial cavity	Decapoda – Thoridae
Bopyroides shiinoi	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae & Thoridae
Bopyrophryxus branchiabdominalis	ectoparasitic	branchial cavity	Decapoda – Parapaguridae
Bopyrosa phryxiformis	ectoparasitic	branchial cavity (?)	unknown
Bopyrus bimaculatus	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Bopyrus crangorum	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Bourdonia tridentata	ectoparasitic; hyperparasitic	marsupium	Isopoda – Bopyridae
Braga amapaensis	ectoparasitic	buccal cavity	Teleostei – Acestorhynchidae
Braga bachmanni	ectoparasitic	buccal cavity (?)	Teleostei (?)
Braga brasiliensis	ectoparasitic	unknown	Teleostei (?)
Braga cichlae	ectoparasitic	buccal cavity	Teleostei – Cichlidae

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Braga fluviatilis	ectoparasitic	buccal cavity	Teleostei – Anostomidae
Braga nasuta	ectoparasitic	skin close to pectoral fins and anus	Teleostei – Arapaimidae
Braga patagonica	ectoparasitic	branchial cavity; buccal cavity; skin behind dorsal fin	Teleostei – Serrasalmidae, Bryconidae, Cynodontidae, Sciaenidae & Cichlidae
Branchiophryxus caulleryi	ectoparasitic	branchial cavity	Euphausiacea – Euphausiidae
Branchiophryxus koehleri	ectoparasitic	branchial cavity	Euphausiacea – Euphausiidae
Branchiophryxus nyctiphanae	ectoparasitic	branchial cavity	Euphausiacea – Euphausiidae
Brucethoa bharata	ectoparasitic	branchial cavity	Teleostei – Argentinidae
Bythognathia yucatanensis	ectoparasitic	unknown	Teleostei (?)
Cabirnalia nausicaa	ectoparasitic	abdomen; pereopods	Decapoda – Cryptochiridae
Cabirops bombyliophila	ectoparasitic; hyperparasitic	marsupium	Isopoda – Bopyridae
Cabirops codreanui	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops fraissei	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops ibizae	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops lernaeodiscoides	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops lobiformis	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops marsupialis	ectoparasitic; hyperparasitic	marsupium	Isopoda – Bopyridae
Cabirops montereyensis	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops orbionei	ectoparasitic; hyperparasitic	marsupium	Isopoda – Bopyridae
Cabirops perezi	ectoparasitic; hyperparasitic	marsupium	Isopoda – Bopyridae
Cabirops pseudioni	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops reverberii	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Cabirops tenuis	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Cabirops tuberculatus	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Bopyridae
Caecognathia abyssorum	ectoparasitic	body surface (?)	unknown
Caecognathia agwillisi	ectoparasitic	body surface (?)	unknown
Caecognathia akaroensis	ectoparasitic	body surface (?)	unknown
Caecognathia albescenoides	ectoparasitic	body surface (?)	unknown
Caecognathia andamanensis	ectoparasitic	body surface (?)	unknown
Caecognathia antarctica	ectoparasitic	body surface (?)	unknown
Caecognathia arctica	ectoparasitic	body surface (?)	unknown
Caecognathia bicolor	ectoparasitic	body surface (?)	unknown
Caecognathia branchyponera	ectoparasitic	body surface (?)	unknown
Caecognathia caeca	ectoparasitic	body surface (?)	unknown
Caecognathia calva	ectoparasitic	body surface (?)	unknown
Caecognathia cerina	ectoparasitic	body surface (?)	unknown
Caecognathia consobrina	ectoparasitic	body surface (?)	unknown
Caecognathia coralliophila	ectoparasitic	body surface	Teleostei – Labridae & Serranidae
Caecognathia crenulatifrons	ectoparasitic	body surface (?)	unknown
Caecognathia cryptopais	ectoparasitic	body surface (?)	unknown
Caecognathia diacamma	ectoparasitic	body surface (?)	unknown
Caecognathia dolichoderus	ectoparasitic	body surface (?)	unknown
Caecognathia elongata	ectoparasitic	body surface (?)	unknown
Caecognathia floridensis	ectoparasitic	body surface (?)	unknown
Caecognathia galzini	ectoparasitic	body surface (?)	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Caecognathia gnamptogenys	ectoparasitic	body surface (?)	unknown
Caecognathia hirsuta	ectoparasitic	body surface (?)	unknown
Caecognathia hodgsoni	ectoparasitic	body surface (?)	unknown
Caecognathia huberia	ectoparasitic	body surface (?)	unknown
Caecognathia leptanilla	ectoparasitic	body surface (?)	unknown
Caecognathia multispinis	ectoparasitic	body surface (?)	unknown
Caecognathia nieli	ectoparasitic	body surface (?)	unknown
Caecognathia nipponensis	ectoparasitic	body surface (?)	unknown
Caecognathia pacifica	ectoparasitic	body surface (?)	unknown
Caecognathia paratrechia	ectoparasitic	body surface (?)	unknown
Caecognathia pilosipes	ectoparasitic	body surface (?)	unknown
Caecognathia polaris	ectoparasitic	body surface (?)	unknown
Caecognathia polythrix	ectoparasitic	body surface (?)	unknown
Caecognathia pustulosa	ectoparasitic	body surface (?)	unknown
Caecognathia regalis	ectoparasitic	body surface (?)	unknown
Caecognathia rhektos	ectoparasitic	body surface (?)	unknown
Caecognathia robusta	ectoparasitic	body surface (?)	unknown
Caecognathia sanctaecrucis	ectoparasitic	body surface (?)	unknown
Caecognathia schistifrons	ectoparasitic	body surface (?)	unknown
Caecognathia serrata	ectoparasitic	body surface (?)	unknown
Caecognathia stygia	ectoparasitic	body surface (?)	unknown
Caecognathia trachymesopus	ectoparasitic	body surface (?)	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Caecognathia vanhoeffeni	ectoparasitic	body surface (?)	unknown
Caecognathia vemae	ectoparasitic	body surface (?)	unknown
Caecognathia wagneri	ectoparasitic	body surface (?)	unknown
Cancricepon anagibbosus	ectoparasitic	branchial cavity (?)	Decapoda – Nanocassiopidae
Cancricepon beibusinus	ectoparasitic	branchial cavity (?)	Decapoda – Scalopidiidae
Cancricepon castroi	ectoparasitic	branchial cavity	Decapoda – Trapeziidae
Cancricepon choprae	ectoparasitic	branchial cavity	Decapoda – Panopeidae, Pseudorhombilidae, Domeciidae & Xanthidae
Cancricepon elegans	ectoparasitic	branchial cavity	Decapoda – Pilumnidae
Cancricepon garthi	ectoparasitic	branchial cavity	Decapoda – Dacryopilumnidae
Cancricepon knudseni	ectoparasitic	branchial cavity (?)	Decapoda – Eriphiidae
Cancricepon multituberosum	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Cancricepon pilula	ectoparasitic	branchial cavity (?)	Decapoda – Xanthidae
Cancricepon savignyi	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Cancricepon xanthi	ectoparasitic	branchial cavity (?)	Decapoda – Xanthidae
Cancrion australiensis	endoparasitic	visceral cavity	Decapoda – Portunidae
Cancrion cancrorum	endoparasitic	visceral cavity (?)	Decapoda – Xanthidae
Cancrion carolinus	endoparasitic	visceral cavity	Decapoda
Cancrion deltoides	endoparasitic	visceral cavity	Decapoda – Pilumnidae
Cancrion floridus	endoparasitic	visceral cavity (?)	Decapoda – Xanthidae
Cancrion khanhensis	endoparasitic	visceral cavity	Decapoda – Portunidae
Cancrion miser	endoparasitic	visceral cavity (?)	Decapoda – Pilumnidae
Cancrion needleri	endoparasitic	visceral cavity	Decapoda

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Capitetragonia alphei	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Capitetragonia elliptica	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Capitoniscus australis	ectoparasitic	unknown	unknown
Capitoniscus cumacei	ectoparasitic	marsupium	Cumacea – Lampropidae
Capitoniscus peruvicus	ectoparasitic	unknown	unknown
Carcinione platypleura	ectoparasitic	branchial cavity	Decapoda – Cryptochiridae
Cardiocepon pteroides	ectoparasitic	branchial cavity (?)	Decapoda – Gecarcinidae
Carocryptus laticephalus	ectoparasitic	unknown	unknown
Castrione digiticaudata	ectoparasitic	branchial cavity (?)	Decapoda – Micheleidae
Castrione longicaudata	ectoparasitic	branchial cavity (?)	Decapoda – Micheleidae
Cataphryxus primus	ectoparasitic	branchial cavity (?)	Decapoda – Lysmatidae
Catoessa ambassae	ectoparasitic	buccal cavity	Teleostei – Ambassidae
Catoessa boscii	ectoparasitic	buccal cavity	Teleostei – Carangidae
Catoessa gruneri	ectoparasitic	branchial cavity	Teleostei – Leiognathidae
Catoessa scabricauda	ectoparasitic	buccal cavity (?)	Teleostei (?)
Ceratothoa africanae	ectoparasitic	buccal cavity	Teleostei – Sparidae
Ceratothoa angulata	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Ceratothoa arimae	ectoparasitic	buccal cavity	Teleostei – Kyphosidae
Ceratothoa banksii	ectoparasitic	buccal cavity	Teleostei – Carangidae, Scombridae, Salmonidae, Latridae, Scatophagidae, Kyphosidae, Mugilidae, Pomatomidae & Hemiramphidae
Ceratothoa barracuda	ectoparasitic	buccal cavity	Teleostei – Sphyraenidae
Ceratothoa capri	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Caproidae, Sparidae & Mugilidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Ceratothoa carinata	ectoparasitic	buccal cavity	Teleostei – Carangidae & Lutjanidae
Ceratothoa collaris	ectoparasitic	buccal cavity	Teleostei – Sparidae, Serranidae & Moronidae
Ceratothoa famosa	ectoparasitic	buccal cavity	Teleostei – Sparidae
Ceratothoa gilberti	ectoparasitic	buccal cavity	Teleostei – Mugilidae
Ceratothoa globulus	ectoparasitic	unknown	Teleostei (?)
Ceratothoa gobii	ectoparasitic	unknown	Teleostei – Gobiidae
Ceratothoa guttata	ectoparasitic	buccal cavity	Teleostei – Exocoetidae
Ceratothoa imbricata	ectoparasitic	buccal cavity	Teleostei – Scatophagidae
Ceratothoa italica	ectoparasitic	buccal cavity	Teleostei – Sparidae
Ceratothoa marisrubri	ectoparasitic	buccal cavity	Teleostei – Mullidae & Sparidae
Ceratothoa oestroides	ectoparasitic	buccal cavity	Teleostei – Sparidae & Moronidae
Ceratothoa oxyrrhynchaena	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Sparidae & Acropomatidae; Elasmobranchii (rays & sharks) – Rajidae, Torpedinidae & Scyliorhinidae
Ceratothoa parallela	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Scorpaenidae, Sparidae, Citharidae, Gadidae, Mullidae, Merlucciidae & Triglidae; Elasmobranchii (rays) – Rajidae
Ceratothoa retusa	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Ceratothoa springbok	ectoparasitic	buccal cavity	Teleostei – Sparidae
Ceratothoa steindachneri	ectoparasitic	buccal cavity	Teleostei – Trachinidae, Sparidae, Serranidae & Chlorophthalmidae
Ceratothoa toyamaensis	ectoparasitic	buccal cavity (?)	Teleostei (?)
Ceratothoa trigonocephala	ectoparasitic	unknown	Teleostei (?)
