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Diversity and geographical distribution of tapeworms of the order Diphyllobothriidea in Pinnipedia

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Annotation

The aim of the study was to obtain and elaborate information focused on tapeworms of the order Diphyllobothiidea and their hosts of marine environment (Pinnipedia). Faecal material of *Phoca vitulina* was obtained from the Seal Rehabilitation and Research Centre, Zeehondencrèche in Netherlands and and examined by two different coprological methods (flotation and sedimentation).

Declaration

I hereby declare that I have worked on my bachelor's thesis independently and used only the sources listed in the bibliography.

I hereby declare that, in accordance with Article 47b of Act No. 111/1998 in the valid wording, I agree with the publication of my bachelor thesis, in full version, to be kept in the Faculty of Science archive, in electronic form in publicly accessible part of the STAG database operated by the University of South Bohemia in České Budějovice accessible through its web pages.
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CONTENTS

1. INTRODUCTION

Tapeworms (Cestoda) belong to the exclusively parasitic group, called Neodermata (Lophotrochozoa: Platyhelminthes) and include almost 6000 species with the adult stages inhabiting predominantly a digestive tract of vertebrates (Caira & Littlewood 2013). They are traditionally divided in two subgroups, the Cestodaria composed of two primitive orders Amphilinidea and Gyrocotylidea and the rest of "true cestodes" represented by Eucestoda, comprising 17 orders (Khalil et al. 1994; www.tapewormdb.uconn.edu[1]). Phylogenetic relationships among the members of Eucestoda have not been clearly resolves so far, nevertheless they have been divided into "lower" bothriate cestodes (Bothriocephalidea, Caryophyllidea, Diphyllidea, Diphyllobothriidea, Haplobothriidea Litobothriidea, Spathebothriidea, Trypanorhyncha) and "higher" acetabulate cestodes (Cathetocephalidea, Cyclophyllidea, Lecanicephalidea, Nippotaeniidea, Proteocephalidea, Phyllobothriidea, Rhinebothriidea, Tetrabothriidea and polyphyletic "Tetraphyllidea") (www.tapewormdb.uconn.edu[1]). The most specious and derived order is Cyclophyllidea with around half of the known tapeworm species parasitizing mainly in birds and mammals. However, majority of the orders (9 out of 19) – Cathetocephalidea, Diphyllidea, Gyrocotylidea, Litobothriidea, Trypanorhyncha Lecanicephalidea, Phyllobothriidea, Rhinebothriidea, "Tetraphyllidea" parasitize in Elasmobranchs (Caira & Littlewood 2013).

This study is focused on species composition and distribution of members of one of the less known orders, Diphyllobothriidea, which parasitizes mainly in marine mammals, namely seals. Diphyllobothriidean tapeworms parasitize in all groups of tetrapods (mammals, birds, reptiles and amphibians), including man (Bray et al. 1994; Delyamure et al. 1985). This group of cestodes is cosmopolite with 74 % of species living in the marine environment, especially in intestine of mammals as seals and cetaceans (Kuchta et al. 2008).

The basis of the thesis was focused on the diversity and geographical distribution of tapeworms of the order Diphyllobothriidea in Pinnipeds. Furthemore, the faecal material of *Phoca vitulina* L. was collected and examined from the Netherlands, during an volunteering work in Research and Rehabilitation Center of seals. Faeces samples were elaborated for the presence of endoparasites.

2. LITERATURE SURVEY

2.1. Cestoda

Cestodes are parasitic flatworms with complex life cycles including usually two or more hosts. Adult cestodes inhabit almost exclusively digestive system of their definitive host (DH), all groups of vertebrates. The larval forms are harboured in organs as well as in intestine of their intermediate host (IH), mainly invertebrates, but in some cases also vertebrates (Elsheikha & Khan 2011).

The body structure of the cestodes is generally composed off two basic parts: scolex, and strobila (Caira & Littlewood 2013). The scolex ("head") is located anteriorly and is used to attach to the intestinal wall or spiral valve of its host. The attachment is often supported by additional attachment organs such as bothria or acetabulum (bothridia or suckers), or by additional specialized structures such as rostellum, apical organs, hooks or tentacles (Khalil et al. 1994). The cestode taxonomy is based mainly on the organisation and types of scoleces (Caira & Littlewood 2013; www.tapewormdb.uconn.edu[1]). The neck, an undifferentiated narrow zone, is usually localized between the scolex and the strobila. The neck may be of various length and contains germ cells, responsible for production of new segments. If the neck is absent, the germ cells occur in the posterior part of the scolex (Roberts & Janovy 2009). The rest of the tapeworm body is called strobila. The most of the cestodes are known to be segmented or proglottized, but there are also species with just a single set of genital organs in a strobilus (i.e. monozoic) such as Caryophyllidea, or their strobilus is composed off several proglottids (i.e. polyzoic), but is not segmented as Spathebothriidea (Caira & Littlewood 2013; www.tapewormdb.uconn.edu[1]). In case of segmented strobilus, the layout of segments is divided into two forms: craspedote (each segment is overlapped by the previous segment) or acraspedote (without overlapping segments).

Forming of segments is caused by asexual process known as strobilation. At this stage, segments increase in size and maturity, with the result of (usually) wider than long units carrying fully functional and active sexual organs (Elsheikha & Khan 2011). Mature proglottids situated at the end of strobila leave the body of oviparous tapeworm and migrate as independent, self- propelled segments (apolytic) or they pass in faeces out of the DH. Gravid segments leaving the body may disintegrate and release their eggs. In some species of tapeworms, proglottids are retained on the strobila (anapolytic) throughout the life of their host. In this case, eggs are released through uterine pores (Khalil et al. 1994; Elsheikha & Khan 2011).

The first embryonic form of the tapeworm develops within the tapeworm egg. These larvae may be divided into two groups based on the number of their embryonic hooks. Decatanths, also called lycophore, possesses 10 embryonic hooks and are present in Cestodaria. Six-hooked-larvae-hexacanth (or oncosphere) are known in all Eucestodes (Elsheikha & Khan 2011). The embryo possessing three pairs of hooks, also called coracidium, is covered by ciliated epithelium, intended for movement in water (Conn & Swiderski 2008).

The larvae (metacestodes) ingested by the specific IH hatch and develop into an immature stage. The stage called procercoid is always situated in the first IH. If the larval stage is harboured in invertebrate IH, tapeworm will be localized in haemocoel and develop to the procercoid form. The metacestodes harboured in the second IH, including both vertebrates and invertebrates, occur in different morphological types as plerocercus, cysticercus, plerocercoid or merocercoid (Chervy 2002).

As mentioned above, cestodes are usually harboured in two or more hosts. The two-host life cycle is typical for members of the genus *Taenia* Linnaeus, 1758 (Cyclophyllidea) or *Bothriocephalus* Rudolphi, 1808 (Bothriocephalidea), while the three-host life cycle is typical for members of the genus *Diphyllobothrium* Lühe, 1910 or *Spirometra* Faust, Campbell et Kellogg, 1929 (Diphyllobothriidea). Only few cestode species are able to develop in a single host, for example *Hymenolepis nana* (Siebold, 1852) (Cyclophyllidea) or *Archigetes* Leuckart, 1878 (Caryophyllidea).

Cestodes are almost exclusively hermaphrodites, usually in form of simultaneous hermaphroditism. The simultaneous hermaphrodites contain both male and female reproductive organs, mostly with faster ripening male organs (protandry). Few species (Cyclophyllidea: Anoplocephalidae, Schistotaeniidae, Hymenolepididae) are opposite, with the faster-growing female system (protogyny) (Warner 1975). It is considered that these two types of development prevent self-fertilization in the same segment (Khalil et al.1994). However, a few species are with a dioecious reproduction, such as *Infula macrophallus* Coil, 1955 (Cyclophyllidea).

Each segment of strobila usually contains one or rarely more sets of male and female reproductive systems (Khalil et al. 1994). The male reproductive organs include various amounts of testes linked to vas deferens carrying sperm to the terminal genitalia through a thin channel called vas efferens. Vas deferens opens into cirrus sac, in which the male copulatory organ called cirrus is localized. Female reproductive organs contain a single ovary which produces eggs. Formation of eggs is unconditionally supported by a vitellarium.

Vitellarium generates yolk-filled cells to nourish the developing eggs (embryos) and also compounds involving production of egg membrane. Vitellarium can also support forming of eggshell. Mature oocytes leave the ovary through the oviduct, often provided with a muscular orifice, known as sphincter or oocapt (Conn & Świderski 2008). Fertilization occurs most frequently between two adjacent tapeworms when the cirrus of both of them is connected through the genital pore and sperm cells (spermatozoa) are exchanged. Spermatozoa travelling from the genital pore, move from base of the vagina into the ootype. Some groups contain a vagina constituting a seminal receptacle which stores these male reproductive cells. The male and female ducts usually open into a common genital atrium through a common genital pore or separately through the male and female genital pores. The developing embryo enters the uterus after leaving the ootype (Khalil et al. 1994; www.tapewormdb.uconn.edu[1]).

The Eucestodes lack the digestive tract. Therefore they absorb nutrients through the specialized surface named tegument, an external cellular structure of the body (neodermis), covered by highly specialized microvilli, known as microtriches (Chervy 2009). The neodermis with its morphological variations of microtriches make up unique defining structures in cestodes. The external layer of microtriches consists of carbohydrate complex called glycocalyx. Microtriches are divided based on their size into two essential groups. The filitriches are specialized microtriches with the basal width ≤ 200 nm. Those with the basal width > 200 nm are known as spinitriches. There are three types of filitriches and 25 types of spinitriches (Chervy 2009).

The surface is responsible for absorption of bile salts, cations, for membrane transport of low molecular weight substances such as carbohydrates, amino acids, fatty acids, vitamins, and for pinocytosis (Cheng 1986). Tapeworms are unable to synthesize lipids which are significant for mechanism of reproduction. Therefore, the absorption of fatty acids is especially important (Mondal 2009).

However, at least one tapeworm species, termed as *Sanguilevator yearsleyi* Caira, Mega & Ruhnke, 2005 (Cathetocephalidea) is known to absorb blood cells. It is supposed, that they separate both leukocytes and erythrocytes within their scolex. They store white blood cells in spherical chambers and red blood cells in transverse channels. As mentioned before, cestodes are considered to absorb small molecules due to their lack of digestive tract. Therefore, it is improbable to consume these hematocytes with the aim of nutrition. The reason of consumption of blood cells by this parasite has not yet been established (Caira et al. 2005).

Additional function of tegument include protective cover to inhibit response from digestive enzymes of the external environment. The structure also acts as a sensory system for detection of the environmental conditions and target sites of anthelmintic drugs (Mansour 2002). At the level of morphological structures, it is supposed that microtriches help to prevent contact with host immune effector cells (Wedekind & Little 2004).

Process of absorbing of nutrients is as important as discarding waste materials. Cestodes use protonephridia, also termed “flame cells”, as a main functional unit of excretory system. They are attached to a tube cell, supported by microtriches, which help to move liquid through the tube. These “cup-shaped” flagellated cells regulate the osmotic pressure of tapeworm, and maintain its ionic balance (Ruppert et al.2004).

Cestodes belong to the group of acoelomates, which exhibit bilateral symmetry and have no body cavity. Therefore, the reproductive organs are supported by musculature. Muscles are located directly below the tegument in the form of several thin layers. There are three types of muscles: circular, oblique and longitudinal. Circular musculature occurs in periphery of tapeworm´s body with perpendicularly lying oblique tissues. Longitudinal muscles extend along the length of the cestodes body. Many tapeworms possess longitudinal muscle bundles located lengthwise from the scolex to the end of strobila, which separate the outer cortex and the inner center (medulla) of the body. Some cestodes contain a narrow, muscular enlargement (cephalic peduncle), supporting a posture of the scolex on the tapeworm´s body (www.tapewormdb.uconn.edu[1]).

2.1.1. Diphyllobothriidea Kuchta, Scholz, Brabec et Bray, 2008

The Diphyllobothriidea is an order of bothriate eucestodes characterised by presence of unarmed scolex with two dorsoventrally localised bothria (Kuchta et al. 2008). The scolex is usually round, without apical disc, except the genus *Tetragonoporus* Skryabin, 1961.

The scolex is usually attached to the neck, from which the strobila grows (Khalil et al. 1994). The strobila is segmented with mostly wider than long, anapolytic craspedote segments. Lack of segmentation is rare (*Ligula*, Bloch 1782). Each segment generally contains one set of male and female reproductive organs, except of some genera with multiple reproduction sets in a single segment such as *Diplogonoporus* Lönnberg, 1892 or *Tetragonoporus* (Kuchta et al. 2008). The testes are numerous, and the cirrus sac is covered by a thick muscular wall, and the proximal part of the vas deferens forms muscular external seminal vesicle. The copulatory organ, cirrus, is unarmed. Female reproductive organs contain a compact ovary and a ventral genital pore attached to a tubular uterus. The vitelline follicles are numerous, usually situated in cortical parenchyma (Kuchta et al. 2008).

The Diphyllobothriidea vary greatly in size. Most of them reach 1–2 m. One of the smallest species is *Diphyllobothrium wilsoni* Shipley, 1907 infecting leopard seal (*Hydrurga leptonyx* (de Blainville, 1820)) with high intensity, being aprroximately 10 mm long (Maltsev 2000). However, in less infected animals they could reach up to 5– 9 cm (Markowski 1952a). The largest species is *Tetragonoporus calyptocephalus* Skryabin, 1961 infecting the bile ducts of the sperm whale (*Physeter catodon* L.), and reaching over 30 m (Yurakhno 1992). The longest cestode infecting humans, *Diphyllobothrium latum* (Linnaeus, 1758), may reach the total length up to 25 m, but most frequently reaches 3–10 m (Scholz et al. 2009).