Ceratothoa usacarangis	ectoparasitic	buccal cavity (?)	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Ceratothoa verrucosa	ectoparasitic	buccal cavity	Teleostei – Sparidae
Chimaeroniscus spheramator	ectoparasitic; hyperparasitic	marsupium	Isopoda – Dajidae
Cinusa nippon	ectoparasitic	buccal cavity	Teleostei – Tetraodontidae
Cinusa tetrodontis	ectoparasitic	buccal cavity	Teleostei – Tetraodontidae
Cirolanoniscus willeyi	ectoparasitic	unknown	Isopoda – Cirolanidae
Cironiscus dahli	ectoparasitic	marsupium	Isopoda – Cirolanidae
Claustrathelges macdermotti	ectoparasitic	abdomen	Decapoda – Diogenidae
Clypeoniscus cantacuzenei	ectoparasitic	unknown	Isopoda – Holognathidae
Clypeoniscus hanseni	ectoparasitic	thorax	Isopoda – Idoteidae
Clypeoniscus meinerti	ectoparasitic	unknown	Isopoda – Idoteidae
Clypeoniscus sarsi	ectoparasitic	unknown	unknown
Clypeoniscus stenetrii	ectoparasitic	body surface	Isopoda – Stenetriidae
Colophryxus novangliae	ectoparasitic	unknown	unknown
Coxalione inaequalis	ectoparasitic	branchial cavity	Decapoda – Laomediidae
Creniola breviceps	ectoparasitic	fins	Teleostei – Acanthuridae & Priacanthidae
Creniola laticauda	ectoparasitic	head	Teleostei – Platycephalidae
Creniola saurida	ectoparasitic	unknown	Teleostei (?)
Crinoniscus alepadis	ectoparasitic	unknown	Scalpellomorpha – Heteralepadidae
Crinoniscus cephalatus	ectoparasitic	prosoma	Scalpellomorpha – Scalpellidae
Crinoniscus equitans	ectoparasitic	unknown	Balanomorpha – Balanidae
Crinoniscus politosummus	ectoparasitic	prosoma	Scalpellomorpha – Poecilasmatidae
Crinoniscus stroembergi	ectoparasitic	egg-sac lamella	Scalpellomorpha – Heteralepadidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Cryptobopyrus elongatus	ectoparasitic	unknown	Decapoda
Cryptocancrion brevibrachium	endoparasitic	visceral cavity	Decapoda – Panopeidae
Cryptocisus laevis	ectoparasitic	unknown	unknown
Cryptocotitus acus	ectoparasitic	unknown	unknown
Cryptoniscus paguri	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Peltogastridae
Cryptoniscus planarioides	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Peltogastridae
Cterissa australiensis	ectoparasitic	unknown	Teleostei – Holocentridae
Cterissa sakaii	ectoparasitic	branchial cavity	Teleostei – Holocentridae
Cumoechus insignis	ectoparasitic	marsupium	Cumacea – Diastylidae & Lampropidae
Cymothoa asymmetrica	ectoparasitic	buccal cavity	Teleostei – Sphyraenidae
Cymothoa borbonica	ectoparasitic	buccal cavity	Teleostei – Carangidae
Cymothoa brasiliensis	ectoparasitic	buccal cavity	Teleostei – Carangidae & Sparidae
Cymothoa bychowskyi	ectoparasitic	buccal cavity	Teleostei – Fistulariidae
Cymothoa carangii	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa catarinensis	ectoparasitic	buccal cavity	Teleostei – Sciaenidae
Cymothoa cinerea	ectoparasitic	buccal cavity	Teleostei – Stromateidae
Cymothoa curta	ectoparasitic	buccal cavity	Teleostei – Anablepidae
Cymothoa dufresni	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa elegans	ectoparasitic	buccal cavity	Teleostei – Scatophagidae & Serranidae
Cymothoa epimerica	ectoparasitic	buccal cavity (?)	Teleostei – Serranidae
Cymothoa eremita	ectoparasitic	buccal cavity	Teleostei – Carangidae, Psettodidae, Mugilidae, Stromateidae, Serranidae, Tetraodontidae, Aulopidae, Siganidae, Haemulidae & Sphyraenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Cymothoa excisa	ectoparasitic	buccal cavity	Teleostei – Gerreidae, Haemulidae, Lutjanidae, Priacanthidae, Sciaenidae & Synodontidae
Cymothoa exigua	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Lutjanidae
Cymothoa eximia	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa frontalis	ectoparasitic	buccal cavity	Teleostei – Belonidae
Cymothoa gadorum	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa gerris	ectoparasitic	buccal cavity	Teleostei – Gerreidae
Cymothoa gibbosa	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa globosa	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa guadeloupensis	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa hermani	ectoparasitic	buccal cavity	Teleostei – Scaridae
Cymothoa ianuarii	ectoparasitic	buccal cavity (?)	Teleostei – Priacanthidae & Pleuronectidae
Cymothoa ichtiola	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa indica	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Mugilidae, Gobiidae, Holocentridae, Cichlidae, Sphyraenidae, Synodontidae & Belonidae
Cymothoa liannae	ectoparasitic	buccal cavity	Teleostei – Carangidae
Cymothoa limbata	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa nigropunctata	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa oestrum	ectoparasitic	buccal cavity	Teleostei – Carangidae, Priacanthidae & Sciaenidae
Cymothoa parupenei	ectoparasitic	buccal cavity	Teleostei – Mullidae
Cymothoa plebeia	ectoparasitic	buccal cavity (?)	Teleostei – Haemulidae & Sciaenidae
Cymothoa propria	ectoparasitic	buccal cavity (?)	Teleostei – Carangidae
Cymothoa pulchrum	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Tetraodontidae & Diodontidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Cymothoa recifea	ectoparasitic	buccal cavity	Teleostei – Centropomidae & Carangidae
Cymothoa recta	ectoparasitic	buccal cavity	Teleostei – Balistidae
Cymothoa rhina	ectoparasitic	buccal cavity	Teleostei – Lutjanidae
Cymothoa rotunda	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa scopulorum	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa selari	ectoparasitic	buccal cavity (?)	Teleostei – Carangidae
Cymothoa slusarskii	ectoparasitic	buccal cavity	Teleostei – Sparidae
Cymothoa sodwana	ectoparasitic	buccal cavity	Teleostei – Carangidae
Cymothoa spinipalpa	ectoparasitic	buccal cavity	Teleostei – Carangidae
Cymothoa truncata	ectoparasitic	buccal cavity (?)	Teleostei (?)
Cymothoa vicina	ectoparasitic	buccal cavity	Teleostei – Mugilidae & Plotosidae
Cyproniscus crossophori	ectoparasitic	unknown	Myodocopida – Cypridinidae
Cyproniscus cypridinae	ectoparasitic	unknown	Myodocopida – Cypridinidae
Cyproniscus decemspinosus	ectoparasitic	unknown	unknown
Cyproniscus octospinosus	ectoparasitic	unknown	unknown
Dactylokepon barbuladigitus	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Dactylokepon caribaeus	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Dactylokepon catoptri	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae
Dactylokepon holthuisi	ectoparasitic	branchial cavity (?)	Decapoda – Scyllaridae
Dactylokepon hunterae	ectoparasitic	branchial cavity	Decapoda – Pinnotheridae
Dactylokepon marchadi	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Dactylokepon palaoensis	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Dactylokepon richardsonae	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae
Dactylokepon semipennatus	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae & Pilumnidae
Dactylokepon seychellensis	ectoparasitic	branchial cavity	Decapoda – Tetraliidae
Dactylokepon sulcipes	ectoparasitic	branchial cavity	Decapoda – Leucosiidae
Dajus afromysidis	ectoparasitic	marsupium (?)	Mysida – Mysidae
Dajus mysidis	ectoparasitic	marsupium (?)	Mysida – Mysidae
Dajus profundus	ectoparasitic	marsupium	Mysida – Mysidae
Dajus siriellae	ectoparasitic	marsupium (?)	Mysida – Mysidae
Danalia caulleryi	ectoparasitic	unknown	Decapoda – Galatheidae
Danalia cervix	ectoparasitic	brood cavity	Decapoda – Cryptochiridae
Danalia curvata	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia dohrnii	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia falsicrura	ectoparasitic	abdomen	Decapoda – Cryptochiridae
Danalia galea	ectoparasitic	abdomen; pereopods	Decapoda – Cryptochiridae
Danalia gregaria	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia hapalocarcini	ectoparasitic	carapace	Decapoda – Cryptochiridae
Danalia inopinata	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia larvaeformis	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia longicollis	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia pellucida	ectoparasitic; hyperparasitic	unknown	Rhizocephala – Sacculinidae
Danalia vesica	ectoparasitic	carapace	Decapoda – Cryptochiridae
Danalia ypsilon	ectoparasitic	unknown	Decapoda – Galatheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Dicropleon bifidus	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Dicropleon morator	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Dicropleon periclimenis	ectoparasitic	abdomen	Decapoda – Palaemonidae
Dicropleon processae	ectoparasitic	abdomen (?)	Decapoda – Processidae
Diogenion vermifactus	endoparasitic	visceral cavity	Decapoda – Diogenidae, Calcinidae & Paguridae
Diplophryxus alphei	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Diplophryxus alveolatus	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Diplophryxus gargantua	ectoparasitic	abdomen	Decapoda – Alpheidae
Diplophryxus gracilis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Diplophryxus jordani	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Diplophryxus kempi	ectoparasitic	abdomen	Decapoda – Palaemonidae
Diplophryxus negrimaculatus	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Diplophryxus siankaanensis	ectoparasitic	abdomen	Decapoda – Alpheidae
Discomorphus magnifoliatus	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Discorsobopyrus stebbingi	ectoparasitic	branchial cavity	Decapoda – Pandalidae
Dolichophryxus geminatus	ectoparasitic	unknown	unknown
Elaphognathia aldabrae	ectoparasitic	unknown	Teleostei (?)
Elaphognathia amboinenesis	ectoparasitic	unknown	unknown
Elaphognathia australis	ectoparasitic	unknown	Teleostei (?)
Elaphognathia bacescoi	ectoparasitic	unknown	Teleostei (?)
Elaphognathia bifurcilla	ectoparasitic	unknown	Teleostei (?)
Elaphognathia cornigera	ectoparasitic	unknown	Teleostei – Gobiidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Elaphognathia discolor	ectoparasitic	unknown	unknown
Elaphognathia ferox	ectoparasitic	unknown	Teleostei (?)
Elaphognathia forceps	ectoparasitic	unknown	Teleostei (?)
Elaphognathia froygattella	ectoparasitic	unknown	Teleostei (?)
Elaphognathia gladia	ectoparasitic	unknown	Teleostei (?)
Elaphognathia insolita	ectoparasitic	unknown	Teleostei (?)
Elaphognathia kikuchii	ectoparasitic	unknown	unknown
Elaphognathia korachaensis	ectoparasitic	unknown	unknown
Elaphognathia lucanoides	ectoparasitic	unknown	unknown
Elaphognathia monodi	ectoparasitic	unknown	unknown
Elaphognathia nunomurai	ectoparasitic	unknown	unknown
Elaphognathia queenslandica	ectoparasitic	unknown	Teleostei (?)
Elaphognathia ramosa	ectoparasitic	unknown	Teleostei (?)
Elaphognathia rangifer	ectoparasitic	unknown	unknown
Elaphognathia rimifrons	ectoparasitic	unknown	unknown
Elaphognathia strombosa	ectoparasitic	unknown	unknown
Elaphognathia sugashimaensis	ectoparasitic	unknown	Teleostei (?)