The life cycle of Diphyllobothriidea usually involves three hosts. A ciliated free-swimming aquatic larva (coracidium) hatches from the thick-walled egg developing in water. Then, the coracidium is eaten by a copepod (Crustacea) and harboured in its body cavity. These hexacanth develop in copepods to another stage named procercoid, which is infective for another host, usually a vertebrate (fish or amphibian). In infected vertebrates, a next larval stage called plerocercoid, develops. The adult diphyllobothriids parasitize in the digestive tract of tetrapodes, mainly marine mammals and birds including humans (Kuchta et al. 2008). The members of the genus *Tetragonoporus* Skryabin, 1961 invade a biliary duct of cetaceans (Kuchta et al. 2008). The two-host-life cycle occurs only in *Cephalochlamys namaquensis* (Cohn, 1906), with a single intermediate copepod host (*Thermocyclops infrequens* (Kiefer, 1929)) and a single DH, known as African clawed frog (*Xenopus* Daudin, 1802) (Thurston 1967; Jackson & Tinsley 2001).

Diphyllobothriidea is actually divided into three families (Kuchta et al. 2008):

I. Cephalochlamydidae Yamaguti, 1959
   - Genus: *Cephalochlamys* Jackson & Tinsley, 2001
   - Genus: *Paracephalochlamys* Jackson & Tinsley, 2001

II. Solenophoridae Monticelli et Crety, 1981
   - Genus: *Scyphocephalus* Riggenbach, 1898
   - Genus: *Bothridium* Blainville, 1824
   - Genus: *Duthiersia* Perrier, 1873

III. Diphyllobothriidae, Lühe, 1910
   - Genus: *Adenocephalus* Nybelin, 1931
   - Genus: *Baylisia* Markowski, 1952

Genus: *Baylisiella* Markowski, 1952

Genus: *Diphyllobothrium* Cobbold, 1858

Genus: *Diplogonoporus* Lönnberg, 1892

Genus: *Flexobothrium* Yurakhno, 1979

Genus: *Glandicephalus* Fuhrmann, 1921

Genus: *Ligula* Bloch, 1782

Genus: *Plicobothrium* Rausch & Margolis, 1969

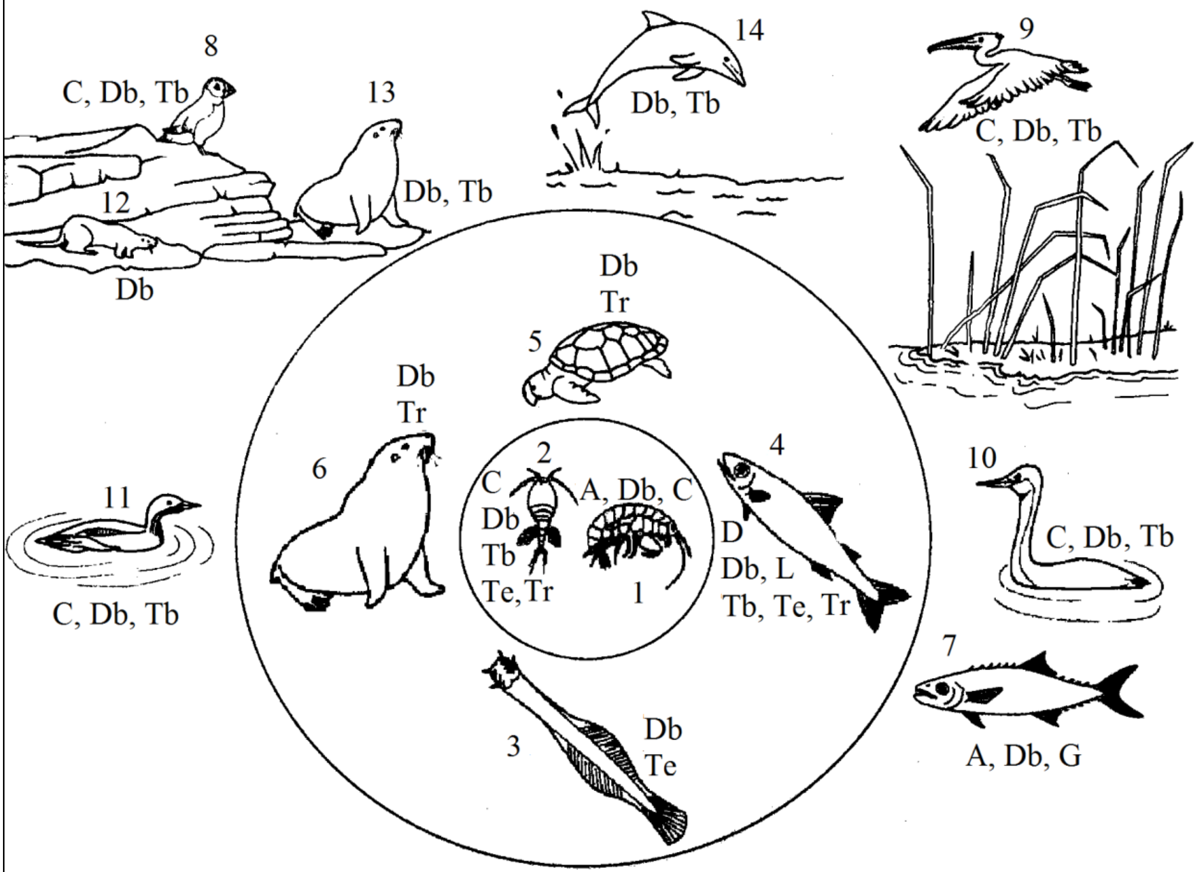
Genus: *Pyramicocephalus* Monticelli, 1890

Genus: *Schistocephalus* Creplin, 1829

Genus: *Spirometra* Faust, Campbell & Kellog, 1929

Genus: *Tetragonoporus* Skryabin, 1961

The family Cephalochlamydidae parasitizes African amphibians of the genus *Xenopus*. Tapeworms of the family Solenophoridae invade reptiles of Africa, Asia, Australia and South America and the members of the family Diphyllobothriidae colonize a wide range of birds and mammals worldwide (Kuchta et al. 2008). The majority of cestodes (including Diphyllobothriidean tapeworms) are invading animals living in the aquatic environment (Caira & Pickering 2013). The following scheme (Fig. 1.) shows tapeworm orders with three various categories of their regular hosts. These hosts are also common for the order Diphyllobothriidea.



Cestode orders	First IH (inner circle)	Second IH (middle circle)	Definitive host
A, Amphilinidea	1. Arthropoda:	3. Chaetognatha	7. Osteichthyes
C, Cyclophyllidea	Euphausiacea	4. Osteichthyes	8. Aves: Charadriiformes
D, Diphyllidea	2. Arthropoda:	5. Reptilia: Chelonia	9. Aves: Pelecaniformes
Db, Diphyllobothriidea	Copepoda	6. Mammalia:	10. Aves: Podicipediformes
G, Gyrocotylidea		Pinnipedia	11. Aves: Gaviiformes
L, Lecanicephalidea			12. Mammalia: Mustelidae
Tb, Tetrabothriidea			13. Mammalia: Pinnipedia
Te, Tetraphyllidea			14. Mammalia: Cetacea
Tr, Trypanorhyncha			

Fig. 1. Aquatic vertebrates and invertebrates serving as an IH (inner circle), second IH (middle circle) and DH (outside of circle) for cestodes (including Diphyllobothriidea) (adaptedd from Énumération des cestodes du plankton et des invertébrés marins by Dollfus R.P. 1976, Annales de Parasitologie Humaine et Comparee, 51, 207-22.)

2.2. Classification and evolution of Pinnipedia

Members of Pinnipedia are semi-aquatic, fin-footed marine mammals belonging to the order Carnivora, with sister groups of terrestrial carnivorous mammals (Yonezawa et al. 2009). Pinnipeds are divided into three monophyletic families: Phocidae, Otariidae and Odobenidae (Perrin et al. 2009). Phocidae consists of two monophyletic subfamilies Phocinae (Tab. 1.) and Monachinae (Tab. 2.), with 12 genera and 17 species described so far, while Otariidae comprises 7 genera and 14 species (Tab. 3.). In Odobenidae, the only living species is *Odobenus rosmarus* L. (Perrin et al. 2009; Yonezawa et al. 2009; Berta & Churchill 2012). Walruses are divided into two living subspecies: Atlantic walrus (*Odobenus r. rosmarus* L.) and Pacific walrus (*Odobenus r. divergens* (Illiger, 1811)), while both of them are distributed in northern hemisphere.

Tab. 1. List of the family Phocidae of the Phocinae Subfamily with their geographic distribution (Rice 1988; Wilson & Reeder 2005; Yonezawa et al. 2009; Berta & Churchill 2012).

Genus	Species	Geographic distribution
Cystophora	*Cystophora cristata* (Erxleben, 1777)	Arctic, North Atlantic North America (Canada), Iceland, Greenland
Erignathus	*Erignathus barbatus* (Erxleben, 1777)	Arctic- North America (Canada, Greenland), central Eurasia
Halichoerus	*Halichoerus grypus* (Fabricius, 1791)	Atlantic - North America, Europe (from Estonia to Denmark), Baltic Sea
Pagophilus	*Pagophilus groenlandicus* (Erxleben, 1777)	Arctic (Eastern Canada, Greenland, Iceland, Norway) North Atlantic
Phoca	*Phoca largha* Pallas, 1811	North Pacific (from Alaska to Japan, exlucding China)

Tab. 1. (Continued).

Genus	Species	Geographic distribution
Phoca	*Phoca vitulina* Linnaeus, 1758	Northern Hemisphere North Atlantic - from James & Hudson Bays (Canada) to Southern Greenland, USA (Massachusetts) East Atlantic - from Barents Sea to Portugal Pacific - west coastal area of North America, Eastern Asia- Hokaido (Japan)
Pusa	*Pusa caspica* Gmelin, 1788	Caspian Sea
	Pusa hispida (Schreber, 1775)	Arctic Ocean, Bering Sea, Northern Europe (Finland), Northern Baltic Sea Pacific Ocean (Kamchatka, Hokkaido) Northern Asia - Lake Ladoga (Russia)
	Pusa sibirica (Gmelin, 1788)	Lake Baikal (Russia)

Tab. 2. Species of the family Phocidae with the subgroup Monachinae and their geographic distribution (Rice 1988; Wilson & Reeder 2005; Yonezawa et al. 2009; Berta & Churchill 2012).

Genus	Species	Geographic distribution
Hydrurga	*Hydrurga leptonyx* (de Blainville, 1820)	Southern Ocean - South America, South Africa, Australia, New Zealand, Antarctica
*Leptonychote*s	*Leptonychotes weddellii* (Lesson, 1826)	Southern Ocean - Antarctica

Tab. 2. (Continued).

Genus	Species	Geographic distribution
Lobodon	*Lobodon carcinophaga* (Hombron & Jacquinot, 1842)	Southern Ocean - Antarctica
Mirounga	*Mirounga angustirostris* Gill, 1866	North Pacific - North America
	Mirounga leonina Linnaeus, 1758	Southern Ocean- Macquarie; Pacific - Chatham Islands; Atlantic - Falkland Islands, Valdez Peninsula
Monachus	*Monachus monachus* (Hermann, 1779)	Atlantic - Canary Islands Mediterranean, Black Sea
	Monachus schauinslandi Matschie, 1905	Pacific - Hawaiian Islands
Ommatophoca	*Ommatophoca rossii* Gray, 1844	Southern Ocean - Antarctica

Tab. 3. Geographic distribution of the family Otariidae (Brunner 2004; Berta & Churchill 2012; Higdon et al. 2007; Maloney et al. 2008; Repenning 1971; Wilson & Reeder 2005; Yonezawa et al. 2009; Waerebeek & Würsig 2008).

Genus	Species	Geographic distribution
Arctocephalus	*Arctocephalus australis* (Zimmermann, 1783)	South Ocean - Falkland Islands East Pacific - South America
	Arctocephalus forsteri (Lesson, 1828)	Pacific - New Zealand, Australia, Sub – Antarctic islands
	Arctocephalus gazella (Peters, 1875)	Southern Ocean - Antarctic
	Arctocephalus philippii (Peters, 1866)	East Pacific - The Juan Fernández Islands (Chile)

Tab. 3. (Continued).

Genus	Species	Geographic distribution
	Arctocephalus pusillus (Schreber, 1775)	Indian - South Africa Pacific - Australia, Tasmania; Atlantic Ocean, African coastal regions from Namibia to Algoa Bay (South Africa)
	Arctocephalus townsendi Merriam, 1897	East Pacific - Guadalupe Island (Mexico), Channel Islands (California)
	Arctocephalus tropicalis (Gray 1872)	Indian- Amsterdam, Crozet, Marion; Pacific - Macquarie; Atlantic - Gough, Tristan
Callorhinus	*Callorhinus ursinus* Linnaeus, 1758	Pacific (Canada, Mexico, Japan, USA, Russia) Bering Sea, Sea of Okhotsk
Eumetopias	*Eumetopias jubatus* (Schreber, 1776)	Pacific (Canada, China, Japan, Russia, USA)
Neophoca	*Neophoca cinerea* (Péron, 1816)	Australia
Otaria	*Otaria flavescens* Shaw, 1800	Coast of South America (Argentina, Brazil, Chile, Peru, Urugay, Panama, Ecuador (Galapagos Islands)
Phocarctos	*Phocarctos hookeri* (Gray, 1844)	Southern Ocean - Auckland, Campbell (New Zealand subantarctic islands)

Tab. 3. (Continued).

Genus	Species	Geographic distribution
Zalophus	*Zalophus californianus* (Lesson 1828)	Pacific - western North America
	Zalophus wollebaeki Sivertsen, 1953	Pacific - Galapagos Islands (Equador), Columbia

2.2.1. General characteristics

Pinnipeds differ from other marine mammals like cetaceans or sirenians in their ability of terrestrial locomotion. These carnivorous, amphibious mammals need to mate, give birth, suckle their young, moult and rest on land (Geraci & Lounsbury 2005). However, they obtain food mainly from marine environments, less frequently also from inland or tropical freshwater systems (www.britannica.com[2]).