Elaphognathia wolffi	ectoparasitic	unknown	unknown
Elocryptus amplitruncus	ectoparasitic	unknown	unknown
Elthusa acutinasa	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa alvaradoensis	ectoparasitic	branchial cavity	Teleostei – Synodontidae
Elthusa arnoglossi	ectoparasitic	branchial cavity	Teleostei – Bothidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Elthusa atlantniroi	ectoparasitic	branchial cavity	Teleostei – Cepolidae
Elthusa californica	ectoparasitic	branchial cavity	Teleostei – Cottidae
Elthusa caudata	ectoparasitic	branchial cavity	Teleostei – Ophidiidae
Elthusa emarginata	ectoparasitic	branchial cavity	Teleostei – Mullidae
Elthusa epimerias	ectoparasitic	branchial cavity (?)	unknown
Elthusa epinepheli	ectoparasitic	branchial cavity	Teleostei – Serranidae
Elthusa fistularia	ectoparasitic	branchial cavity	Teleostei – Fistulariidae
Elthusa foveolata	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa frontalis	ectoparasitic	branchial cavity (?)	Teleostei – Balistidae
Elthusa menziesi	ectoparasitic	branchial cavity	Teleostei – Cottidae
Elthusa methepia	ectoparasitic	branchial cavity	Teleostei – Achiridae
Elthusa moritakii	ectoparasitic	branchial cavity	Teleostei – Ereuniidae
Elthusa myripristae	ectoparasitic	branchial cavity	Teleostei – Holocentridae
Elthusa nanoides	ectoparasitic	branchial cavity (?)	unknown
Elthusa neocytta	ectoparasitic	buccal cavity	Teleostei – Oreosomatidae
Elthusa nierstraszi	ectoparasitic	branchial cavity (?)	unknown
Elthusa ochotensis	ectoparasitic	branchial cavity (?)	unknown
Elthusa parabothi	ectoparasitic	branchial cavity	Teleostei – Bothidae
Elthusa parva	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa philippinensis	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa poutassouiensis	ectoparasitic	branchial cavity	Teleostei – Gadidae
Elthusa propinqua	ectoparasitic	branchial cavity	Teleostei – Macrouridae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Elthusa pseudorhombus	ectoparasitic	branchial cavity	Teleostei – Paralichthyidae
Elthusa raynaudii	ectoparasitic	buccal cavity	Teleostei – Lutjanidae
Elthusa rotunda	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa sacciger	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Synaphobranchidae
Elthusa samariscii	ectoparasitic	branchial cavity	Teleostei – Samaridae
Elthusa samoensis	ectoparasitic	branchial cavity (?)	unknown
Elthusa sigani	ectoparasitic	branchial cavity	Teleostei – Scorpaenidae & Siganidae
Elthusa sinuata	ectoparasitic	branchial cavity	Teleostei – Cepolidae, Pleuronectidae, Sparidae, Gobidae, Bramidae, Trichiuridae & Argentinidae; Elasmobranchii (rays) – Rajidae; also found on cephalopods
Elthusa splendida	ectoparasitic	buccal cavity	Teleostei – Squalidae
Elthusa tropicalis	ectoparasitic	branchial cavity	Teleostei – Ogcocephalidae
Elthusa turgidula	ectoparasitic	branchial cavity (?)	Teleostei (?)
Elthusa uranoscopus	ectoparasitic	branchial cavity	Teleostei – Uranoscopidae
Elthusa vulgaris	ectoparasitic	branchial cavity	Teleostei – Paralichthyidae, Hexagrammidae & Sebastidae
Elthusa winstoni	ectoparasitic	branchial cavity	Teleostei – Acanthuridae
Elthusa xena	ectoparasitic	branchial cavity (?)	Teleostei – Clinidae
Emetha adriatica	ectoparasitic	buccal cavity (?)	Teleostei (?)
Emetha audouini	ectoparasitic	buccal cavity	Teleostei – Sparidae
Enthylacus trivinctus	endoparasitic; hyperparasitic	mantel cavity (?)	Rhizocephala – Sacculinidae
Entione achaei	endoparasitic	visceral cavity (?)	Decapoda – Alpheidae
Entione cavolinii	endoparasitic	visceral cavity (?)	Decapoda – Grapsidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Entionella eriphiae	endoparasitic	visceral cavity (?)	Decapoda – Eriphiidae
Entionella fluviatilis	endoparasitic	visceral cavity (?)	Decapoda – Varunidae
Entionella monensis	endoparasitic	visceral cavity	Decapoda – Majidae
Entionella okayamaensis	endoparasitic	visceral cavity (?)	Decapoda – Sesarmidae
Entoniscoides okadai	endoparasitic	visceral cavity (?)	Decapoda – Xanthidae
Entoniscus creplinii	endoparasitic	visceral cavity	Decapoda – Porcellanidae
Entoniscus japonicus	endoparasitic	visceral cavity	Decapoda – Porcellanidae
Entoniscus muelleri	endoparasitic	visceral cavity (?)	Decapoda – Porcellanidae
Entoniscus porcellanae	endoparasitic	visceral cavity (?)	Decapoda – Porcellanidae
Entophilus omnitectus	endoparasitic	visceral cavity	Decapoda – Munididae
Eophrixus adriaticus	ectoparasitic	abdomen (?)	Decapoda – Hippolytidae
Eophrixus brevicauda	ectoparasitic	abdomen	Decapoda – Alpheidae
Eophrixus caudatus	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Eophrixus enchophyllus	ectoparasitic	abdomen	Decapoda – Alpheidae
Eophrixus kuboi	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Eophrixus laevimanus	ectoparasitic	abdomen	Decapoda – Alpheidae
Eophrixus leptochelae	ectoparasitic	abdomen (?)	Decapoda – Pasiphaeidae
Eophrixus lysmatae	ectoparasitic	abdomen	Decapoda – Lysmatidae
Eophrixus nigrocinctus	ectoparasitic	abdomen	Decapoda – Palaemonidae
Eophrixus pikei	ectoparasitic	abdomen	Decapoda – Palaemonidae
Eophrixus shojii	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Eophrixus subcaudalis	ectoparasitic	abdomen	Decapoda – Alpheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Eophrixus yusakiensis	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Epicepon belema	ectoparasitic	branchial cavity	Decapoda – Cyclodorippidae
Epicepon indicum	ectoparasitic	branchial cavity	Decapoda – Cyclodorippidae
Epicepon japonicum	ectoparasitic	branchial cavity	Decapoda – Cyclodorippidae
Epipenaeon elegans	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Epipenaeon fissurae	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Epipenaeon grande	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Epipenaeon ingens	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Epipenaeon latifrons	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Epipenaeon oviforme	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Epipenaeon pestai	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Epiphrixus adriaticus	ectoparasitic	abdomen	Decapoda – Alpheidae
Eragia profunda	ectoparasitic	branchial cavity	Decapoda – Crangonidae
Eremitione biacuta	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Eremitione brandaoi	ectoparasitic	branchial cavity (?)	Decapoda – Diogenidae
Eremitione calcinii	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Eremitione clibanaricola	ectoparasitic	branchial cavity	Decapoda – Diogenidae & Calcinidae
Eremitione giardi	ectoparasitic	branchial cavity	Decapoda – Paguridae
Eremitione hyndmanni	ectoparasitic	branchial cavity (?)	Decapoda – Paguridae
Eremitione lata	ectoparasitic	branchial cavity (?)	Decapoda – Paguridae
Eremitione nobilii	ectoparasitic	branchial cavity (?)	Decapoda – Pylochelidae
Eremitione quasimodo	ectoparasitic	branchial cavity	Decapoda – Diogenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Eremitione tuberculata	ectoparasitic	branchial cavity (?)	Decapoda – Lithodidae
Ergyne cervicornis	ectoparasitic	branchial cavity (?)	Decapoda – Polybiidae
Eriphrixus obesus	ectoparasitic	abdomen (?)	Decapoda – Palaemonidae
Eumetor liriopides	endoparasitic	visceral cavity (?)	Rhizocephala – Sacculinidae
Euneognathia gigas	ectoparasitic	unknown	Teleostei (?)
Falsanathelges mariae	ectoparasitic	abdomen	Decapoda – Calcinidae & Diogenidae
Falsanathelges muelleri	ectoparasitic	abdomen (?)	Decapoda – Diogenidae & Calcinidae
Filophryxus dorsalis	ectoparasitic	abdomen	Decapoda – Palaemonidae
Galathocrypta acaudata	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Gareia arafurae	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Gibbagnathia europalothrix	ectoparasitic	unknown	Teleostei (?)
Gigantione bouvieri	ectoparasitic	branchial cavity	Decapoda – Pilumnidae & Dromiidae
Gigantione elconaxii	ectoparasitic	branchial cavity (?)	Decapoda – Axiidae
Gigantione giardi	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione hainanensis	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione hawaiiensis	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione ishigakiensis	ectoparasitic	branchial cavity	Decapoda – Xanthidae & Carpiliidae
Gigantione moebii	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione mortenseni	ectoparasitic	branchial cavity	Decapoda – Dromiidae
Gigantione notonyxae	ectoparasitic	branchial cavity	Decapoda – Goneplacidae
Gigantione petalomerae	ectoparasitic	branchial cavity (?)	Decapoda – Dromiidae
Gigantione pikei	ectoparasitic	branchial cavity	Decapoda – Axiidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gigantione pratti	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione rathbunae	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione rhombos	ectoparasitic	branchial cavity	Decapoda – Euryplacidae
Gigantione sagamiensis	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gigantione tau	ectoparasitic	branchial cavity	Decapoda – Goneplacidae
Gigantione tuberculata	ectoparasitic	branchial cavity	Decapoda – Goneplacidae
Gigantione uberlackerae	ectoparasitic	branchial cavity	Decapoda – Axiidae
Glossobius albinae	ectoparasitic	buccal cavity (?)	Teleostei (?)
Glossobius anctus	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Glossobius auritus	ectoparasitic	buccal cavity	Teleostei – Exocoetidae
Glossobius crassa	ectoparasitic	buccal cavity (?)	Teleostei (?)
Glossobius hemiramphi	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Glossobius impressus	ectoparasitic	buccal cavity	Teleostei – Exocoetidae
Glossobius linearis	ectoparasitic	branchial cavity (?)	Teleostei (?)
Glossobius ogasawarensis	ectoparasitic	branchial cavity (?)	Teleostei (?)
Gnathia acrorudus	ectoparasitic	unknown	unknown
Gnathia africana	ectoparasitic	body surface; fins	Teleostei – Clinidae
Gnathia albescens	ectoparasitic	unknown	unknown
Gnathia albipalpebrata	ectoparasitic	branchial cavity	Elasmobranchii (sharks)
Gnathia alces	ectoparasitic	unknown	unknown
Gnathia andrei	ectoparasitic	unknown	unknown
Gnathia antonbruunae	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia arabica	ectoparasitic	unknown	unknown
Gnathia asperifrons	ectoparasitic	unknown	unknown
Gnathia aureamaculosa	ectoparasitic	body surface; branchial cavity	Teleostei – Acanthuridae, Balastidae, Caesionidae, Gobiidae, Haemulidae, Labridae, Mullidae, Scaridae, Siganidae & Tetraodontidae
Gnathia aureola	ectoparasitic	unknown	unknown
Gnathia beethoveni	ectoparasitic	unknown	unknown
Gnathia bengalensis	ectoparasitic	unknown	unknown
Gnathia bermudensis	ectoparasitic	unknown	unknown
Gnathia biorbis	ectoparasitic	unknown	unknown
Gnathia brachyuropus	ectoparasitic	unknown	unknown
Gnathia brucei	ectoparasitic	unknown	unknown
Gnathia bungoensis	ectoparasitic	unknown	unknown
Gnathia calamitosa	ectoparasitic	unknown	unknown
Gnathia calmani	ectoparasitic	unknown	Teleostei – Balistidae
Gnathia calsi	ectoparasitic	unknown	unknown
Gnathia camponotus	ectoparasitic	unknown	unknown
Gnathia camuripenis	ectoparasitic	unknown	unknown
Gnathia capillata	ectoparasitic	branchial cavity	Elasmobranchii (rays) – Dasyatidae
Gnathia capitellum	ectoparasitic	body surface	Teleostei – Platycephalidae & Tetraodontidae
Gnathia capricornica	ectoparasitic	unknown	unknown
Gnathia carinodenta	ectoparasitic	unknown	unknown
Gnathia clementensis	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia cooki	ectoparasitic	unknown	unknown
Gnathia coralmaris	ectoparasitic	unknown	unknown
Gnathia cornuta	ectoparasitic	unknown	unknown
Gnathia coronadoensis	ectoparasitic	unknown	unknown
Gnathia dejimagi	ectoparasitic	branchial cavity	Elasmobranchii (sharks)
Gnathia dentata	ectoparasitic	unknown	unknown
Gnathia derzhavini	ectoparasitic	unknown	unknown
Gnathia disjuncta	ectoparasitic	unknown	unknown
Gnathia epopstruma	ectoparasitic	unknown	unknown
Gnathia eumeces	ectoparasitic	unknown	unknown
Gnathia excavata	ectoparasitic	unknown	unknown
Gnathia falcipenis	ectoparasitic	unknown	Teleostei – Carangidae, Pomacentridae, Belonidae, Acanthuridae, Albulinidae, Chanidae, Scaridae & Balistidae
Gnathia fallax	ectoparasitic	unknown	unknown
Gnathia firingae	ectoparasitic	unknown	unknown
Gnathia formosa	ectoparasitic	unknown	unknown
Gnathia fragilis	ectoparasitic	unknown	unknown
Gnathia glauca	ectoparasitic	unknown	unknown
Gnathia glaucostega	ectoparasitic	unknown	Elasmobranchii (rays) – Glaucostegidae
Gnathia gonzalezi	ectoparasitic	unknown	unknown
Gnathia grandilaris	ectoparasitic	branchial cavity; buccal cavity	Elasmobranchii (rays & sharks) – Dasyatidae & Carcharhinidae
Gnathia grutterae	ectoparasitic	unknown	Teleostei – Balistidae, Labridae & Tetraodontidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia gurjanovae	ectoparasitic	unknown	unknown
Gnathia halei	ectoparasitic	unknown	unknown
Gnathia hamletgast	ectoparasitic	unknown	unknown
Gnathia hemingwayi	ectoparasitic	unknown	unknown
Gnathia hirsuta	ectoparasitic	unknown	unknown
Gnathia illepidus	ectoparasitic	unknown	unknown
Gnathia incana	ectoparasitic	unknown	unknown
Gnathia indoinsularis	ectoparasitic	unknown	unknown
Gnathia inopinata	ectoparasitic	unknown	unknown
Gnathia iridomyrmex	ectoparasitic	unknown	unknown
Gnathia johanna	ectoparasitic	unknown	unknown
Gnathia koreana	ectoparasitic	unknown	unknown
Gnathia kumejimensis	ectoparasitic	unknown	unknown
Gnathia lacunacapitalis	ectoparasitic	unknown	unknown
Gnathia latidens	ectoparasitic	unknown	unknown
Gnathia lignophila	ectoparasitic	unknown	unknown
Gnathia limicola	ectoparasitic	unknown	unknown
Gnathia luxata	ectoparasitic	unknown	unknown
Gnathia maculosa	ectoparasitic	branchial cavity	Elasmobranchii (rays) – Dasyatidae
Gnathia magdalenensis	ectoparasitic	unknown	unknown
Gnathia malaysiensis	ectoparasitic	unknown	unknown
Gnathia margaritarum	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia marionis	ectoparasitic	unknown	unknown
Gnathia marleyi	ectoparasitic	unknown	Teleostei – Haemulidae, Lutjanidae, Serranidae, Holocentridae, Pomacentridae, Acanthuridae, Chaetodontidae & Scaridae
Gnathia masca	ectoparasitic	unknown	Teleostei – Tetraodontidae, Serranidae & Lethrinidae
Gnathia maxillaris	ectoparasitic	body surface; base of fins; around eyes; near operculum	Teleostei – Sparidae, Blenniidae, Lotidae, Cottidae & Labridae
Gnathia meticola	ectoparasitic	unknown	unknown
Gnathia micheli	ectoparasitic	unknown	unknown
Gnathia mortenseni	ectoparasitic	unknown	unknown
Gnathia mulieraria	ectoparasitic	unknown	unknown
Gnathia mutsuensis	ectoparasitic	unknown	unknown
Gnathia mystrium	ectoparasitic	unknown	unknown
Gnathia nasuta	ectoparasitic	unknown	unknown
Gnathia nicembola	ectoparasitic	unknown	unknown
Gnathia nkulu	ectoparasitic	unknown	unknown
Gnathia notostigma	ectoparasitic	unknown	unknown
Gnathia nubila	ectoparasitic	branchial cavity	Elasmobranchii (rays) – Aetobatidae
Gnathia obtusispina	ectoparasitic	unknown	unknown
Gnathia odontomachus	ectoparasitic	unknown	unknown
Gnathia oxyuraea	ectoparasitic	fins	Teleostei – Gadidae, Triglidae & Sciaenidae
Gnathia panousei	ectoparasitic	unknown	unknown
Gnathia pantherina	ectoparasitic	branchial cavity; nares; buccal cavity	Elasmobranchii (rays & sharks) – Scyliorhinidae & Torpedinidae; Teleostei – Serranidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia parvirostrata	ectoparasitic	branchial cavity	Elasmobranchii (sharks)
Gnathia perimulica	ectoparasitic	unknown	unknown
Gnathia phallonajopsis	ectoparasitic	branchial cavity; buccal cavity	Teleostei – Gadidae
Gnathia philogona	ectoparasitic	unknown	unknown
Gnathia pilosus	ectoparasitic	unknown	Teleostei – Blenniidae, Pomacentridae, Acanthuridae, Sparidae, Serranidae, Labridae, Gobiidae, Scorpaenidae & Terapontidae
Gnathia pipinde	ectoparasitic	body surface	Teleostei – Tetraodontidae
Gnathia piscivora	ectoparasitic	unknown	unknown
Gnathia productatridens	ectoparasitic	unknown	unknown
Gnathia prolasius	ectoparasitic	unknown	unknown
Gnathia puertoricensis	ectoparasitic	unknown	unknown
Gnathia rathi	ectoparasitic	unknown	unknown
Gnathia rectifrons	ectoparasitic	unknown	unknown
Gnathia rhytidoponera	ectoparasitic	unknown	unknown
Gnathia ricardoi	ectoparasitic	unknown	unknown
Gnathia rufescens	ectoparasitic	branchial cavity	Elasmobranchii (rays) – Dasyatidae
Gnathia samariensis	ectoparasitic	unknown	unknown
Gnathia sanrikuensis	ectoparasitic	unknown	unknown
Gnathia scabra	ectoparasitic	unknown	unknown
Gnathia schmidti	ectoparasitic	unknown	unknown
Gnathia serrula	ectoparasitic	unknown	unknown
Gnathia serrulatifrons	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Gnathia sifae	ectoparasitic	unknown	unknown
Gnathia somalia	ectoparasitic	unknown	unknown
Gnathia spongicola	ectoparasitic	unknown	unknown
Gnathia steveni	ectoparasitic	unknown	unknown
Gnathia stigmacros	ectoparasitic	unknown	unknown
Gnathia stoddarti	ectoparasitic	unknown	unknown
Gnathia taprobanensis	ectoparasitic	unknown	unknown
Gnathia teissieri	ectoparasitic	unknown	unknown
Gnathia teruyukiae	ectoparasitic	branchial cavity; buccal cavity	Elasmobranchii (rays & sharks) – Dasyatidae & Ginglymostomatidae
Gnathia tridens	ectoparasitic	unknown	Teleostei – Clinidae
Gnathia trilobata	ectoparasitic	unknown	unknown
Gnathia trimaculata	ectoparasitic	branchial cavity; buccal cavity; fins; body surface	Elasmobranchii (rays & sharks) – Carcharhinidae, Squatinidae, Orectolobidae, Ginglymostomatidae, Stegostomatidae, Sphyrnidae, Myliobatidae, Dasyatidae, Urolophidae, Gymnuridae, Rhinobatidae, Rajidae & Rhincodontidae; Teleostei – Tripterygiidae
Gnathia triospathiona	ectoparasitic	unknown	unknown
Gnathia tuberculata	ectoparasitic	unknown	unknown
Gnathia tuberculosa	ectoparasitic	unknown	unknown
Gnathia ubatuba	ectoparasitic	unknown	unknown
Gnathia varanus	ectoparasitic	unknown	unknown
Gnathia variobranchia	ectoparasitic	unknown	unknown
Gnathia vellosa	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Gnathia venusta	ectoparasitic	unknown	unknown
Gnathia virginalis	ectoparasitic	unknown	unknown
Gnathia vorax	ectoparasitic	body surface; fins; branchial cavity; buccal cavity	Teleostei – Serranidae, Apogonidae, Bothidae, Carangidae, Pomacentridae, Labridae, Sparidae & Mugilidae
Gnathia wistari	ectoparasitic	unknown	unknown
Gnathia zanzibarensis	ectoparasitic	unknown	unknown
Gnomoniscus podasconis	ectoparasitic; hyperparasitic	marsupium (?)	Isopoda – Podasconidae
Goleathopseudione bilobatus	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Gorgoniscus incisodactylus	endoparasitic	brood cavity	Laurida – Synagogidae
Grapsicepon belizeianum	ectoparasitic	branchial cavity (?)	Decapoda – Mithracidae
Grapsicepon edwardsi	ectoparasitic	branchial cavity	Decapoda – Grapsidae
Grapsicepon magnum	ectoparasitic	branchial cavity	Decapoda – Majidae
Grapsicepon messoris	ectoparasitic	branchial cavity (?)	Decapoda – Grapsidae
Grapsicepon micronesianum	ectoparasitic	branchial cavity (?)	Decapoda – Tetraliidae & Trapeziidae
Grapsicepon rotundum	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Gyge angularis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Gyge branchialis	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Gyge fujianensis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Gyge irregularis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Gyge ovalis	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Hemiarthrus abdominalis	ectoparasitic	abdomen	Decapoda – Pandalidae & Thoridae
Hemiarthrus alphei	ectoparasitic	abdomen	Decapoda – Alpheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Hemiarthrus nematocarcini	ectoparasitic	abdomen	Decapoda – Campylonotidae
Hemiarthrus surculus	ectoparasitic	abdomen	Decapoda – Alpheidae
Hemiarthrus synalphei	ectoparasitic	abdomen	Decapoda – Alpheidae
Hemicepon muelleri	ectoparasitic	branchial cavity (?)	Decapoda
Hemioniscus anatifae	ectoparasitic; hyperparasitic	unknown	Scalpellomorpha – Lepadidae
Hemioniscus balani	endoparasitic	ovaries	Balanomorpha – Balanidae & Chthamalidae
Hemioniscus pagurophilus	ectoparasitic; hyperparasitic	mantle cavity	Lithoglyptida – Trypetesidae
Hemiphryxus malindiae	ectoparasitic	abdomen	Decapoda – Palaemonidae
Heterocepon marginatum	ectoparasitic	branchial cavity	Decapoda – Pinnotheridae
Heterophryxus appendiculatus	ectoparasitic	cephalothorax	Euphausiacea – Euphausiidae
Heterophryxus australis	ectoparasitic	unknown	unknown
Heterophryxus elongatus	ectoparasitic	unknown	unknown
Heterophryxus pacificus	ectoparasitic	unknown	unknown
Holophryxus acanthephyrae	ectoparasitic	carapace	Decapoda – Acanthephyridae
Holophryxus alaskensis	ectoparasitic	carapace	Decapoda – Pasiphaeidae
Holophryxus citriformis	ectoparasitic	carapace	Decapoda – Acanthephyridae
Holophryxus fusiformis	ectoparasitic	carapace (?)	Decapoda – Sergestidae
Holophryxus giardi	ectoparasitic	carapace	Decapoda – Benthesicymidae
Holophryxus polyandrus	ectoparasitic	unknown	unknown
Holophryxus quadratahumerale	ectoparasitic	unknown	unknown
Holophryxus richardi	ectoparasitic	unknown	Decapoda – Sergestidae
Holophryxus septapodus	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Holophryxus truncatus	ectoparasitic	unknown	unknown
Hypercepon guamensis	ectoparasitic	branchial cavity	Decapoda – Trapeziidae
Hyperphrixus castrensis	ectoparasitic	abdomen	Decapoda – Palaemonidae
Hyperphrixus filiformis	ectoparasitic	abdomen (?)	Decapoda – Alpheidae
Hyperphrixus tattersalli	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Hypocepon enoeensis	ectoparasitic	unknown	Decapoda – Pinnotheridae
Hypocepon globosus	ectoparasitic	unknown	Decapoda – Pinnotheridae
Hypohyperphrixus latilamellaris	ectoparasitic	abdomen	Decapoda – Alpheidae
Ichthyoxenos africana	ectoparasitic	unknown	Teleostei – Cichlidae
Ichthyoxenos amurensis	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos asymmetrica	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos circularius	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos dentimaxillus	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos expansus	ectoparasitic	unknown	Teleostei – Distichodontidae
Ichthyoxenos formosanus	ectoparasitic	unknown	Teleostei – Cyprinidae
Ichthyoxenos fushanensis	ectoparasitic	unknown	Teleostei – Cyprinidae
Ichthyoxenos geei	mesoparasitic	flesh-burrowing	Teleostei – Cyprinidae
Ichthyoxenos hsiakowensis	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos japonensis	mesoparasitic	flesh-burrowing; near pectoral fin	Teleostei – Cyprinidae
Ichthyoxenos jellinghausii	mesoparasitic	flesh-burrowing; behind lateral fin	Teleostei – Cyprinidae
Ichthyoxenos longenditus	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos micronyx	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Ichthyoxenos minabensis	ectoparasitic	buccal cavity	Teleostei – Chaunacidae
Ichthyoxenos montanus	mesoparasitic	flesh-burrowing (?)	Teleostei – Cyprinidae
Ichthyoxenos opisthopterygium	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos puhi	ectoparasitic	branchial cavity	Teleostei – Muraenidae
Ichthyoxenos quadratus	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos sinensis	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos tanganyikae	ectoparasitic	buccal cavity	Teleostei – Cichlidae
Ichthyoxenos tchangi	ectoparasitic	unknown	Teleostei (?)