The members of Phocoidea have torpedo-shaped bodies with a broad middle and tapered at the head and hindquarters. They use four limbs modified into webbed flippers for the movement. Pinnipeds swim by paddling their flippers, compared to sirenians and cetaceans moving their tails or flukes up and down. They tend to be slower swimmers than cetaceans (Shirihai & Jarrett 2006). On the other hand, pinnipeds are more flexible and agile, typically swimming at 9–28 km/h (Riedman 1990). Pinnipeds reach depths on average over 200 metres for not more than 10 minutes during diving (Stirling & Kooyman 1971; MacDonald 1984; Georges, et al. 2000). Elephant seals (genus *Mirounga* Gray, 1827) can reach depth of 1.5 km and can also dive regularly for more than an hour (Riedman 1990).

The body size varies from 1 to 5 m, reaching the weight from about 45 kg to 3000 kg (Berta 2009). Males and females differ in size on the basis of sexual dimorphism. The adult males in otariids such as southern elephant seals (*Mirounga leonina* L.) are significantly larger than females. They can reach the mass up to 4000 kg, compared to females weighing not more than 800 kg. Adult females of odobenids weigh generally two-thirds as much as males. In phocines, the males are generally little smaller than females. Sexual dimorphism also comprises differences in colour, development of appendages, thickness of fur or vocalization (Ralls & Mesnick 2009; Le Boeuf & Campagna 2013). These traits are present mostly in males, used in defense of females as well as defending of territories during breeding season. Most differences of secondary sex characteristics in males occur in polygynous species (Ralls & Mesnick 2009). The mating system of pinnipeds is also related to breeding on land or ice. Land- breeding otariids tend to be polygynous, as females gather to groups (Riedman 1990). Phocids and walruses use to be monogamous and include mostly ice- or water- breeding species. While otariids tend to return to the same place for many years, the ice- breeding seals use to change their breeding sites every season (Riedman 1990; Ralls & Mesnick 2009).

The lifespan of pinnipeds is generally 20–30 years, when females typically mature faster and live longer than males (Fay 1960; Berta 2012). The sexual maturity of these marine mammals varies among species, mostly attaining within 2–12 years (Riedman 1990).

All of pinnipeds, whether old or young, must be aware of predators both on land or underwater. Whereas they spend most of their time in water, they are hunted by killer whales (*Orcinus orca* L.) and few species of sharks, as a great white shark (*Carcharadon carcharias* L.). Their natural predatos on land are polar bears (*Ursus maritimus* Phipps, 1774) or terrestrial predators such as canids (Nowak 2005; Weller 2009; Brown et al. 2010).

As noted above, pinnipeds are widespread, mostly living in cold and nutrient-rich waters of Northern and Southern Hemispheres. Their natural habitat includes waters of Polar regions with temperatures below 20 °C. The average air temperature is generally lower than 10°C (Longton 1988). While most species live in coastal areas, several members inhabit freshwaters systems. The only exlusively freshwater species is the Baikal seal (*Pusa sibirica* (Gmelin, 1788)), endemic to the Lake Baikal (Reeves et al. 2002). Other seals, like the monk seals (genus *Monachus*) and few species of otariids, live in tropical and subtropical areas. Only two species have been reported from both, marine and freshwater ecosystems, the harbor seals (*Phoca vitulina* L.) and the ringed seal (*Pusa hispida* (Schreber, 1775)), respectively (Riedman 1990).

The digestive system of seals usually include enormously long small intestine compared to common carnivorous mammals. The length of small intestine of Southern Elephant Seal is 25–42 times the body length (Laws 1953). The length of the gut and content of water affect the passage of food, which usually runs about less than 5 hours (Helm 1984).

The diet of pinnipeds includes variety of fishes, cephalopods and other marine invertebrates (Riedman 1990; Hobson et al. 1997). The leopard seal represents an exception, feeding on penguins or other seals, especially pups of crabeatear seals (*Lobodon carcinophaga* (Hombron & Jacquinot, 1842)) (Riedman 1990; Siniff & Bengtson 1977). There are also other feeding specialists such as pacific walrus (*Odobenus rosmarus divergens* (Illiger, 1815)) or atlantic walrus (*Odobenus rosmarus rosmarus* L.), which are main predators of bivalve mollusks in the Arctic (Fukuyamaa & Olivera 1985). Pinnipeds are generally known to prey and feed underwater. The pattern of consumption depends on the species of seal and size of their prey. Too heavy seal catches are pulled out of the water and processed on land (Roffe & Mate 1984). Walruses typically ingest their prey directly in water by suction feeding (Berta 2012).

3. MATERIAL AND METHODS

3.1. Literature review

Information material for this work was obtained from majority of articles including data of the order Diphyllobothriidea related to Carnivoran families of Pinnipedia. The resources were obtained from databases as NHM, CiNii, BHL, BioMedSearch, CJO, GoogleBooks, HathiTrust, JSTOR, NRC Reseach Press, PubMed, ScienceDirect, SpringerLink, Taylor & Francis, WOS. The keywords of the publications were processed using Endnote Basic software. The original version of the data has been reduced because some records were duplicated or did not contain the necessary information. The literarure survey included study of over 150 publications focused on geographical distribution and prevalence of tapeworms infecting seals. The relationships between seals and tapeworms of the order Diphyllobothriidea were possible to determine due to the obtained data compared to the information of pinnipeds.

3.2. Collection of material

Due to the possibility to work as a volunteer at the Seal Rehabilitation and Research Centre (SRRC), Zeehondencrèche located in Pieterburen, the Netherlands (www.zeehondencreche.nl[3]), for two months, I also had an opportunity to gather material in the field for this work.

The fieldwork included fresh faecal material collection during an internship in the SRRC. In the agreement with the veterinarians in the SRRC, the faecal sampling from seals placed at the Centre was approved. Samples were transferred to the Faculty of Science, University of South Bohemia in České Budějovice (Czech Republic) after finishing the work, where they were analyzed under supervision of specialists.

The Center works to save injured, weakened or sick wild seals and release them back to the nature for over 40 years (http://www.zeehondencreche.nl/historie). The internship lasted 2 months (from 16.8.to 10.10.), when members of the SRRC mostly took care of juveniles of Common seal (*Phoca vitulina*). In order to keep all important aims of the Centre (to rescue, cure and release the seals in to the wild), it was necessary to maintain strict hygiene protocols, nutritional and medical schemas with the seal patients. To keep the seals wild and stress free, it was important to avoid human interaction as much as possible.

My work concerned the Seal Care Department, where direct contact with seals was necessary. This work comprised mostly 2 or more feedings of seal patients per day, and extense morning sanitation of all areas in contact with seals and people working with them. Before entering the enclosure where the seal is housed, visual check of the health status was necessary. When considering negative status of the seal patient, it was necessary to adapt to the situation and take action, which usually involved closer contact with the animal (measurement of body temperature, giving medication and wound cleaning). Due to these facts, it was possible to collect samples during labour. The collection of the samples was discussed and coordinated by veterinary experts of the SRRC, and it was always personally agreed by a nurse in a given situation.

At first, faecal samples were gathered from new seals, which arrived into the SRRC during a period of my internship. All patients of *Phoca vitulina* were captured from the locality of Wadden Sea (Zuid Holland, Friesland, Vlieland, Noord Holland, Schiermonnikoog and Terschelling), due to their poor health condition. Their age was estimated under one year (juveniles), except one case of adult harbour seal.Faecal material was collected immediately after intake, and then after 24-48 hours or later (if possible). During intake were given anti-parasitics (Praziquantel, Mebendazole) to seals, to treat cestodes, nematodes, trematodes or other diseases. For sampling, nitrile powder-free gloves were used. Faecal material was placed in sampling bottles filled up with pure ethanol at room temperature (20-23 °C / 68-73.4 °F). After the internship, a total of 60 faecal samples from 20 individuals (70% males, 30% females) were coprologically analysed by two qualitative coprological concentration techniques (Flotation, Sedimentation) for the presence of endoparasites of the order Diphyllobothriidea.

3.3. Coprological examination

Faeces were examined by two different coprological methods, flotation and sedimentation, to examine the parasitofauna of the digestive tract of seals, focusing on parasites of the order Diphyllobothriidea. During this research the attention was payed especially on sensitivity and efficiency of both methods focused on the above mentioned helminths.

3.3.1. Flotation

Cestodes of the order Diphyllobothriidea can be diagnosed by identifying of their eggs or proglottids from faeces. Flotation is one of the standard parasitological methods for separation components of stool with different buoyancy. Less dense material as helminth eggs, cysts, oocysts, proglottids or larval forms are concentrated on the surface of the faecal float solution (with an appropriate specific gravity), while the heavier parts of the faecal material are located at the bottom (Dryden et al. 2005). We used Sheather´s sucrose solution of the specific gravity 1.30 as a flotation fluid.

Sheather´s sucrose solution of the specific gravity 1.30 was prepared by boiling 1 kg of granulated sugar dissolved in 700 ml of tap water. After cooling down, the mixture was enriched with 10 ml of liquid phenol for stabilization and durability.

Flotation apparatus was composed of a stand, nylon tea strainer, laboratory clamp holder, ring clamp and glass test tubes without cap. Faeces were homogenized in the original homeopathic bottle by shaking or with tweezers. Approximately 2 g of the mixture was poured through a tea strainer into the test tube. The rest of faecal material stuck on the nylon sieve was poured through with tap water to fill the tube ca. 1.5 cm below the rim. Such prepared samples were centrifuged for 10 minutes at 1106, 82 g. The supernate was poured. The sediment was mixed with a small amount of Sheather´s sugar solution and subsequently filled with it ca. 1 cm below the rim of the test tube. Samples were then centrifuged for another 10 minutes at 1106, 82 g and then were prepared for light microscopy.

For the microscopy, the following equipment was required: test tubes with samples processed by flotation, test tube rack, light microscope (Olympus CX31), microslides, coverslips, inoculation loop, cotton, flask and tap water. From the test tube, a drop of the membrane from the top of the flotated liquid was picked with an inoculation loop and transferred on a microslide. This process was repeated with another drop and then the microslide was covered with a coverslip. Such a native mount was microscoped and the results consulted with specialists. Eggs were measured and photographed by the specialists using an Olympus BX53 light microscope equipped with digital camera and OLYMPUS cellSens Standard 1.13 imaging software. All measurements were given in μm. Prevalence was estimated as the percentage of infected seals.

3.3.2.Sedimentation

The sedimentation technique is based on removing light unintended fragments from the faecal material. Heavy components as eggs of trematodes (e.g. *Fasciola hepatica* Linnaeus, 1758), oocysts of Conoidasida (e.g. *Eimeria leuckarti* Flesch, 1883), or larvae of nematodes (e.g. *Trichinella spiralis* Owen, 1835) fall to the bottom of a faecal suspension (Leiper 1949; Bauer 1988; Kaufmann 1996; Baker 2007). This coprological method is also commonly used to diagnose eggs of cestodes (e.g. *Diphyllobothrium latum* (Linnaeus, 1758)) (Thienpont et al. 1979; Zajac & Conboy 2012).

For the sedimentation technique, following equipment was used: glass test tubes, cork stoppers, test tube rack, glass funnel, gauze, wooden spatulas, 3 ml plastic pipettes, laboratory hood, AMS III solution (SG 1.080), Triton solution, and ether (Hunter et al. 1948).

The AMS solution was prepared by dissolving of 115.2 g anhydrous Na_2SO_4 in a medium consisting of 540 ml HCl and 660 ml H_2O. The Triton solution consisted of 16.5 ml Triton X-100 and 33.5 ml H_2O.

Faeces were homogenized in the original sampling bottle by shaking or with a wooden spatula. The test tube was filled up with approximately 3 g of faeces samples fixed by ethanol and 6 ml of AMS solution. The compound was poured through the funnel with gauze to another clean test tube. The mixture was filled up with 3 drops of the Triton solution and 3 ml of diethylether inside the safety hood. Such prepared samples were closed with cork stoppers, homogenized by shaking and centrifuged for 2 minutes at 600 g. The supernatant was poured off. The sediment was used for light microscopy; after being slightly stirred, several drops were put on a microslide and examined using 40x10 and 60x10 magnification.

The results were consulted with specialists. Eggs were measured and photographed with an Olympus Camedia C-5060, light microscope equipped with digital camera and Quick PHOTO MICRO 2.3 imaging software. All measurements are given in μm. Prevalence was calculated as the percentage of infected seals.

4. RESULTS

4.1. Literature review

The publications, containing information of the order Diphyllobothriidea invading the digestive tract of Phocidae, Otariidae and Odobenidae, were elaborated and reduced due to unclear, false or duplicated the same data. Relevant information was identified from over 150 publications and modified to required categories. The following table (Tab. 4.) is showing specific species of Diphyllobothriidean tapeworms invading seals (Phocidae) and sea lions (Otariidae).

Almost all species of the family Diphyllobothriidae infect Phocids and Otariids, except the genera *Plicobothrium* and *Spirometra*. The Otariids species are predominantly infected by *Adenocephalus pacificus* (Nybelin, 1931), which is not invading any member of Phocids. More than a half of the given species of Diphyllobothriideans invade only one species of seal or sea lion. The species *Baylisia baylisi* Markowski, 1952, *B. supergonoporis* Yurakhno, 1989 and *D. lobodoni* Yurakhno & Maltsev, 1994 infect only *Lobodon carcinophagus*. Other member of Phocidae, *Mirounga leonina* is the only host within Phocids and Otariids for *Baylisiella tecta* (Linstow, 1892) and *Flexobothrium microovatum* Yurakhno, 1989. *D. archeri* Leiper & Atkinson, 1914 and *Glandicephalus perfoliatus* (Rennie & Reid, 1912) are invading only *Leptonychotes weddellii*. The Hawaiian monk Seal (*Monachus schauinslandi*) is the only host for *D. cameroni* Rausch, 1969, *D. minutus* Andersen, 1987 and *D. rauschi* Andersen, 1987. Other species of the genus *Glandicephalus* invading seals and sea lions, *G. antarticus* (Baird, 1853), has the only pinniped host, *Ommatophoca rossii*. *Diphyllobothrium pterocephalum* Delyamure & Skryabin, 1966 parasitizes only *Cystophora cristata*. The only tapeworm representing the genus *Ligula* in Phocids is *L. colymbi* Zeder, 1803 harboured by *Phoca caspica*. This endemic seal to the Caspian Sea is only pinniped host also for *D. phocarum* Delyamure, Kurochkin & Skryabin, 1964 (Berta et al. 2006). The leopard seal (*Hydrurga leptonyx*) is the only pinniped host to *D. pseudowilsoni* Wojciechowska & Zdzitowiecki, 1995. Other species of diphyllobothriidean tapeworms invade more than one species of Phocidae or Otariidae. A detailed description of the geographical distribution of the Phocidae and Otariidae host species is given below (Tab. 5.). In publications occur unspecified species of parasite, *D. sp.* Cobbold, 1858, which are mentioned in both lists only in case of new locations of tapeworm (genus: *Diphyllobothrium*) in a host.