Ichthyoxenos yunnanensis	ectoparasitic	unknown	Teleostei (?)
Idusa carinata	ectoparasitic	buccal cavity	Teleostei – Mugilidae
Idusa dieuzeidei	ectoparasitic	branchial cavity	Teleostei – Cynoglossidae
Idusa plagusiae	ectoparasitic	branchial cavity (?)	Teleostei (?)
Ione cornuta	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Ione ovata	ectoparasitic	branchial cavity	Decapoda – Callianassidae & Callichiridae
Ione taiwanensis	ectoparasitic	branchial cavity (?)	Decapoda – Callianideidae
Ione thompsoni	ectoparasitic	branchial cavity	Decapoda – Callianassidae
Ione thoracica	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Ione tubulata	ectoparasitic	branchial cavity (?)	Decapoda (?)
Ionella agassizii	ectoparasitic	branchial cavity	Decapoda – Callianassidae
Ionella compressa	ectoparasitic	branchial cavity (?)	Decapoda – Callichiridae & Callianassidae
Ionella maculata	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Ionella murchisoni	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Isonebula acanthopleon	ectoparasitic	buccal cavity	Teleostei – Curimatidae
Isonebula maculatus	ectoparasitic	body surface; buccal cavity	Teleostei – Serrasalmidae
Izuohshimaphryxus hoshinoi	ectoparasitic	abdomen	Decapoda – Palaemonidae
Joryma brachysoma	ectoparasitic	branchial cavity	Teleostei – Pristigasteridae, Clupeidae & Scombridae
Joryma engraulidis	ectoparasitic	branchial cavity	Teleostei – Engraulidae
Joryma hilsae	ectoparasitic	branchial cavity	Teleostei – Pristigasteridae, Dorosomatidae & Engraulidae
Joryma malabaricus	ectoparasitic	branchial cavity	Teleostei – Pristigasteridae & Dorosomatidae
Joryma sawayah	ectoparasitic	branchial cavity	Teleostei – Pristigasteridae & Terapontidae
Joryma tartoor	ectoparasitic	branchial cavity	Teleostei – Carangidae & Pristigasteridae
Kepon grapsi	ectoparasitic	branchial cavity	Decapoda – Grapsidae
Kepon halimi	ectoparasitic	unknown	Decapoda – Majidae
Kepon orientalis	ectoparasitic	branchial cavity (?)	Decapoda – Varunidae
Kepon typus	ectoparasitic	branchial cavity (?)	Decapoda – Grapsidae
Kolourione premordica	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Kuna insularis	ectoparasitic	branchial cavity	Teleostei – Pomacentridae
Lathraena insidiosa	ectoparasitic	buccal cavity	Teleostei – Engraulidae
Leidya bimini	ectoparasitic	branchial cavity	Decapoda – Sesarmidae
Leidya distorta	ectoparasitic	branchial cavity	Decapoda – Ocypodidae
Leidya infelix	ectoparasitic	branchial cavity (?)	Decapoda – Grapsidae
Leidya ucae	ectoparasitic	branchial cavity (?)	Decapoda – Ocypodidae
Liriopsis monophthalmus	ectoparasitic; hyperparasitic	mantle cavity	Rhizocephala – Peltogastridae
Liriopsis pygmaea	ectoparasitic; hyperparasitic	mantle cavity	Rhizocephala – Peltogastridae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Litobopyrus longicaudatus	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Livoneca bowmani	ectoparasitic	branchial cavity	Teleostei – Sciaenidae, Atherinidae & Clupeidae
Livoneca ovalis	ectoparasitic	branchial cavity; buccal cavity	Elasmobranchii (rays) – Pristidae
Livoneca redmanii	ectoparasitic	branchial cavity	Teleostei – Moronidae, Carangidae, Mugilidae, Soleidae, Cichlidae & Sciaenidae
Lobothorax laevis	ectoparasitic	buccal cavity	Teleostei (?)
Lobothorax nicosmiti	ectoparasitic	buccal cavity	Teleostei – Gempylidae
Lobothorax typus	ectoparasitic	buccal cavity	Teleostei – Trichiuridae
Loki athanus	ectoparasitic	abdomen	Decapoda – Alpheidae
Loki circumsaltanus	ectoparasitic	abdomen	Decapoda – Thoridae
Mediophrixus pinuum	ectoparasitic	abdomen	Decapoda – Alpheidae
Megacepon choprai	ectoparasitic	branchial cavity	Decapoda – Sesarmidae
Megacepon disparatum	ectoparasitic	branchial cavity	Decapoda – Varunidae
Megacepon goetici	ectoparasitic	branchial cavity (?)	Decapoda – Varunidae
Megacepon pleopodatopus	ectoparasitic	branchial cavity (?)	Decapoda – Sesarmidae & Varunidae
Megacepon sesarmae	ectoparasitic	branchial cavity (?)	Decapoda – Sesarmidae
Megacepon sheni	ectoparasitic	branchial cavity	Decapoda – Varunidae
Mesocepon tosizimensis	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Mesophryxus ventralis	ectoparasitic	abdomen	Decapoda – Palaemonidae
Metacepon leidyi	ectoparasitic	branchial cavity (?)	Decapoda – Matutidae
Metacepon pleopodata	ectoparasitic	branchial cavity (?)	Decapoda – Litocheiridae
Metaphrixus carolii	ectoparasitic	abdomen	Decapoda – Hippolytidae
Metaphrixus intutus	ectoparasitic	abdomen	Decapoda – Palaemonidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Metaphrixus rastriferis	ectoparasitic	abdomen	Decapoda – Palaemonidae
Metaphrixus setouchiensis	ectoparasitic	abdomen (?)	decapoda – Hippolytidae
Metathelges muelleri	ectoparasitic	branchial cavity (?)	Decapoda
Micippion asymmetricus	endoparasitic	visceral cavity (?)	Decapoda – Majidae
Microniscus acartii	ectoparasitic	unknown	Copepoda – Acartiidae
Microniscus calani	ectoparasitic	thorax	Copepoda – Calanidae
Microniscus eucalani	ectoparasitic	unknown	Copepoda – Eucalanidae
Microniscus fuscus	ectoparasitic	unknown	Copepoda
Microniscus latyfrons	ectoparasitic	unknown	Copepoda – Acartiidae & Paracalanidae
Microniscus ornatus	ectoparasitic	unknown	Copepoda (?)
Micropodiphryxus richardsonae	ectoparasitic	abdomen	Decapoda – Crangonidae
Minicopenaeon apertum	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Minicopenaeon crosnieri	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Minicopenaeon intermedium	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Minicopenaeon liuruiyui	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Minimathelges minutus	ectoparasitic	abdomen (?)	Decapoda
Minimathelges nanus	ectoparasitic	abdomen	Decapoda – Paguridae
Miophrixus latreutidis	ectoparasitic	abdomen	Decapoda – Hippolytidae
Monodgnathia colobostruma	ectoparasitic	unknown	Teleostei (?)
Monodgnathia cristatipes	ectoparasitic	unknown	unknown
Monodgnathia ponera	ectoparasitic	unknown	Teleostei (?)
Monodgnathia poteriophora	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Mothocya affinis	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Mothocya andoni	ectoparasitic	branchial cavity	Teleostei – Monodactylidae
Mothocya argenosa	ectoparasitic	branchial cavity; buccal cavity	Teleostei – Atherinopsidae, Hemiramphidae & Atherinidae
Mothocya arrosor	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya belonae	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya bermudensis	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya bertlucy	ectoparasitic	branchial cavity	Teleostei – Blenniidae
Mothocya bohlkeorum	ectoparasitic	branchial cavity	Teleostei – Apogonidae
Mothocya collettei	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya epimerica	ectoparasitic	branchial cavity; buccal cavity	Teleostei – Atherinidae
Mothocya gilli	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya girellae	ectoparasitic	branchial cavity	Teleostei – Kyphosidae
Mothocya halei	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae & Kyphosidae
Mothocya kaorui	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya karobran	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya komatsui	ectoparasitic	branchial cavity (?)	unknown
Mothocya lineata	ectoparasitic	buccal cavity	Teleostei – Hemiramphidae
Mothocya longicopa	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya melanosticta	ectoparasitic	branchial cavity	Teleostei – Exocoetidae
Mothocya nana	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya omidaptria	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya panamica	ectoparasitic	branchial cavity	Teleostei – Belonidae

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Mothocya parvostis	ectoparasitic	branchial cavity (?)	Teleostei – Hemiramphidae
Mothocya plagulophora	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya powelli	ectoparasitic	branchial cavity	Teleostei (?)
Mothocya renardi	ectoparasitic	branchial cavity	Teleostei – Belonidae
Mothocya rosea	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya sajori	ectoparasitic	branchial cavity	Teleostei – Hemiramphidae
Mothocya taurica	ectoparasitic	branchial cavity	Teleostei – Alosidae, Clupeidae & Engraulidae
Mothocya waminda	ectoparasitic	branchial cavity	Teleostei – Atherinidae
Mothocya xenobranchia	ectoparasitic	branchial cavity	Teleostei – Belonidae
Munidion cubense	ectoparasitic	branchial cavity	Decapoda – Munididae
Munidion irritans	ectoparasitic	branchial cavity	Decapoda – Munididae
Munidion laterale	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Munidion longipedis	ectoparasitic	branchial cavity	Decapoda – Munididae
Munidion parvum	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Munidion pleuroncodis	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Munidion princeps	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Munnoniscus marsupialis	ectoparasitic	marsupium (?)	Isopoda – Munnopsidae
Munnoniscus sarsi	ectoparasitic	marsupium (?)	Isopoda – Munnopsidae
Nalocryptus longicaudatus	ectoparasitic	unknown	unknown
Neophryxus globicaudatus	ectoparasitic	pleopods	Decapoda – Palaemonidae
Neritoniscus euphoticus	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Nerocila acuminata	ectoparasitic	body surface; fins	Teleostei – Ariidae, Anablepidae, Megalopidae, Auchenipteridae, Engraulidae, Atherinidae, Serranidae, Mugilidae, Embiotocidae & Sciaenidae; Elasmobranchii (rays) – Rhinobatidae
Nerocila armata	ectoparasitic	fins	Teleostei – Priacanthidae
Nerocila arres	ectoparasitic	caudal peduncle; caudal fin	Teleostei – Terapontidae & Nemipteridae
Nerocila barramundae	ectoparasitic	body surface; fins	Teleostei – Ariidae
Nerocila benrosei	ectoparasitic	head; below or above eye	Teleostei – Labridae
Nerocila bivittata	ectoparasitic	caudal peduncle	Teleostei – Blenniidae, Labridae, Scorpaenidae, Cottidae, Sparidae, Mugilidae, Centracanthidae, Merluccidae, Monacanthidae, Sciaenidae, Mullidae, Gobiidae, Serranidae, Triglidae & Platycephalidae
Nerocila blainvillei	ectoparasitic	unknown	Teleostei (?)
Nerocila californica	ectoparasitic	fins; body surface	Teleostei – Mugilidae, Paralichthyidae, Engraulidae, Carangidae, Embiotocidae, Ariidae, Serranidae, Atherinidae & Scorpaenidae
Nerocila congener	ectoparasitic	unknown	Teleostei (?)
Nerocila depressa	ectoparasitic	fins; body surface	Teleostei – Dorosomatidae, Engraulididae, Cyprinidae & Pristigasteridae
Nerocila donghaiensis	ectoparasitic	body surface	Teleostei – Sciaenidae
Nerocila excisa	ectoparasitic	unknown	Teleostei (?)
Nerocila exocoeti	ectoparasitic	body surface	Teleostei – Exocoetidae
Nerocila falcata	ectoparasitic	unknown	Teleostei (?)
Nerocila falklandica	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Nerocila fluviatilis	ectoparasitic	body surface	Teleostei – Priacanthidae
Nerocila hemirhamphusi	ectoparasitic	unknown	Teleostei – Hemiramphidae
Nerocila heterozota	ectoparasitic	unknown	Teleostei (?)
Nerocila japonica	ectoparasitic	fins	Teleostei – Lateolabracidae, Cyprinidae, Mugilidae, Triglidae, Latidae, Sparidae, Terapontidae, Embiotocidae, Gobiidae, Labridae, Monacanthidae, Molidae & Kyphosidae
Nerocila kisra	ectoparasitic	caudal peduncle; lower jaw	Teleostei – Terapontidae, Scombridae, Lutjanidae, Polynemidae, Pristigasteridae, Sparidae & Sciaenidae
Nerocila lanceolata	ectoparasitic	fins	Teleostei – Lobotidae & Sparidae
Nerocila laticeps	ectoparasitic	unknown	Teleostei (?)
Nerocila livida	ectoparasitic	branchial cavity	Teleostei (?)
Nerocila lomatia	ectoparasitic	branchial cavity	Teleostei – Clupeidae
Nerocila longispina	ectoparasitic	body surface; fins	Teleostei – Terapontidae & Sciaenidae
Nerocila loveni	ectoparasitic	caudal peduncle	Teleostei – Leiognathidae, Haemulidae, Carangidae & Dorosomatidae
Nerocila milesensis	ectoparasitic	anal fin	Teleostei – Scorpaenidae
Nerocila monodi	ectoparasitic	body surface	Teleostei – Sparidae & Cichlidae
Nerocila munda	ectoparasitic	dorsal fin	Elasmobranchii (sharks) – Triakidae

(Continued)

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Nerocila orbignyi	ectoparasitic	body surface; fins	Teleostei – Mugilidae, Moronidae, Pleuronectidae, Cichlidae, Salmonidae, Sparidae, Soleidae, Gempylidae, Rhombosoleidae, Clupeidae, Batrachoididae, Lophiidae, Belonidae, Triglidae, Gadidae, Serranidae, Arripidae, Carangidae, Labridae, Scorpaenidae, Scombridae, Dactylopteridae, Girellidae, Molidae, Platycephalidae, Pomatomidae, Sillaginidae & Trachinidae; Holocephali – Chimaeridae & Callorhinchidae; Elasmobranchii (rays) – Rajidae
Nerocila phaiopleura	ectoparasitic	body surface	Teleostei – Sphyraenidae, Carangidae, Dussumieriidae, Chirocentridae, Clupeidae, Engraulidae, Pristigasteridae, Mugilidae, Ariidae, Plotosidae, Istiophoridae, Leiognathidae, Polynemidae & Scombridae
Nerocila philippensis	ectoparasitic	unknown	Teleostei (?)
Nerocila pigmentata	ectoparasitic	unknown	Teleostei (?)
Nerocila pulicatensis	ectoparasitic	body surface	Teleostei – Plotosidae & Bagridae
Nerocila recurvispina	ectoparasitic	body surface	unknown
Nerocila serra	ectoparasitic	body surface	Teleostei – Ariidae, Plotosidae & Bagridae; also found on sea snakes
Nerocila sigani	ectoparasitic	caudal fin	Teleostei – Siganidae
Nerocila sundaica	ectoparasitic	body surface; fins	Teleostei – Carangidae, Pristigasteridae, Sciaenidae & Terapontidae
Nerocila swainsoni	ectoparasitic	unknown	Teleostei (?)
Nerocila tenuipes	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Nerocila trichiura	ectoparasitic	body surface	Teleostei – Exocoetidae
Nerocila trivittata	ectoparasitic	unknown	Teleostei (?)
Nikione natalensis	ectoparasitic	branchial cavity	Decapoda – Processidae
Nococryptus angustus	ectoparasitic	unknown	unknown
Norileca borealis	ectoparasitic	body surface	Teleostei – Scombridae
Norileca indica	ectoparasitic	branchial cavity	Teleostei – Scombridae, Carangidae, Dorosomatidae, Nemipteridae & Leiognathidae
Norileca triangulata	ectoparasitic	body surface; branchial cavity	Teleostei – Carangidae, Scombridae, Dorosomatidae & Exocoetidae
Notophryxus clypeatus	ectoparasitic	branchial cavity	Mysida – Mysidae
Notophryxus globularis	ectoparasitic	unknown	Euphausiacea – Euphausiidae
Notophryxus lateralis	ectoparasitic	branchial cavity	Euphausiacea – Euphausiidae
Notophryxus lobatus	ectoparasitic	unknown	Mysida – Mysidae
Notophryxus lobus	ectoparasitic	branchial cavity	Mysida
Notophryxus longicaudatus	ectoparasitic	unknown	unknown
Notophryxus ocellatus	ectoparasitic	abdomen	Mysida – Mysidae
Notophryxus ovalis	ectoparasitic	unknown	unknown
Notophryxus ovoides	ectoparasitic	abdomen	Mysida – Mysidae
Oculophryxus bicaulis	ectoparasitic	eyestalk	Euphausiacea – Euphausiidae
Ogyridione caroliniana	ectoparasitic	branchial cavity	Decapoda – Ogyrididae
Olencira lamarckii	ectoparasitic	unknown	Teleostei (?)
Olencira praegustator	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Alosidae
Onisocryptus kurilensis	ectoparasitic	carapace	Myodocopida – Cypridinidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Onisocryptus ovalis	ectoparasitic	trunk	Myodocopida – Cypridinidae
Onisocryptus sagittus	ectoparasitic	brood cavity	Myodocopida – Cypridinidae
Onkokepon articulatus	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Onkokepon beibuensis	ectoparasitic	branchial cavity (?)	Decapoda – Leucosiidae
Onocryptus alatus	ectoparasitic	unknown	unknown
Onychocepon giardi	ectoparasitic	branchial cavity (?)	Decapoda – Pinnotheridae
Onychocepon harpax	ectoparasitic	branchial cavity (?)	Decapoda – Pinnotheridae
Onychocepon resupinum	ectoparasitic	branchial cavity (?)	Decapoda – Pinnotheridae
Orbimorphus constrictus	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Orbione angusta	ectoparasitic	branchial cavity (?)	Decapoda – Solenoceridae
Orbione aristei	ectoparasitic	branchial cavity (?)	Decapoda – Aristeidae
Orbione bonnieri	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Orbione digitata	ectoparasitic	branchial cavity (?)	Decapoda – Solenoceridae
Orbione halipori	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Orbione izuensis	ectoparasitic	branchial cavity (?)	Decapoda – Aristeidae
Orbione penei	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Orbione thielemanni	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Orophryxus shiinoi	ectoparasitic	carapace	Decapoda – Palaemonidae
Orthione furcata	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Orthione griffenis	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Orthione mesoamericana	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Osicryptus hirsutus	ectoparasitic	unknown	unknown

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Ourozeuktes bopyroides	mesoparasitic	flesh-burrowing	Teleostei – Monacanthidae
Ovobopyrus alphezemiotes	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Ovoionella obovata	ectoparasitic	branchial cavity	Decapoda – Munididae
Pagurion tuberculata	ectoparasitic	branchial cavity (?)	Decapoda – Calcinidae
Paguristione uniuropodus	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Paguritherium alatum	endoparasitic	visceral cavity	Decapoda – Paguridae
Paguritherium manggagaway	endoparasitic	visceral cavity	Decapoda – Calcinidae
Pagurocryptella holthuisi	ectoparasitic	branchial cavity (?)	Decapoda – Paguridae
Pagurocryptella paguri	ectoparasitic	branchial cavity	Decapoda – Parapaguridae
Palaemonellione cebuensis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Parabopyrella angulosa	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella angusta	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella australiensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella barnardi	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella bonnieri	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Parabopyrella choprai	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae & Hippolytidae (?)
Parabopyrella crenulata	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella cuspidata	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella delagoae	ectoparasitic	branchial cavity (?)	Decapoda
Parabopyrella distincta	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella elongata	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella essingtoni	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Parabopyrella hodgarti	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella incisa	ectoparasitic	branchial cavity (?)	Decapoda
Parabopyrella indica	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella intermedia	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella lata	ectoparasitic	branchial cavity	Decapoda – Alpheidae & Lysmatidae
Parabopyrella megatelson	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella mortenseni	ectoparasitic	branchial cavity	Decapoda – Alpheidae & Lysmatidae
Parabopyrella nierstraszi	ectoparasitic	branchial cavity	Decapoda – Lysmatidae
Parabopyrella pacifica	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Parabopyrella perplexa	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella richardsonae	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella saronae	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Parabopyrella setoensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella symmetros	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella tanyensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrella thomasi	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Parabopyriscus stellatus	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Parabopyrus kiiensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Paracepon nierstraszi	ectoparasitic	branchial cavity (?)	Decapoda (?)
Paracepon stebbingi	ectoparasitic	branchial cavity (?)	Decapoda – Inachidae
Paracymothoa astyanaxi	ectoparasitic	buccal cavity	Teleostei – Characidae
Paracymothoa parva	ectoparasitic	buccal cavity	Teleostei – Characidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Paracymothoa tholoceps	ectoparasitic	unknown	Teleostei – Erythrinidae
Paraeragia kiribatiensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Paragigantione americana	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Paragigantione indica	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Paragigantione papillosa	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Paragnathia formica	ectoparasitic	unknown	Teleostei – Anguillidae, Pleuronectidae, Clupeidae & Scombridae
Paranikione distorta	ectoparasitic	branchial cavity (?)	Decapoda – Processidae
Paranikione sibogae	ectoparasitic	branchial cavity (?)	Decapoda – Processidae
Parapagurion calcinicola	ectoparasitic	branchial cavity	Decapoda – Calcinidae & Diogenidae
Parapagurion imbricata	ectoparasitic	branchial cavity (?)	Decapoda – Parapaguridae & Diogenidae
Parapenaeon consolidata	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeon diatropa	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeon georgei	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeon lobulatum	ectoparasitic	branchial cavity (?)	Decapoda
Parapenaeon richardsonae	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeon sicyoniae	ectoparasitic	branchial cavity (?)	Decapoda – Sicyoniidae
Parapenaeonella coarctatum	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeonella distincta	ectoparasitic	branchial cavity	Decapoda – Penaeidae
Parapenaeonella expansa	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeonella minutopoda	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeonella secunda	ectoparasitic	branchial cavity (?)	Decapoda – Penaeidae
Parapenaeonella tertia	ectoparasitic	branchial cavity	Decapoda – Penaeidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Parapleurocrypta alphei	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Parapleurocrypta digitata	ectoparasitic	branchial cavity (?)	Decapoda (?)