Due to odobenids are hosts probably only for 4 species of the order Diphyllobothriidea, the next table (Tab. 6.) was made separately.

The only known genus of the family Diphyllobothriidea, which infect walruses (Odobenidae), is *Diphyllobothrium*. The following species are mentioned: *Diphyllobothrium cordatum*, *D. latum*, *D. fayi* n. sp. Rausch 2005 and *D. roemeri* Zschokke 1903. Common diphyllobothriidean parasites in walruses are *D. cordatum* and *D. fayi*, while *D. fayi* invades only subspecies *Odobenus rosmarus divergens*. Hilliard and Douglas (1972) studied unspecified species of the genus *Diphyllobothrium* which was localized in walrus at Kodiak Island. Species *D. roemeri*, *D. latum* in walrus were mentioned by Dailey (1975) with unknown locality. Another case of no locality of *D. roemeri* in intestine of walrus was written by Lauckner (1985).

Tab. 4. Tapeworms of the order Diphyllobothriidea invading Phocidae and Otariidae.

Host / Tapeworm	*Hydrurga leptonyx*	*Leptonychotes weddellii*	*Lobodon carcinophaga*	*Mirounga angustirostris*	*Mirounga leonina*	*Monachus monachus*	*Monachus schauinslandi*	*Ommatophoca rossii*	*Cystophora cristata*	*Erignathus barbatus*	*Pagophilus groenlandicus*	*Phoca largha*	*Phoca vitulina*	*Pusa caspica*	*Pusa hispida*	*Arctocephalus australis*	*Arctocephalus gazella*	*Arctocephalus philippii*	*Arctocephalus pusillus*	*Arctocephalus tropicalis*	*Callorhinus ursinus*	*Eumetopias jubatus*	*Neophoca cinerea*	*Otaria flavescens*	*Zalophus californianus*	*Zalophus wollebaeki*
Adenocephalus pacificus																+	+	+	+	+	+	+	+	+	+	+
Baylisia baylisi			+																							
B. supergonoporis			+																							
Baylisiella tecta					+																					
Diphyllobothrium archeri		+																								
D. cameroni							+																			
D. cordatum										+	+	+	+									+				
D. ditremum		+											+		+											
D. elegans						+			+																	
D.hians						+				+			+		+											
D. lanceolatum										+	+		+		+							+				
D. lashleyi		+						+																		
D. lobodoni			+																							
D. minutus							+																			
D. mobile		+						+																		
D. phocarum														+												
D. pseudowilsoni	+																									
D. pterocephalum									+																	
D. rauschi							+																			
D. quadratum	+	+	+																							

Tab. 4. Continued.

D. scoticum	+																									
D. schistochilos										+	+		+													
*D. sp**				+			+						+	+												
D. wilsoni	+	+	+					+																		
Diplogonoporus tetrapterus									+	+	+	+	+		+						+	+				
Flexobothrium microovatum					+																					
Glandicephalus antarcticus								+																		
G. perfoliatus		+																								
Pyramicocephalus phocarum									+	+		+	+		+						+	+				
Ligula colymbi														+												
Schistocephalus solidus															+											

* Unspecified Diphyllobothrium with previously not mentioned location of infecting the given Phocid.

Tab. 5. List of diphyllobothriidean parasites invading Phocidae and Otariidae with their geographical distribution.

Parasite	**Host**		**Locality**		**References**
Species	**Subfamily**	**Species**	**Ocean**	**Land/ Island/ Archipelago/Sea**	
Adenocephalus pacificus	Otariinae	*Arctcocephalus australis*	Atlantic Ocean	Isla Arce	Hernández-Orts et al. 2013
				Isla de Lobos	Morgades et al. 2006
				Northern Patagonia	Hernández-Orts et al. 2013
			Pacific Ocean	Galapagos Islands	Dailey 1975
				Robinson Crusoe Island	Nybelin 1931
		Arctocephalus gazella	Southern Ocean	Avian Island	Rengifo-Herrera 2013
				South Shetland	Rengifo-Herrera 2013
				King George Island	Rengifo-Herrera 2013

Tab. 5. Continued.

A. pacificus	Otariinae	*Arctocephalus philippii*	Pacific Ocean	Alejandro Selkirk Island/ Juan Fernández Islands	Cattan et al. 1980, Sepulveda & Alcaino 1993
		Artocephalus pusillus	Atlantic Ocean	Namibia	Pansegrouw 1990
				South Africa	Delyamure & Parukhin 1968
			Indian Ocean	Lady Julia Percy Island	Drummond 1937
		Arctocephalus tropicalis	Atlantic Ocean	Cape Town	Shaughnessy & Ross 1980
				Gough Island	Bester 1989
				Richards Bay-Natal	Shaughnessy & Ross 1980
		Callorhinus ursinus	Pacific Ocean	California Coast /Año Nuevo Island	Gerber et al. 1993
				Kamchatka	Cholodkovsky 1914
				Hokaido	Maejima et al. 1981
				Honshu	Machida 1969, Yamaguti 1951
				Russian Far East	Afanassjew 1941
			Pacific Ocean,	St. George Island/ Pribilof Islands	Stiles 1899
				St. Paul´s Island	Wardle et al. 1947, Kuzmina et al. 2015
			Pacific Ocean,	Tuleniy Island	Chupakhina 1971, Krotov & Delyamure 1952

Tab. 5. Continued.

A. pacificus	Otariinae	*Eumetopias jubatus*	Pacific Ocean	Aleutian Islands	Dailey 1975
				Alaska	Fay et al. 1978
				California Coast/ Año Nuevo Island	Dailey & Hill 1970
				Oregon Coast	Stroud 1978
				Vancouver Island	Margolis 1956
				Bering Sea	Shults 1986
				Sea of Okhotsk	Dailey 1975
		Neophoca cinerea	Indian Ocean	Pearson Islands	Johnston 1937
		Otaria flavescens	Atlantic Ocean	Isla de Lobos	Cattan et al. 1977. Morgades et al. 2006
				Northern Patagonia	Hernández-Orts et al. 2013
			Pacific Ocean	(Islotes Los Leones) Guañape Islands	Baer 1969, Miranda et al. 1968
				Isla Santa Maria	George-Nascimento & Carvajal 1981
				Juan de Marcona	Tantalean 1993
			Southern Ocean	Falkland Islands	Baylis & Hamilton 1934
		Zalophus californianus	Pacific Ocean	California Coast/ Año Nuevo Island	Dailey & Hill 1970
		Zalophus wollebaeki		Galapagos Islands	Dailey 1975
Baylisia baylisi	Monachinae	*Lobodon carcinophaga*	Southern Ocean	South Shetland Islands / Graham Land	Markowski 1952a

Tab. 5. Continued.

B. baylisi	Monachinae	*Lobodon carcinophaga*	Southern Ocean	King George Island/ South Shetland Islands	Wojciechowska & Zdzitowiecki 1995
				Balleny Islands (D'Urville Sea)	Yurakhno & Maltsev 1997
Baylisia supergonoporis		*L. carcinophaga*		Balleny Islands (D'Urville Sea)	Yurakhno 1989a, Yurakhno & Maltsev 1997
Baylisiella tecta		*Mirounga leonina*	Southern Ocean	South Georgia	Linstow 1892, Markowski 1952b
				King George V Land	Johnston 1937
				Adelie Land	Johnston 1937
				Queen Mary Land	Johnston 1937
Diphyllobothrium archeri		*Leptonychotes weddellii*	Southern Ocean	Balleny Islands (D'Urville Sea)	Maltsev & Zhdamirov 1995
				Cape Denison	McEwin 1957
				Commonwealth bay	Johnston 1937
				Falkland Islands	Maltsev & Zhdamirov 1995
				Graham Land	Markowski 1952b
				King George V Land	McEwin 1957, Wojciechowska & Zdzitowiecki 1995
				McMurdo Sound	Beverley-Burton 1971
				Ross Sea	Maltsev & Zhdamirov 1995
				South Georgia	Maltsev & Zhdamirov 1995

Tab. 5. Continued.

D. archeri	Monachinae	*Leptonychotes weddellii*	Southern Ocean	South Shetland	Markowski 1952b, Maltsev & Zhdamirov 1995, Wojciechowska & Zdzitowiecki 1995
				Palmer Archipelago	Markowski 1952b
D. cameroni		*Monachus schauinslandi*	Pacific Ocean	Midway Atoll/Hawaii	Andersen 1987, Rausch 1969
D. cordatum	Phocinae	*Erignathus barbatus*	Arctic Ocean	Bernard Habour	Cooper 1921
				Novaya Zemlya (west coast)	Vagin 1933
				Disko Island	Krabbe 1868
				Svalbard	Markowski 1952a
			Pacific Ocean	St. Lawrence Island	Hilliard 1960, Fiscus et al. 1976
		Pagophilus groenlandicus	Arctic Ocean	Disko Island	Ariola 1899
				Svalbard	Markowski 1952a
		Phoca largha	Arctic Ocean	Alaska	Shults 1982
		Phoca vitulina	Atlantic Ocean	Kattegat-Skagerrak/ Baltic Sea	Heide-Jorgensen 1992
				Wadden Sea	Strauss et al. 1991
			Arctic Ocean	Svalbard	Zschokke 1903
	Otariinae	*Eumetopias jubatus*	Pacific Ocean	Oregon Coast	Stroud 1978
D. ditremum	Monachinae	*L. weddellii*	Southern Ocean	McMurdo Sound	Nieland 1962

Tab. 5. Continued.

D. ditremum	Phocinae	*Phoca vitulina*	Pacific Ocean	Alaska	Margolis & Dailey 1972
		Pusa hispida	Atlantic Ocean	Lake Saimaa	Sinisalo et al. 2003
D. elegans	Monachinae	*Monachus monachus*		St.George Arm (Black Sea)	Schnapp et al. 1962
	Phocinae	*Cystophora cristata*	Arctic Ocean	Disko Island	Krabbe 1868
D. hians	Monachinae	*M. monachus*	Atlantic ocean	Genoa (Italy)	Ariola 1900
				Tunis	Stossich 1895
	Phocinae	*Erignathus barbatus*		Island	Diesing 1850
			Arctic Ocean	Svalbard	Markowski 1952a
		Phoca vitulina	Atlantic Ocean	Mecklenburg (Baltic Sea)	Braun 1891
				Warnemünde (Baltic Sea)	Matz 1892
		Pusa hispida		Gryphiae (Baltic Sea)	Diesing 1850
D. lanceolatum		*E. barbatus*	Arctic Ocean	Disko Island	Krabbe 1868
				Chukchi Sea	Cooper 1921
				Kara Sea	Stunkard & Schoenborn 1936
				Kotelny Island	Linstow 1905
				Novaya Zemlya	Vagin 1933
				Taymyr Island	Linstow 1905

Tab. 5. Continued.

D. lanceolatum	Phocinae	*Erignathus barbatus*	Arctic Ocean	Svalbard	Fiscus et al. 1976, Guiart 1935, Markowski 1952a, Zschokke 1903,
			Pacific Ocean	Bering Sea	Cooper 1921. Lyster 1940
				Sea of Okhotsk	Cooper 1921, Hilliard 1960, Shulman & Popov 1982
				St. Lawrence Island	Hilliard 1960, Popov 1975, Delyamure & Popov 1975, Delyamure et al. 1976, Stunkard & Schoenborn 1936
		Pagophilus groenlandicus	Arctic Ocean	Baffin Island	Lyster 1940
		Phoca vitulina		Kvichak River	Rausch & Hilliard 1970
			Pacific Ocean	Russian Coast	Popov 1982
		Pusa hispida	Arctic Ocean	Novaya Zemlya	Vagin 1933, Delyamure & Alekseev 1965
			Pacific Ocean	Sea of Okhotsk	Krotov & Delyamure1952
	Otariinae	*Eumetopias jubatus*		Kuril Islands/ Sea of Okhotsk	Kovalenko 1975
D. lashleyi	Monachinae	*Leptonychotes weddellii*	Southern Ocean	Balleny Islands (D'Urville Sea)	Leiper & Atkinskon 1914, Maltsev 1995

Tab. 5. Continued.

D. lashleyi	Monachinae	*Leptonychotes weddellii*	Southern Ocean	Bellingshausen Sea	Leiper & Atkinskon 1914, Maltsev & Zhdamirov 1995
				Graham Land	Leiper & Atkinskon 1914, Maltsev & Zhdamirov 1995
				Ross Sea	Leiper & Atkinskon 1914, Maltsev & Zhdamirov 1995
				South Shetland	Leiper & Atkinskon 1914, Maltsev & Zhdamirov 1995
				Weddell Sea	Leiper & Atkinskon 1914, Maltsev & Zhdamirov 1995
		Ommatophoca rossii		Balleny Islands (D'Urville Sea)	Maltsev & Zhdamirov 1995
				Bellingshausen Sea	Maltsev & Zhdamirov 1995
				Graham Land	Maltsev & Zhdamirov 1995
				Ross Sea	Maltsev & Zhdamirov 1995
				South Shetland	Maltsev & Zhdamirov 1995
				Weddell Sea	Maltsev & Zhdamirov 1995
D. lobodoni		*Lobodon carcinophaga*		Balleny Islands (D'Urville Sea)	Yurakhno & Maltsev 1994

Tab. 5. Continued.