Parapleurocryptella elasmonoti	ectoparasitic	branchial cavity (?)	Decapoda – Munidopsidae
Parapleurocryptella minuta	ectoparasitic	branchial cavity (?)	Decapoda – Chirostylidae
Parapodascon stebbingi	ectoparasitic	marsupium	Amphipoda – Uristidae
Parargeia ornata	ectoparasitic	branchial cavity (?)	Decapoda – Crangonidae
Paraspidophryxus terminalis	ectoparasitic	unknown	unknown
Parasymmetrione tuberculineata	ectoparasitic	branchial cavity	Decapoda – Diogenidae
Parasymmetrorbione bicauda	ectoparasitic	branchial cavity (?)	Decapoda – Solenoceridae
Parathelges aniculi	ectoparasitic	abdomen	Decapoda – Calcinidae
Parathelges cardonae	ectoparasitic	abdomen	Decapoda – Diogenidae
Parathelges carolii	ectoparasitic	abdomen	Decapoda – Diogenidae
Parathelges enoshimensis	ectoparasitic	abdomen	Decapoda – Paguridae
Parathelges foliatus	ectoparasitic	abdomen	Decapoda – Paguridae
Parathelges neotenuicaudis	ectoparasitic	abdomen	Decapoda – Paguridae
Parathelges occidentalis	ectoparasitic	abdomen	Decapoda – Diogenidae
Parathelges piriformis	ectoparasitic	abdomen	Decapoda – Paguridae
Parathelges racovitzai	ectoparasitic	abdomen	Decapoda – Diogenidae
Parathelges tumidipes	ectoparasitic	abdomen	Decapoda – Calcinidae
Parione ischyrandra	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parione lamellata	ectoparasitic	branchial cavity (?)	Decapoda
Parione pachychelii	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Parione paucisecta	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Parione pisidiae	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parionella decidens	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parionella elegans	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Parionella notexocha	ectoparasitic	branchial cavity (?)	Decapoda – Eumunididae
Parionella richardsonae	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parionina chinensis	ectoparasitic	branchial cavity (?)	Decapoda
Parionina pacifica	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Parioninella astridae	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parioninella liuruiyui	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Parioninella pacifica	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Pauperella rotunda	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Perezina gregaria	ectoparasitic; hyperparasitic	mantle cavity	Rhizocephala – Sacculinidae
Philostomella cigarra	ectoparasitic	buccal cavity	Teleostei – Characidae
Phyllodurus abdominalis	ectoparasitic	abdomen (?)	Decapoda – Upogebiidae
Pinnixion sexdecennia	endoparasitic	visceral cavity	Decapoda – Pinnotheridae
Pinnotherion setoensis	endoparasitic	visceral cavity	Decapoda – Inachidae
Pinnotherion vermiforme	endoparasitic	visceral cavity	Decapoda – Pinnotheridae
Pleonobopyrus kumanonadensis	ectoparasitic	abdomen	Decapoda – Crangonidae
Pleopodias diaphus	ectoparasitic	body surface; anterior to dorsal fin	Teleostei – Myctophidae
Pleopodias elongatus	ectoparasitic	unknown	Teleostei (?)
Pleopodias nielbrucei	ectoparasitic	unknown	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Pleopodias vigilans	ectoparasitic	unknown	Teleostei (?)
Pleurocrypta dubia	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pleurocrypta floridana	ectoparasitic	branchial cavity	Decapoda – Galatheidae
Pleurocrypta galateae	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pleurocrypta keiensis	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocrypta longibranchiata	ectoparasitic	branchial cavity	Decapoda – Galatheidae
Pleurocrypta macrocephala	ectoparasitic	branchial cavity	Decapoda – Porcellanidae
Pleurocrypta meridionalis	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocrypta microbranchiata	ectoparasitic	branchial cavity	Decapoda – Galatheidae
Pleurocrypta petrolisthis	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Pleurocrypta piriformis	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pleurocrypta porcellanaelongicornis	ectoparasitic	branchial cavity (?)	Decapoda – Porcellanidae
Pleurocrypta strigosa	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pleurocryptella altalis	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Pleurocryptella crassandra	ectoparasitic	branchial cavity (?)	Decapoda – Munidopsidae
Pleurocryptella fimbriata	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocryptella formosa	ectoparasitic	branchial cavity (?)	Decapoda – Sternostylidae
Pleurocryptella infecta	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocryptella laevis	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocryptella latilamellaris	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pleurocryptella shinkai	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Pleurocryptella superba	ectoparasitic	branchial cavity (?)	Decapoda – Munidopsidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Pleurocryptella wolffi	ectoparasitic	branchial cavity	Decapoda – Munidopsidae
Pleurocryptina indica	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pliophrixus philonika	ectoparasitic	abdomen (?)	Decapoda – Processidae
Plotor indus	ectoparasitic	unknown	Teleostei (?)
Podascon chevreuxi	ectoparasitic	marsupium (?)	Amphipoda – Ampeliscidae
Podascon dellavallei	ectoparasitic	marsupium (?)	Amphipoda – Ampeliscidae
Podascon haploopis	ectoparasitic	marsupium (?)	Amphipoda – Ampeliscidae
Podoniscus multidentatus	ectoparasitic	unknown	unknown
Pontobopyrus abyssorum	ectoparasitic	branchial cavity (?)	Decapoda – Crangonidae
Portunion bourdoni	endoparasitic	visceral cavity (?)	Decapoda – Epialtidae
Portunion conformis	endoparasitic	visceral cavity	Decapoda – Varunidae
Portunion flavidus	endoparasitic	visceral cavity	Decapoda – Grapsidae & Plagusiidae
Portunion kossmanni	endoparasitic	visceral cavity	Decapoda – Carcinidae
Portunion maenadis	endoparasitic	visceral cavity	Decapoda – Carcinidae
Portunion moniezii	endoparasitic	visceral cavity (?)	Decapoda – Polybiidae
Portunion salvatoris	endoparasitic	visceral cavity (?)	Decapoda – Polybiidae
Priapion fraissei	endoparasitic	visceral cavity	Decapoda – Polybiidae
Probopyrinella heardi	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Probopyrinella latreuticola	ectoparasitic	branchial cavity	Decapoda – Hippolytidae
Probopyrione plana	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Probopyriscus novempalensis	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Probopyrus aberrans	ectoparasitic	branchial cavity (?)	Decapoda – Thoridae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Probopyrus abhoyai	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus alcocki	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus annandalei	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus ascendens	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus bengalensis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus bithynis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus borrei	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus brachysoma	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus brevipes	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus buitendijki	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus demani	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus floridensis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus fluviatilis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus gangeticus	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus giardi	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus godaveriensis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus incertus	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus insularis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus iriomotensis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus marinus	ectoparasitic	branchial cavity (?)	Decapoda (?)
Probopyrus markhami	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus pacificensis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Probopyrus palaemoni	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus pandalicola	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus pica	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probopyrus prashadi	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus ringuelti	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probopyrus semperi	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Probynia obstipa	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Probynia pleurocephala	ectoparasitic	branchial cavity (?)	Decapoda
Probynia ramiroromani	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Procepon horridulum	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Procepon insolitum	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Procepon liuruiyui	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Prodajus bigelowiensis	ectoparasitic	marsupium	Mysida – Mysidae
Prodajus bilobatus	ectoparasitic	lower surface of thorax	Mysida – Mysidae
Prodajus curviabdominalis	ectoparasitic	marsupium	Mysida – Mysidae
Prodajus gastrosacci	ectoparasitic	marsupium	Mysida – Mysidae
Prodajus lobiancoi	ectoparasitic	marsupium	Mysida – Mysidae
Prodajus ostendensis	ectoparasitic	branchial cavity	Mysida – Mysidae
Prodajus ovatus	ectoparasitic	marsupium	Mysida – Mysidae
Progebiophilus assisi	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus bakeri	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus brevis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Progebiophilus bruscai	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Progebiophilus chapini	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus elongatus	ectoparasitic	branchial cavity	Decapoda – Callianassidae
Progebiophilus euxinicus	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Progebiophilus filicaudatus	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus insperatus	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus kensleyi	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus sinicus	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Progebiophilus upogebiae	ectoparasitic	branchial cavity	Decapoda – Upogebiidae
Progebiophilus villosus	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Prophryxus alascensis	ectoparasitic	unknown	unknown
Propseudione rhombicosoma	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Pseudione affinis	ectoparasitic	branchial cavity	Decapoda – Pandalidae
Pseudione ampla	ectoparasitic	branchial cavity (?)	Decapoda – Pandalidae
Pseudione andamanicae	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pseudione atlantica	ectoparasitic	branchial cavity (?)	Decapoda – Nephropidae
Pseudione borealis	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Pseudione callianassae	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Pseudione chiesai	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione chiloensis	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae
Pseudione clevai	ectoparasitic	branchial cavity	Decapoda – Stylodactylidae
Pseudione cognata	ectoparasitic	branchial cavity (?)	Decapoda – Crangonidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Pseudione crenulata	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione dohrni	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Pseudione fibriata	ectoparasitic	branchial cavity (?)	Decapoda
Pseudione galacanthae	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione hanseni	ectoparasitic	branchial cavity (?)	Decapoda – Axiidae
Pseudione hayi	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pseudione incerta	ectoparasitic	branchial cavity (?)	Decapoda
Pseudione indica	ectoparasitic	branchial cavity	Decapoda – Crangonidae & Pandalidae
Pseudione intermedia	ectoparasitic	branchial cavity	Decapoda – Paguridae
Pseudione itsindrae	ectoparasitic	branchial cavity (?)	Decapoda – Munidopsidae
Pseudione ivanklini	ectoparasitic	branchial cavity	Decapoda – Nephropidae
Pseudione japanensis	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae
Pseudione kossmanni	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pseudione longicauda	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Pseudione magna	ectoparasitic	branchial cavity (?)	Decapoda – Pandalidae
Pseudione minimocrenulata	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione munidae	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione murawaiensis	ectoparasitic	branchial cavity	Decapoda – Callianassidae
Pseudione nephropsi	ectoparasitic	branchial cavity (?)	Decapoda – Nephropidae
Pseudione parviramus	ectoparasitic	branchial cavity (?)	Decapoda – Pandalidae
Pseudione pontocari	ectoparasitic	branchial cavity	Decapoda – Crangonidae
Pseudione sagamiensis	ectoparasitic	branchial cavity (?)	Decapoda – Galatheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Pseudione serejoae	ectoparasitic	branchial cavity	Decapoda – Munididae
Pseudione stylopoda	ectoparasitic	branchial cavity	Decapoda – Stylodactylidae
Pseudione subcrenulata	ectoparasitic	branchial cavity (?)	Decapoda – Munididae
Pseudione tanimbarensis	ectoparasitic	branchial cavity (?)	Decapoda – Nephropidae
Pseudione tattersalli	ectoparasitic	branchial cavity	Decapoda – Pandalidae
Pseudionella akuaku	ectoparasitic	branchial cavity	Decapoda – Calcinidae
Pseudionella attenuata	ectoparasitic	branchial cavity	Decapoda – Paguridae
Pseudionella deflexa	ectoparasitic	branchial cavity	Decapoda – Paguridae
Pseudionella markhami	ectoparasitic	branchial cavity	Decapoda – Paguridae
Pseudionella spiropaguri	ectoparasitic	branchial cavity (?)	Decapoda – Paguridae
Pseudirona laeopsi	ectoparasitic	branchial cavity	Teleostei – Bothidae
Pseudostegias atlantica	ectoparasitic	abdomen	Decapoda – Diogenidae
Pseudostegias dulcilacuum	ectoparasitic	abdomen (?)	Decapoda – Diogenidae
Pseudostegias hapalogasteri	ectoparasitic	abdomen	Decapoda – Lithodidae
Pseudostegias mossambica	ectoparasitic	abdomen (?)	Decapoda
Pseudostegias otagoensis	ectoparasitic	abdomen	Decapoda – Diogenidae
Pseudostegias setoensis	ectoparasitic	abdomen	Decapoda – Calcinidae & Diogenidae
Pseudostegias trisagitta	ectoparasitic	abdomen	Decapoda – Calcinidae
Quadripediphryxus mayuzumii	ectoparasitic	abdomen	Decapoda – Alpheidae
Renocila alkoo	ectoparasitic	unknown	Teleostei (?)