D. minutus	Monachinae	*Monachus schauinslandi*	Pacific Ocean	Midway Atoll (Hawaii)	Andersen 1987, Rausch 1969
D. mobile		*Leptonychotes weddellii*	Southern Ocean	Balleny Islands (D'Urville Sea)	Maltsev 2000
				Graham Land	Maltsev 2000, Markowski 1952b
				McMurdo Sound	Beverley-Burton 1971
				Petermann Island	Maltsev 2000
				Ross Sea	Maltsev 2000
		Ommatophoca rossii		Balleny Islands (D'Urville Sea)	Maltsev 2000
				Drygalski Island off Queen Mary Land	Johnston 1937
				Graham Land	Maltsev 2000
				Petermann Island	Maltsev 2000
				Ross Sea	Maltsev 2000
D. phocarum	Phocinae	*Pusa caspica*	-	Caspian Sea	Delyamure et al. 1964
D. pseudowilsoni	Monachinae	*Hydrurga leptonyx*		South Shetland	Wojciechowska & Zdzitowiecki 1995
D. pterocephalum	Phocinae	*Cystophora cristata*	Arctic Ocean	Disko Island	Delyamure & Skryabin 1966
D. rauschi	Monachinae	*Monachus schauinslandi*	Pacific Ocean	Midway Atoll (Hawaii)	Chapin 1927, Rausch 1969, Andersen 1987
D. quadratum		*H. leptonyx*	Indian Ocean	Adelaide	Maltsev 2000
			Southern Ocean	Amundsen Sea	Maltsev 2000
				Argentine Islands	Maltsev 2000

Tab. 5. Continued.

D. quadratum	Monachinae	*Hydrurga leptonyx*	Southern Ocean	Balleny Islands (D'Urville Sea)	Maltsev 2000
				Bellinghausen Sea	Maltsev 2000
				Coronation Island	Maltsev 2000
				Graham Land	Maltsev 2000
				Kerguelen Islands	Joyeux & Baer 1954
				Macquarie Island	Johnston 1937
				McDonald Islands	Maltsev 2000, Markowski 1952b, McEwin 1957
				Petermann Island	Maltsev 2000, Railliet & Henry 1912
				Ross Sea	Maltsev 2000
				South Georgia	Fuhrmann 1921, Linstow 1892, Maltsev 2000
				South Shetland	Maltsev 2000, Wojciechowska & Zdzitowiecki 1995
		Leptonychotes weddellii		Adelaide	Maltsev 2000
				Balleny Islands (D'Urville Sea)	Maltsev 2000
				Coronation Island	Maltsev 2000
				Graham Land	Maltsev 2000
				McDonald Islands	Maltsev 2000
				Petermann Islands	Maltsev 2000
				Ross Sea	Maltsev 2000
				South Georgia	Maltsev 2000

Tab. 5. Continued.

D. quadratum	Monachinae	*Lobodon carcinophaga*	Indian Ocean	Adelaide	Maltsev 2000
			Southern Ocean	Balleny Islands (D'Urville Sea)	Maltsev 2000
				Coronation Island	Maltsev 2000
				Graham Land	Maltsev 2000
				McDonald Islands	Maltsev 2000
				Petermann Islands	Maltsev 2000
				Ross Sea	Maltsev 2000
				South Georgia	Maltsev 2000
D. scoticum		*Hydrurga leptonyx*		Graham Land	Markowski 1952b
				Kerguelen Islands	Joyeux & Baer 1954
				Macquarie Island	Johnston 1937
				McDonald Islands	Maltsev 2000
D. schistochilos	Phocinae	*Erignathus barbatus*	Arctic Ocean	Chukchi Sea	Delyamure 1955
				Novaya Zemlya (west coast)	Vagin 1933
				Svalbard	Germanos 1896, Guiart 1935, Zschokke 1903
		Pagophilus groenlandicus		Svalbard	Guiart 1935
		Phoca vitulina		Siberia	Cholodkovsky 1914
				Svalbard	Guiart 1935
*D. sp.**	Monachinae	*Mirounga angustirostris*	Pacific Ocean	California Coast	Gerber et al. 1993
		Monachus schauinslandi		French Frigate Shoals, Laysan Island (Hawaii Islands)	Dailey et al. 1988

Tab. 5. Continued.

*D. sp.**	Phocinae	*Phoca vitulina*	Atlantic Ocean	Netherlands	Borgsteede et al. 1991
			Pacific Ocean	Gray´s Harbor, Washington	Dailey &Fallace, 1989
				California Coast	Gerber et al. 1993
		Pusa caspica		Kulaly Island (Mangyshlak Peninsula)	Kurochkin & Zablotsky 1985
D. wilsoni	Monachinae	*Hydrurga leptonyx*	Southern Ocean	Antarcitc/ King George Island	Fuhrmann 1921, Maltsev 2000, Wojciechowska & Zdzitowiecki 1995
		Leptonychotes weddellii		South Shetland	Wojciechowska & Zdzitowiecki 1995
				Petermann Island	Fuhrmann 1921, Railliet & Henry 1912
		Lobodon carcinophaga		Amundsen Sea	Maltsev 2000
				Argentine Islands	Maltsev 2000
				Balleny Islands (D'Urville Sea)	Maltsev 2000
				Bellinghausen Sea	Maltsev 2000
				Graham Land	Maltsev 2000
		Ommatophoca rossii		Antarctic	Fuhrmann 1921, Rennie & Reid 1912
Diplogonoporus tetrapterus	Phocinae	*Cystophora cristata*	Arctic Ocean	Greenland Sea	Delyamure 1966
				Iceland	Baer 1962, Krabbe 1868
		Erignathus barbatus		Iceland	Baer 1962

Tab. 5. Continued.

D. tetrapterus	Phocinae	*Pagophilus groenlandicus*	Arctic Ocean	Arctic	Delyamure 1966
				Greenland Sea	Treshchev 1982
		Phoca largha	Pacific Ocean	Bering Sea	Shults 1982
				Navarin-Anadyr	Delyamure et al. 1984
				Karaginsky Gulf (Bering Sea)	Delyamure et al. 1984, Fiscus et al. 1976
				Pribilof Islands, Bristol Bay	Delyamure et al. 1984
		Phoca vitulina		Glacier Bay, Prince William Sound (Alaska)	Herreman et al. 2011
				Sea of Japan (Kit Bay)	Belopolskaya 1960
				Sea of Okhotsk	Popov 1975
		Pusa hispida	Atlantic Ocean	Disko Island	Krabbe 1868
			Arctic Ocean	Kolokolkova Bay (Barents Sea)	Treshchev & Popov 1975
				Salluit (Canada)	Measures & Gosselin 1994
			Pacific Ocean	Alaska	Fiscus et al. 1976
	Otariinae	*Callorhinus ursinus*		Pribilof Islands	Keyes 1965, Kuzmina 2015, Margolis 1954, Stunkard 1948, Rausch 1964
				Russian Far East	Afanassjew 1941
		Eumetopias jubatus		Bering Sea	Shults 1986

Tab. 5. Continued.

D. tetrapterus	Otariinae	*Eumetopias jubatus*	Pacific Ocean	Gulf of Alaska	Shults 1986
				Karaginsky Gulf (Bering Sea)	Yurakhno 1986
				Montague Island	Rausch 1964
				Sea of Okhotsk	Delyamure 1976, Kovalenko 1975, Krotov & Delyamure 1952, Yamaguchi 1978
				St. Lawrence Island	Rausch 1964
Flexobothrium microovatum	Monachinae	*Mirounga angustirostris*	Southern Ocean	Antarctic	Maltsev 2000, Yurakhno 1989b,
Glandicephalus antarcticus		*Ommatophoca rossii*		Antarctic	Baird 1853, Railliet & Henry 1912, Rennie & Reid 1912, Shimpley 1907
				Balleny Islands (D'Urville Sea)	Yurakhno & Maltsev 1995
				Queen Mary Land	Johnston 1937
G. perfoliatus		*Leptonychotes weddellii*		Balleny Islands (D'Urville Sea)	Yurakhno & Maltsev 1995
				Commonwealth Bay	Johnston 1937
				McMurdo Sound	Beverley-Burton 1971
				Petermann Island	Fuhrmann 1921, Railliet & Henry 1912
				South Shetland Island	Wojciechowska & Zdzitowiecki 1995
Pyramicocephalus phocarum	Phocinae	*Cystophora cristata*	Arctic Ocean	Iceland	Zschokke 1903

Tab. 5. Continued.

P. phocarum	Phocinae	*Erignathus barbatus*	Arctic Ocean	Baffin´s Bay (Greenland)	Clarke 1958
				Bernard Harbour	Cooper 1921
				Iceland	Baer 1962
				Karaginsky Gulf/ Bering Sea	Delyamure et al. 1976
				Kotelny Island/ New Siberian Islands	Linstow 1905
				Novaya Zemlya (west coast)	Vagin 1933
				Svalbard	Guiart 1935, Markowski 1952a, Zschokke 1903
			Pacific Ocean	Sea of Okhotsk	Popov 1975, Delyamure & Popov 1975, Maejima et al. 1983
				Kivalina, Chukchi Sea (Alaska)	Fiscus et al. 1976, Johnson et al. 1966, Rice 1963
				St. Lawrence Island	Hilliard 1960
		Phoca largha		Sea of Okhotsk	Popov 1975
				Pribilof Islands	Delyamure et al. 1984
		Phoca vitulina		Sea of Okhotsk	Popov 1975, Popov 1982
		Pusa hispida			
				Alaska	Fiscus et al. 1976
	Otariinae	*Eumetopias jubatus*	Pacific Ocean	Kamchatka	Yurakhno 1986
				Oregon Coast	Stroud 1978

Tab. 5. Continued.

P. phocarum	Otariinae	*Eumetopias jubatus*	Pacific Ocean	Sakhalin/ Kuril Islands/ Sea of Okhotsk	Kovalenko 1975, Krotov & Delyamure 1952
				Sea of Okhotsk	Chupakhina 1971
		Callorhinus ursinus		Sea of Okhotsk	Chupakhina 1971
Ligula colymbi	Phocinae	*Pusa caspica*		Caspian Sea	Delyamure et al. 1964
Schistocephalus solidus		*Pusa hispida*		Baltic Sea	Delyamure et al. 1980

* Unspecified *Diphyllobothrium* with previously not mentioned location of infecting the given Phocid.

Tab. 6. List of diphyllobothriidean parasites invading Odobenidae with their geographical distribution .

Parasite	**Host**		**Locality**		**References**
Species	**Subfamily**	**Species**	**Ocean**	**Land/ Island/ Archipelago/Sea**	
Diphyllobothrium cordatum	Odobenidae	*Odobenus rosmarus*	Arctic Ocean	Disko Island	Ariola 1899
				Siberia	Cholodkovsky 1914
				Chukchi Sea	Protasova 2006
D. fayi			Arctic Ocean	Skull Cliff, Beaufort Sea	Rausch 2005
			Pacific Ocean	St. Lawrence Island, Bering Sea	Rausch 2005
*D. sp.***				Kodiak Island, Alaska	Hilliard 1972

** Unspecified *Diphyllobothrium* in the subfamily Odobenidae.

4.1.1. Maps of the geographical distribution of Pinnipedia and their parasites of the order Diphyllobothriidea

For more pronounced illustration of the relationships among diphyllobothriidean tapeworms, their marine hosts and their geographical distribution, the obtained data were transferred from the Table 5. and Table 6. to maps of geographical distribution of individual species of phocids (Fig. 2. - Fig.14.), otariids (Fig. 15. - Fig. 25.) and odobenids (Fig. 26.) and their diphyllobothriid cestodes with the so far described occurrence.

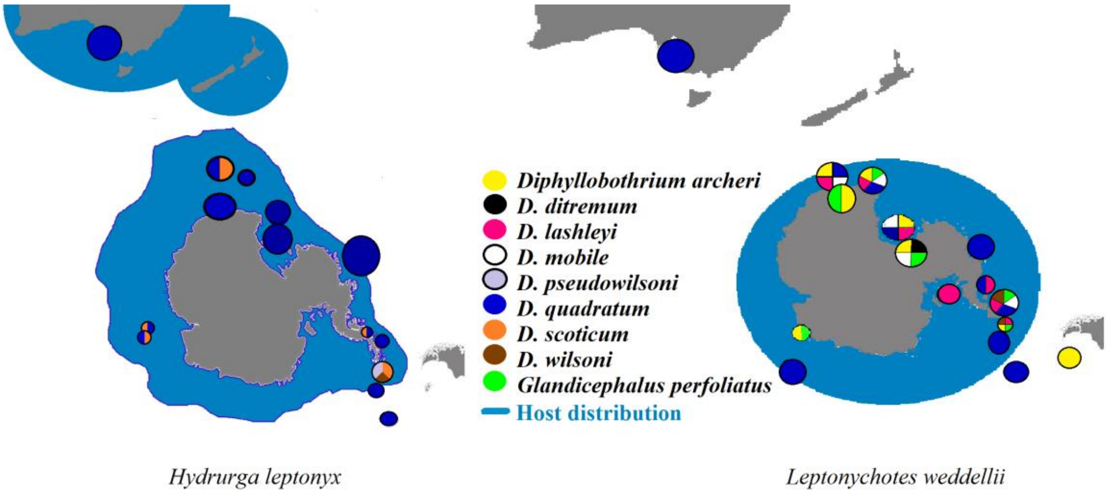


Fig. 2. Occurrence of diphyllobothriidean tapeworms in *Hydrurga leptonyx* and *Leptonychotes weddellii*.

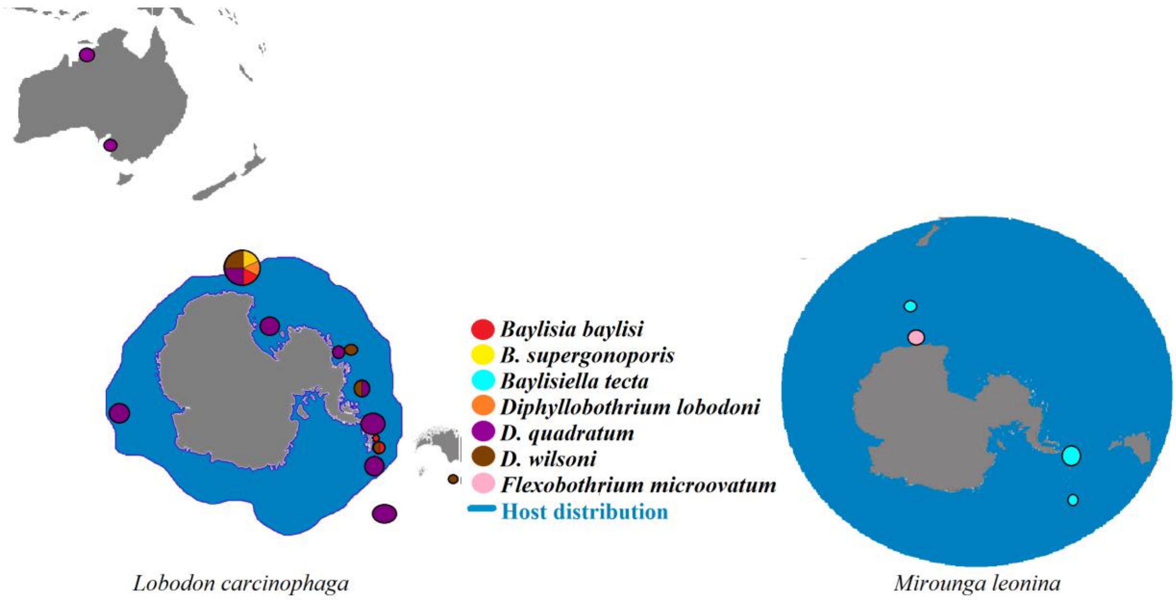


Fig. 3. Occurrence of diphyllobothriidean tapeworms in *Lobodon carcinophaga* and *Mirounga leonina*.

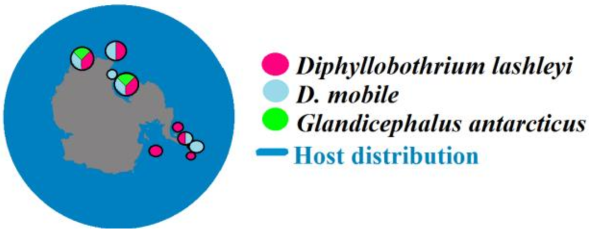


Fig. 4. Occurrence of diphyllobothriidean tapeworms in *Ommatophoca rossii*.

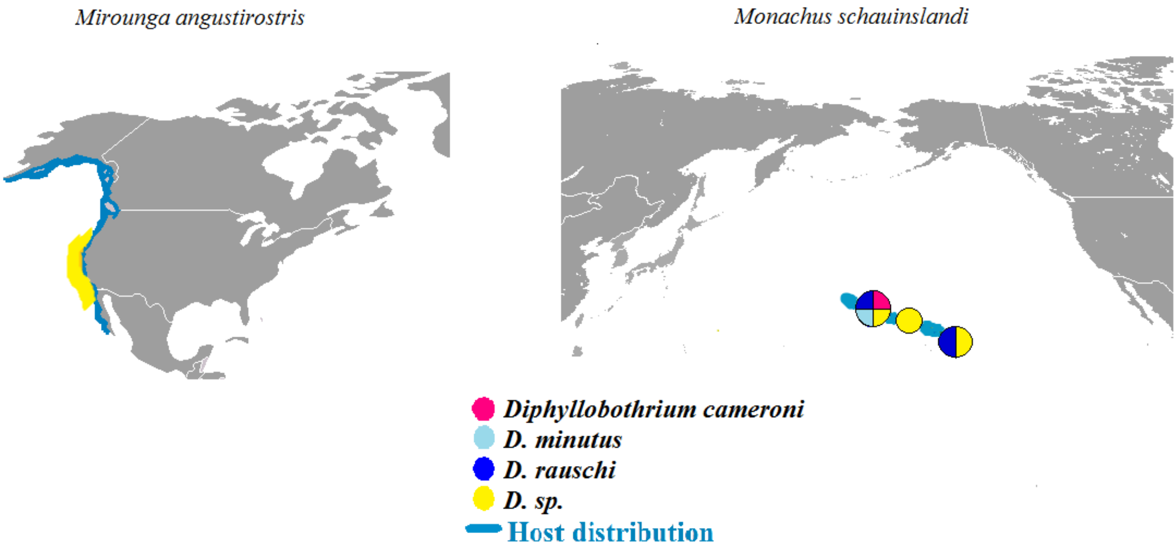


Fig. 5. Occurrence of diphyllobothriidean tapeworms in *Mirounga angustirostris* and *Monachus schauinslandi*.

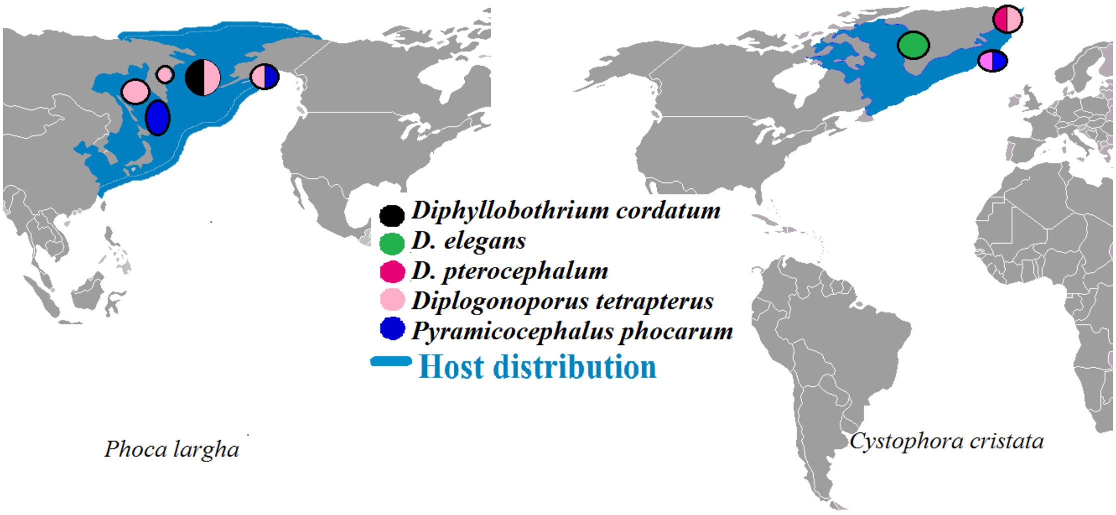


Fig. 6. Occurrence of diphyllobothriidean tapeworms in *Phoca largha* and *Cystophora cristata*.

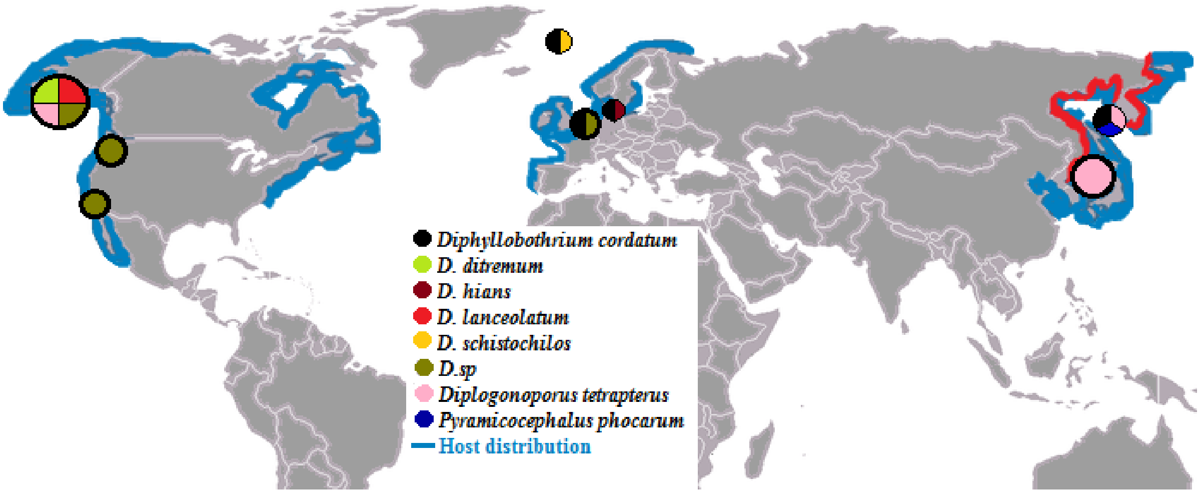


Fig. 7. Occurrence of diphyllobothriidean tapeworms in *Phoca vitulina*.

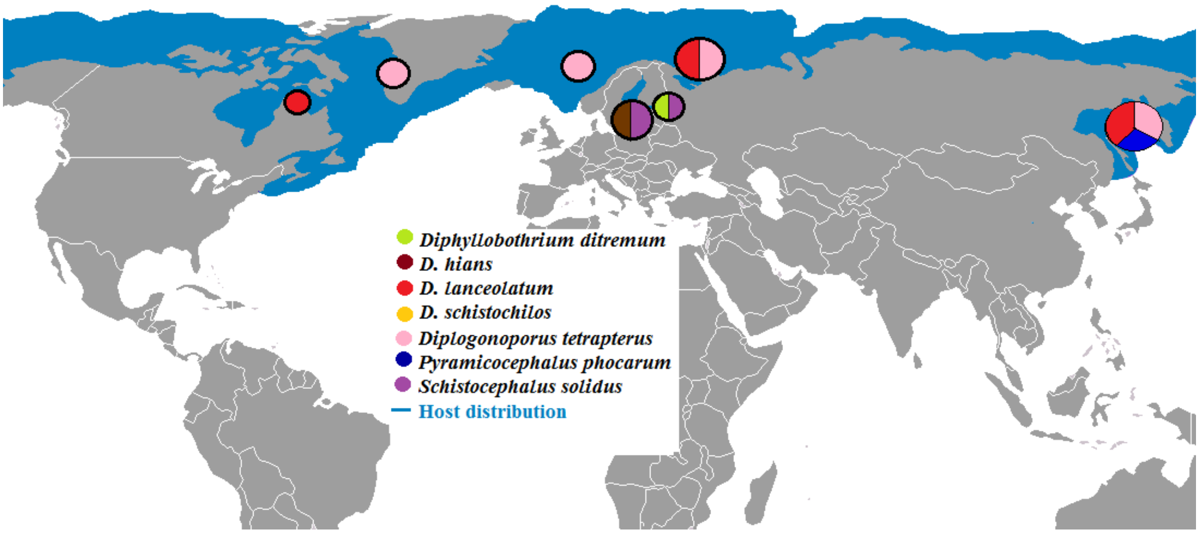


Fig. 8. Occurrence of diphyllobothriidean tapeworms in *Pusa hispida*.

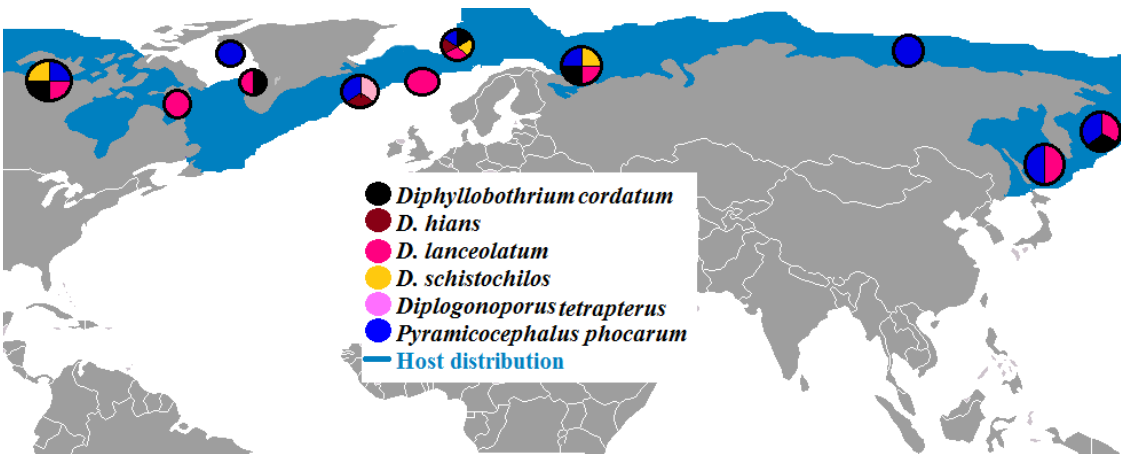


Fig. 9. Occurrence of diphyllobothriidean tapeworms in *Erignathus barbatus*.