Renocila bijui	ectoparasitic	body surface	Teleostei – Acanthuridae
Renocila bollandi	ectoparasitic	body surface	Teleostei – Scorpaenidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Renocila bowmani	ectoparasitic	body surface	Teleostei – Serranidae
Renocila colini	ectoparasitic	body surface	Teleostei – Apogonidae
Renocila curtipinnata	ectoparasitic	body surface (?)	Teleostei – Scorpaenidae
Renocila heterozota	ectoparasitic	body surface	Teleostei – Pomacentridae
Renocila indica	ectoparasitic	unknown	Teleostei (?)
Renocila kohnoi	ectoparasitic	body surface	Teleostei – Pomacanthidae
Renocila limbata	ectoparasitic	body surface (?)	Teleostei (?)
Renocila loriae	ectoparasitic	head; above eye	Teleostei – Apogonidae
Renocila ovata	ectoparasitic	body surface; posterior to pectoral fin	Teleostei – Pomacentridae
Renocila periophthalma	ectoparasitic	unknown	Teleostei – Gobiidae
Renocila plesiopi	ectoparasitic	body surface	Teleostei – Plesiopidae & Apogonidae
Renocila quadrata	ectoparasitic	body surface (?)	Teleostei – Acanthuridae, Zanclidae & Chaetodontidae
Renocila richardsonae	ectoparasitic	body surface	Teleostei – Mullidae
Renocila thresherorum	ectoparasitic	branchial cavity	Teleostei – Apogonidae, Cichlidae, Mugilidae & Soleidae
Renocila trillesi	ectoparasitic	body surface	Teleostei – Serranidae
Renocila waldneri	ectoparasitic	body surface	Teleostei – Serranidae
Renocila yamazatoi	ectoparasitic	head	Teleostei – Pomacentridae
Rhiothra callipia	ectoparasitic	unknown	Teleostei (?)
Rhopalione atrinicolae	ectoparasitic	abdomen	Decapoda – Pinnotheridae
Rhopalione incerta	ectoparasitic	branchial cavity (?)	Decapoda – Pinnotheridae
Rhopalione kali	ectoparasitic	abdomen	Decapoda – Pinnotheridae
Rhopalione sinensis	ectoparasitic	abdomen	Decapoda – Pinnotheridae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Rhopalione uromyzon	ectoparasitic	branchial cavity; abdomen	Decapoda – Pinnotheridae
Riggia acuticaudata	ectoparasitic	peritoneal cavity (?)	Teleostei – Loricariidae
Riggia brasiliensis	mesoparasitic	flesh-burrowing	Teleostei – Pimelodidae & Anostomidae
Riggia cryptocularis	ectoparasitic	peritoneal cavity	Teleostei – Characidae & Loricariidae
Riggia nana	ectoparasitic	peritoneal cavity (?)	Teleostei – Anostomidae, Apteronotidae, Sternopygidae & Hypopomidae
Riggia paranensis	ectoparasitic	peritoneal or pericardial cavity	Teleostei – Curimatidae
Riggia puyensis	ectoparasitic	peritoneal cavity	Teleostei – Loricariidae
Robinione brattstroemi	ectoparasitic	branchial cavity (?)	Decapoda – Callianassidae
Robinione overstreerti	ectoparasitic	branchial cavity	Decapoda – Callichiridae
Rolandoniscus serratus	ectoparasitic; hyperparasitic	unknown	Isopoda – Bopyridae
Ryukyua circularis	ectoparasitic	branchial cavity	Teleostei – Dorosomatidae
Ryukyua globosa	ectoparasitic	branchial cavity	Teleostei – Dorosomatidae
Scalpelloniscus binoculis	ectoparasitic	unknown	unknown
Scalpelloniscus nieli	ectoparasitic	prosoma	Scalpellomorpha – Scalpellidae
Scalpelloniscus penicillatus	ectoparasitic	unknown	Scalpellomorpha – Scalpellidae
Scalpelloniscus vomicus	endoparasitic	embedded in muscular tissue of peduncle or capitulum	Scalpellomorpha – Heteralepadidae & Poecilasmatidae; Calanticomorpha – Calanticidae
Schizobopyrina amakusaensis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Schizobopyrina andamanica	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Schizobopyrina bombyliaster	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Schizobopyrina brachytelson	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae

Table E1. Continued

Scientific name	**Parasitism type**	**Site of attachment**	**Hosts (Order/Class – Family)**
Schizobopyrina bruscai	ectoparasitic	branchial cavity (?)	Decapoda – Thoridae
Schizobopyrina cochinensis	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Schizobopyrina gracilis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Schizobopyrina kossmanni	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Schizobopyrina lobata	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae
Schizobopyrina miyakei	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Schizobopyrina platylobata	ectoparasitic	branchial cavity (?)	Decapoda – Palaemonidae
Schizobopyrina striata	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae & Thoridae
Schizobopyrina urocaridis	ectoparasitic	branchial cavity	Decapoda – Palaemonidae
Scyracepon australiana	ectoparasitic	branchial cavity (?)	Decapoda – Macrophthalmidae
Scyracepon biglobosus	ectoparasitic	branchial cavity	Decapoda – Majidae
Scyracepon distincta	ectoparasitic	branchial cavity	Decapoda – Corystidae
Scyracepon hawaiiensis	ectoparasitic	branchial cavity	Decapoda – Goneplacidae
Scyracepon levis	ectoparasitic	branchial cavity (?)	Decapoda – Epialtidae
Scyracepon oceanicum	ectoparasitic	branchial cavity (?)	Decapoda – Eriphiidae
Scyracepon polynesiensis	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Scyracepon pseudoliomerae	ectoparasitic	branchial cavity	Decapoda – Xanthidae
Scyracepon quadrihamatum	ectoparasitic	branchial cavity (?)	Decapoda – Majidae
Scyracepon thalamitae	ectoparasitic	branchial cavity (?)	Decapoda – Portunidae
Scyracepon tuberculosa	ectoparasitic	branchial cavity (?)	Decapoda – Epialtidae
Septembopyrina tozeumaophila	ectoparasitic	branchial cavity (?)	Decapoda – Hippolytidae
Shiinoella gracilipes	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Sigyn branchialis	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Smenispa convexa	ectoparasitic	buccal cavity; branchial cavity	Teleostei – Carangidae
Smenispa irregularis	ectoparasitic	buccal cavity	Teleostei – Sparidae, Psettodidae & Carangidae
Spathione aprosdovrima	ectoparasitic	abdomen	Decapoda – Cryptochiridae
Stegias andronophoros	ectoparasitic	abdomen (?)	Decapoda
Stegias angusta	ectoparasitic	abdomen (?)	unknown
Stegias clibanarii	ectoparasitic	abdomen	Decapoda – Diogenidae
Stegoalpheon kempi	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Stegoargeia lowisi	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Stegoargeia nierstraszi	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Stellatoniscus shieldsi	endoparasitic; hyperparasitic	marsupium (?)	Isopoda – Entoniscidae
Stellatoniscus tentaculus	endoparasitic; hyperparasitic	marsupium	Isopoda – Entoniscidae
Streptodajus equilibrans	ectoparasitic	unknown	Mysida – Mysidae
Synalpheion giardi	endoparasitic	visceral cavity (?)	Decapoda – Alpheidae
Synsynella choprai	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Synsynella deformans	ectoparasitic	branchial cavity	Decapoda – Alpheidae
Synsynella hayi	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Synsynella inoi	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Synsynella integra	ectoparasitic	branchial cavity (?)	Decapoda – Alpheidae
Telephryxus clypeus	ectoparasitic	antennules	Decapoda – Munidopsidae
Telotha henselii	ectoparasitic	branchial cavity; body surface	Teleostei – Loricariidae; Decapoda – Palaemonidae
Telotha indica	ectoparasitic	branchial cavity (?)	Teleostei (?)

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Telotha lunaris	ectoparasitic	branchial cavity	Teleostei – Apteronotidae
Telotha silurii	ectoparasitic	branchial cavity (?)	Teleostei (?)
Tenerognathia visus	ectoparasitic	unknown	Teleostei – Sebastidae
Tetragonocephalon lutianus	ectoparasitic	branchial cavity (?)	Teleostei – Lutjanidae
Thaumastognathia bicorniger	ectoparasitic	unknown	Elasmobranchii (sharks)
Thaumastognathia diceros	ectoparasitic	unknown	unknown
Thaumastognathia metaphone	ectoparasitic	unknown	Teleostei (?)
Thaumastognathia orectognathus	ectoparasitic	unknown	Teleostei (?)
Thaumastognathia tanseimaruae	ectoparasitic	unknown	unknown
Thaumastognathia wasmannia	ectoparasitic	unknown	Teleostei (?)
Thermaloniscus cotylophorus	ectoparasitic	unknown	unknown
Tiarinion fulvus	endoparasitic	visceral cavity	Decapoda – Epialtidae
Tiarinion texopallium	endoparasitic	visceral cavity	Decapoda – Epialtidae
Trapezicepon amicorum	ectoparasitic	branchial cavity (?)	Decapoda – Trapeziidae
Trapezicepon domeciae	ectoparasitic	branchial cavity	Decapoda – Domeciidae
Trisopodoniscus abyssorum	ectoparasitic	unknown	unknown
Tylokepon biturus	ectoparasitic	branchial cavity (?)	Decapoda – Epialtidae
Tylokepon bonnieri	ectoparasitic	branchial cavity (?)	Decapoda – Epialtidae
Tylokepon marianensis	ectoparasitic	branchial cavity	Decapoda – Epialtidae
Tylokepon micippae	ectoparasitic	branchial cavity (?)	Decapoda – Majidae
Tylokepon naxiae	ectoparasitic	branchial cavity	Decapoda – Majidae
Upogebione bidigitatus	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae

Table E1. Continued

Scientific name	Parasitism type	Site of attachment	Hosts (Order/Class – Family)
Upogebione ovalis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Upogebione phuketensis	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Upogebione tropica	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Upogebiophilus rhadames	ectoparasitic	branchial cavity (?)	Decapoda – Upogebiidae
Urobopyrus processae	ectoparasitic	branchial cavity	Decapoda – Processidae
Vanamea symmetrica	ectoparasitic	buccal cavity	Teleostei – Serrasalmidae, Pimelodidae, Doradidae, Cichlidae, Trichomycteridae & Gasteropelecidae
Xanthion spadix	endoparasitic	visceral cavity	Decapoda – Xanthidae
Zeuxokoma alphei	ectoparasitic	unknown	Decapoda – Alpheidae
Zeuxokoma elogata	ectoparasitic	abdomen	Decapoda – Nematocarcinidae
Zeuxokoma glabra	ectoparasitic	abdomen; thorax	Decapoda – Alpheidae
Zeuxokoma luetzeni	ectoparasitic	abdomen	Decapoda – Alpheidae
Zeuxokoma musaeformis	ectoparasitic	unknown	Decapoda – Pandalidae
Zeuxokoma setosa	ectoparasitic	unknown	Decapoda – Thoridae
Zonophryxus agassizi	ectoparasitic	unknown	unknown
Zonophryxus dodecapus	ectoparasitic	carapace	Decapoda – Pandalidae
Zonophryxus grimaldii	ectoparasitic	unknown	Decapoda – Pandalidae
Zonophryxus probisowa	ectoparasitic	unknown	Decapoda – Pandalidae
Zonophryxus quinquedens	ectoparasitic	carapace	Decapoda – Nematocarcinidae
Zonophryxus retrodens	ectoparasitic	carapace	Decapoda – Pandalidae
Zonophryxus trilobus	ectoparasitic	carapace	Decapoda – Pandalidae