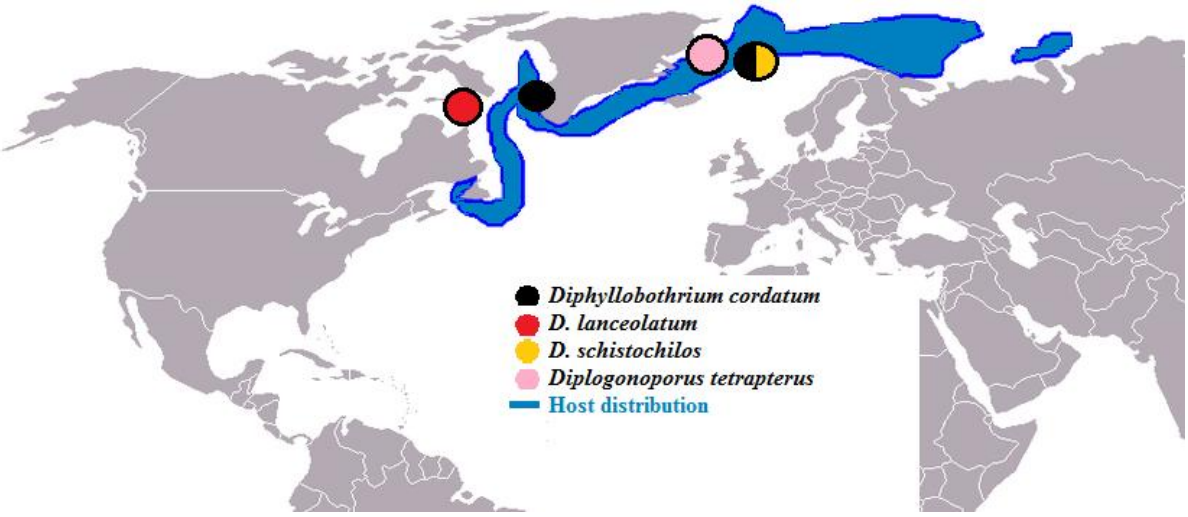


Fig. 10. Occurrence of diphyllobothriidean tapeworms in *Pagophilus groenlandicus*.

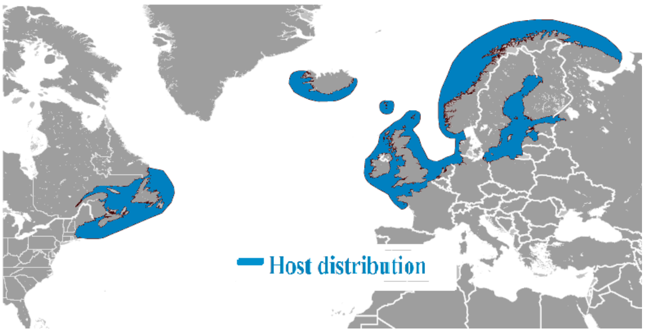


Fig. 11. Occurrence of diphyllobothriidean tapeworms in *Halichoerus grypus*.

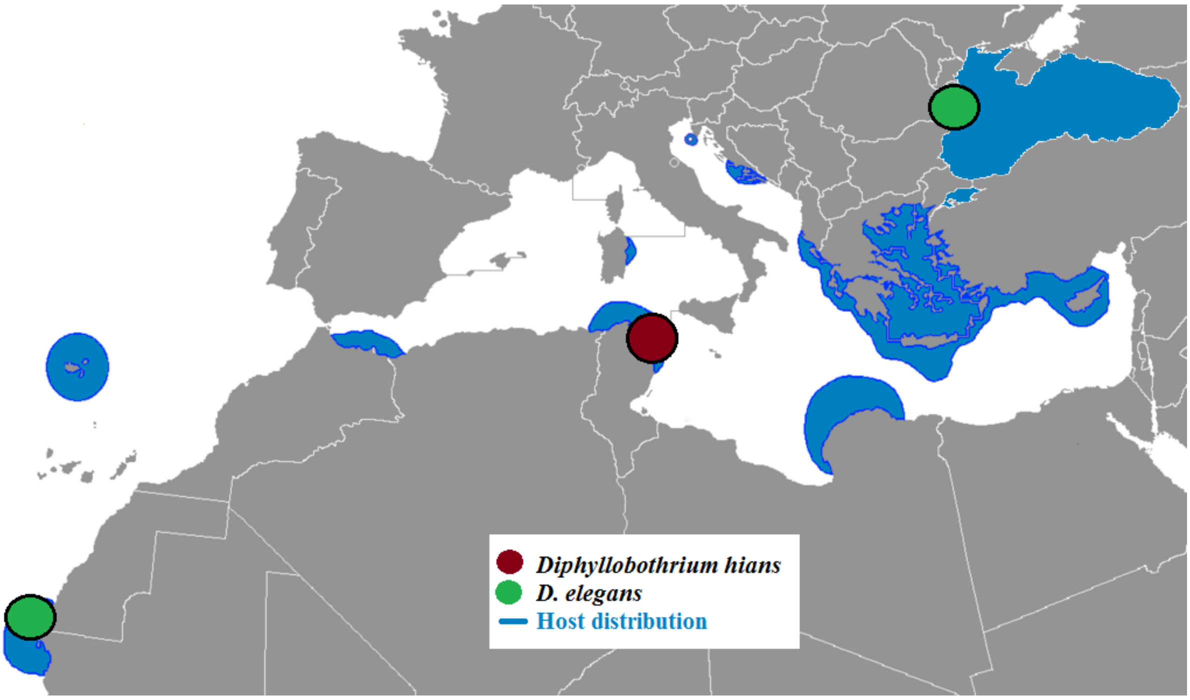


Fig. 12. Occurrence of diphyllobothriidean tapeworms in *Monachus monachus*.

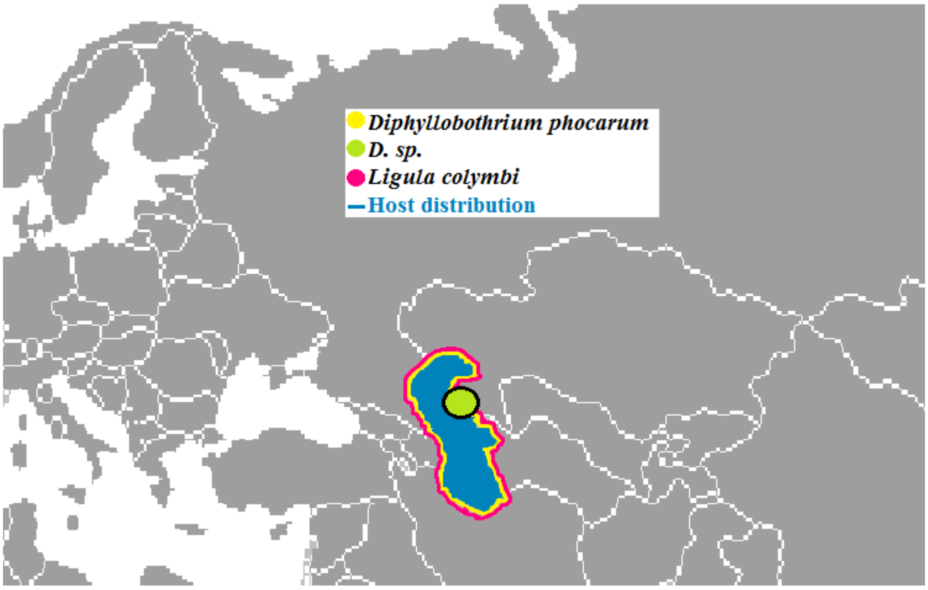


Fig. 13. Occurrence of diphyllobothriidean tapeworms in *Pusa caspica*.

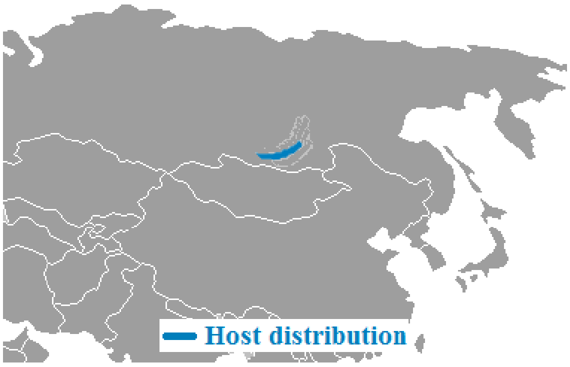


Fig. 14. Distribution of *Pusa sibirica*.

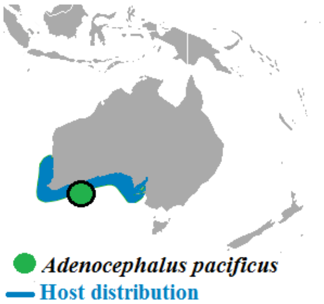


Fig. 15. Occurrence of diphyllobothriidean tapeworms in *Neophoca cinerea*.

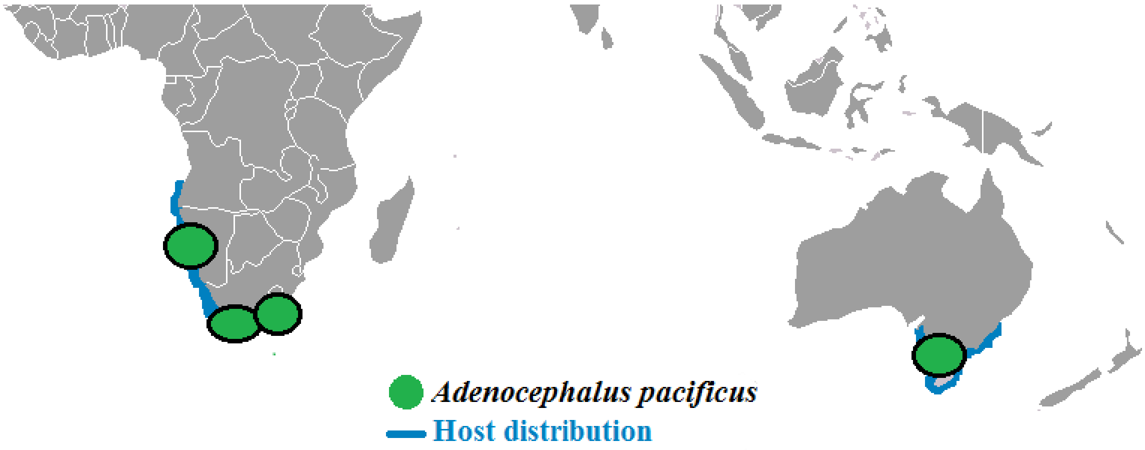


Fig. 16. Occurrence of diphyllobothriidean tapeworms in *Arctocephalus pusillus*.

Fig. 17. Distribution of *Phocarctos hookeri*. Fig. 18. Distribution of *A. forsteri*.

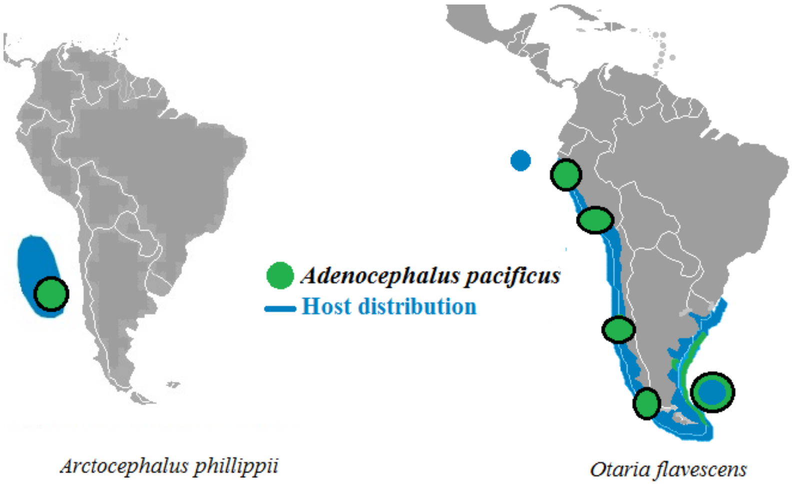


Fig. 19. Occurrence of diphyllobothriidean tapeworms in *A. phillippii* and *Otaria flavescens*.

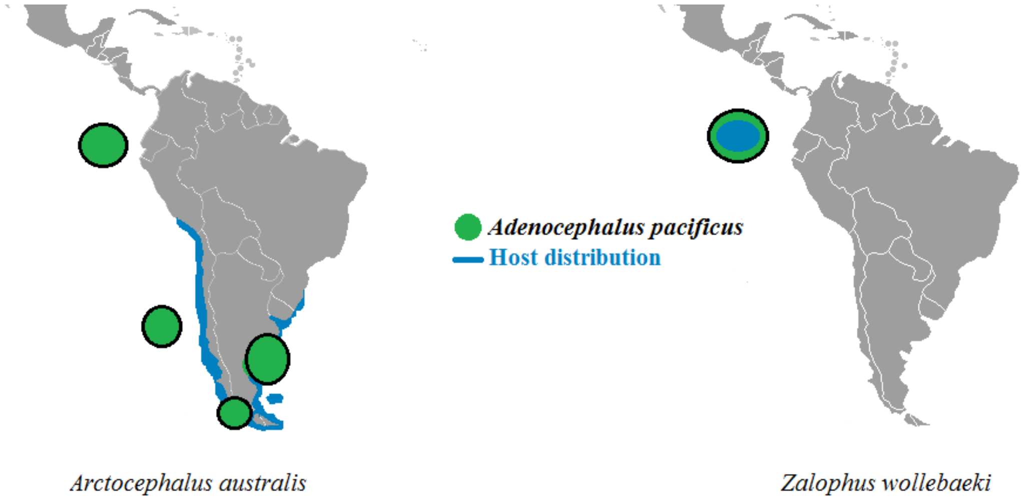


Fig. 20. Occurrence of diphyllobothriidean tapeworms in *A. australis* and *Zalophus wollebaeki.*

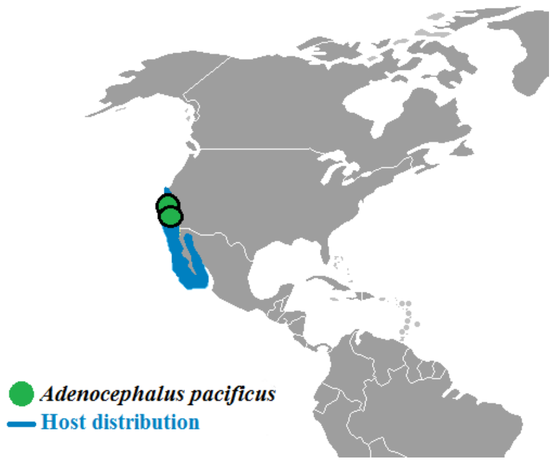


Fig. 21. Occurrence of diphyllobothriidean tapeworms in *Z. californianus*.

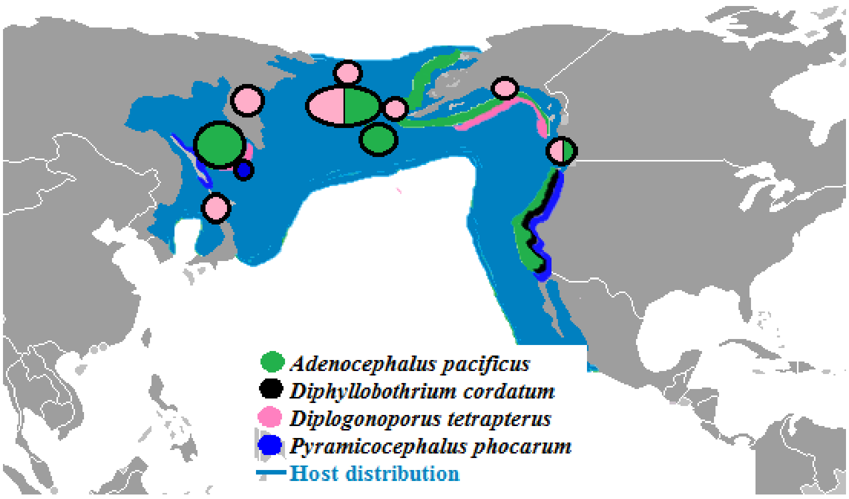


Fig. 22. Occurrence of diphyllobothriidean tapeworms in *Eumetopias jubatus*.

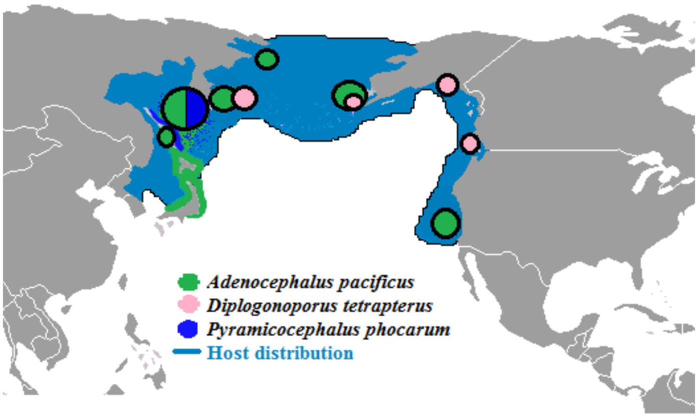


Fig. 23. Occurrence of diphyllobothriidean tapeworms in *Callorhinus ursinus*.

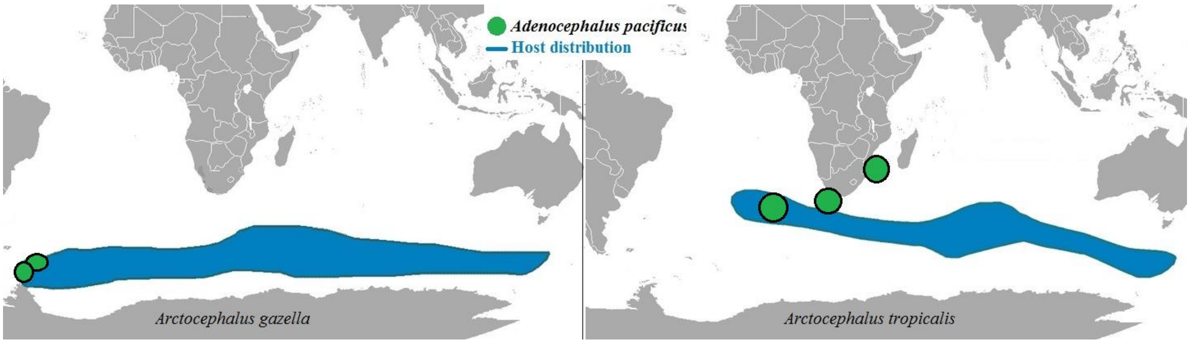


Fig. 24. Occurrence of diphyllobothriidean tapeworms in *A. gazella* and *A. tropicalis*.

Fig. 25.Distribution of *A. townsendi*.

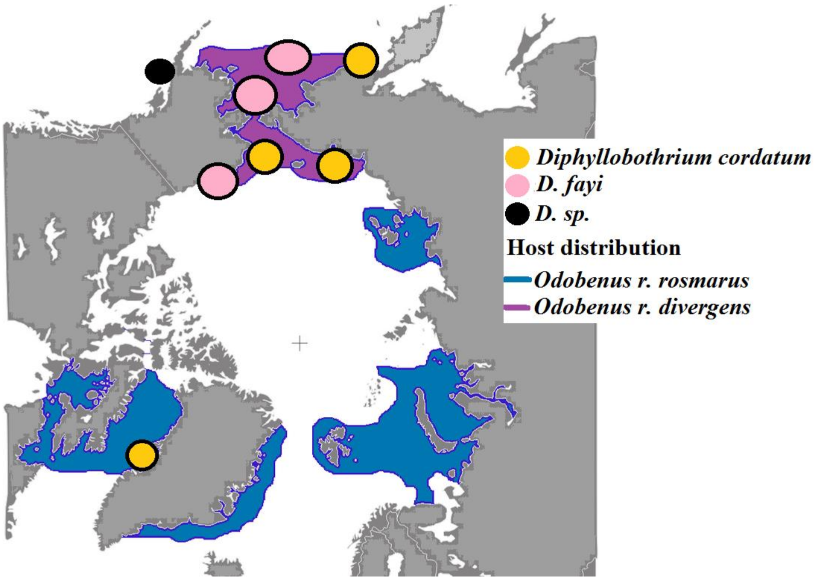


Fig. 26. Occurrence of diphyllobothriidean tapeworms in *Odobenus rosmarus*.

Five members of Phocids belonging to group Monachinae have the same range of distribution. *Hydrurga leptonyx*, *Leptonychotes weddellii* (Fig. 2.), *Lobodon carcinophaga*, *Mirounga leonina* (Fig. 3.) and *Ommatophoca rossii* (Fig. 4.) can be generally localized in the realm of the Arctic Ocean ((Rice 1988; Wilson & Reeder 2005; Yonezawa et al. 2009; Berta & Churchill 2012). Types of their diphyllobothriidean cestodes differ across the phocid species. The Weddell seal (*L. weddelli*) is a host for 7 species of the order Diphyllobothriidea: *Diphyllobothrium archeri, D. ditremum, D. lashleyi, D. mobile, D. quadratum, D. wilsoni* and *Glandicephalus perfoliatus*.

Diphyllobothrium lashleyi is a common tapeworm of the Weddell seal and the Ross seal (*O. rossii*) with the same localities of infection (Tab. 5.). The Leopard seal, Weddel seal and Crabeater seal (*L.carcinophaga*) harbour *D. quadratum*. The same group of seals, with the addition of Ross seal, are hosts for *D. wilsoni*, with the similar localities as mentioned above. The Crabeater seal is a host for *D. wilsoni* in more areas, as Amundsen Sea, Argentine Islands, Balleny Islands, Bellinghausen Sea and Graham Land (Maltsev 2000). Southern elephant seal (*M. leonina*) is the only member across the Phocidae and Otariidae infected by *Baylisiella tecta* and *Flexobothrium microovatum*. Other cestodes, with hosts of the phocids living close to Antarctic, include *Baylisia baylisi, B. supergonoporis, D. cameroni, D. elegans, D. hians, D. lobodoni, D. minutus, D. scoticum, D. rauschi, D. pseudowilsoni, Glandicephalus antarcticus* and *Schistochilos perfoliatus*.

In *Mirounga angustirostris* (Fig. 5.) and *Phoca vitulina* (Fig. 7.) were found unidentified species of the genus Diphyllobothrium along the California coast (Gerber et al.

1993). They belong to the group of seals living in the northern hemisphere. Seals inhabiting North Pacific Ocean, North Atlantic Ocean and Arctic Ocean are hosts for the following diphyllobothriidean parasites: *D. cordatum, D. ditremum, D. elegans, D. hians, D. lanceolatum, D. pterocephalum, D. schistochilos, Diplogonoporus tetrapterus, Ligula colymbi, Pyramicocephalus phocarum and Schistocephalus solidus,* while the parasites of *Diphyllobothrium cameroni, D. minutus* and *D. rauschi* belong to the endemic species of Hawaiian monk seal (Fig. 5.), inhabiting only Hawaii Islands (Rice 1998).

The Baikal seal (*Pusa sibirica*) is endemic to the Baikal Sea with no diphyllobothriidean cestodes (Fig. 14.).

Members of Otariidae predominantly inhabit southern hemisphere, including Australia and South America with adjacent islands, and South Africa. Almost all species of sea lions are hosts of *Adenocephalus pacificus*. Sea lions living in the northern hemisphere (except *Arctocephalus townsendi* and *Zalophus californianus*) are infected by more species of diphyllobothriidean tapeworms. Records from the Steller sea lion (*Eumetopias jubatus*) (Fig. 22.) and the northern fur seal (*Callorhinus ursinus*) (Fig. 23.) are showing occurrence of *A. pacificus*, *Diplogonoporus tetrapterus* and *P. phocarum* in the North Pacific Ocean. In the Steller sea lion was also detected *D. cordatum*. The New Zealand sea lion (*Phocarctos hookeri*) (Fig. 17.), New Zealand fur seal (*Arctocephalus forsteri*) (Fig. 18.) and Guadalupe fur seal (*A.townsendi*) (Fig. 25.) were found negative for cestodes of the order Diphyllobothriidea.

Both subspecies of walrus are distributed in northern hemisphere (Fig. 26.). The Pacific walrus is a host for at least two species of the order Diphyllobothriidea (*D. cordactum*, *D. fayi*), while the only one known species of the Atlantic walrus is *D. cordatum* from the Kodiak Island near Alaska. Diphyllobothriidean parasites of the Pacific walrus are distributed both in Pacific Ocean and Arctic Ocean.

4.2. Coprological examination of *Phoca vitulina*

The examined material was negative for the diphyllobothriidean tapeworms (Cestoda), but positive for several trematode and nematode species. Eggs of parasites were measured and photographed by OLYMPUS cellSens Standard 1.13 imaging software and Quick PHOTO MICRO 2.3 imaging software. Eggs were divided into three different groups based on their size. From the total number of 107 eggs, 43 eggs measured on average 23 × 47 µm (width 19—27, length 42—51), 44 eggs measured on average 18 × 35 µm (width 15—22, length 30—39) and 20 eggs measured on average 11 × 21 µm (width 10— 13, length 18—25).

Faecal samples contained definitely at least three species of nematodes. From 20 seals, 14 patients harboured larvae (Fig. 27.) of the *Anisakis* Dujardin, 1845 complex, which sizes varied from 60 to 299 µm. Samples of another three seals contained eggs of *Anisakis* with average size 47 × 45 µm (Fig. 30.). Eggs of the average size 11 × 21 µm (Fig. 28.) probably belonged to lungworms of the species *Parafilaroides gymnurus* (Railliet, 1899). Eggs of the genus *Capillaria* Zeder, 1800 (Fig. 31.) occurred only in two seal patients (1 adult, 1 juvenile). The size of eggs reached approximately 63 × 30 µm and the species is recognized as *Capillaria delamurei*, Zablotzkii, 1971.

Trematode eggs (Fig. 29.) belonged to the class of Heterophyidae Odhner, 1914 and very probably to the species *Ascocotyle septentrionalis* (van den Broek, 1967).

Fig. 27. Microphotograph of the *Anisakis* complex larva.

Fig. 28. Egg of Nematoda, *Parafilaroides* cf. *gymnurus*.

Fig. 29. Egg of Trematoda, *Ascocotyle septentrionalis*.

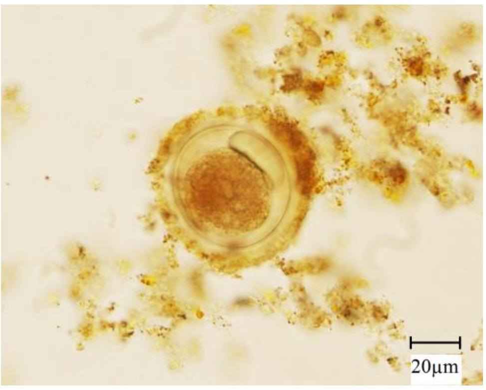


Fig. 30. Egg of Anisakidae (Nematoda). Fig. 31. Egg of *Capillaria* (Nematoda).

Comparing of sedimentation and flotation methods based on the endoparasites mentioned above, have shown following results: larvae of Anisakiidae were more often recognized using the sedimentation technique with 77% success rate (flotation - 48%), while the flotation method was more efficient (with 74% success rate) in occurence of nematode and trematode eggs (sedimentation - 59%).

Due to combination of these two coprological methods, it was possible to conclude that 95% of examined seal patients had trematode eggs and 70 % of mentioned seals were infected by larvae of Anisakiidae. The sedimentation method also revealed the presence of fungi *Alternaria* Nees ex Wallroth, 1816 and digested remains of crustaceans.

The periods between sampling (i.e. from arrival of the patient to 24-48 hours (or more) after giving a medication) was established to determine the effectiveness of medicaments attacking endoparasites of harbour seal. The results showed the reduction of parasites even after 24 hours of taking medicine.

5. DISCUSSION

5.1. Literature review

The first occurrence of diphyllobothriidean tapeworm infecting pinnipeds is dated to 1848, when Siebold described *Diplogonoporus tetrapterus* for the first time in *Phoca vitulina* (Phocidae) (Siebold 1848). At this time, it has not yet been possible to identify species by using molecular-biology techniques, which can provide further information. Until then, the authors of the publications could have been mistaken in determinations of species. In some cases, species of tapeworms or marine hosts were not mentioned at all. Based on the publications, any references on parasites of *Pusa sibirica* (Phocidae) and three members of Otariids, called *Arctocephalus forsteri*, *Arctocephalus townsendi* and *Phocarctos hookeri*, as hosts of Diphyllobothriids, were missing. In *P. sibirica* were previously present species of Anoplura and Nematoda (Felix 2013). The Russian publications, focused on parasites of marine mammals, were precise from the 19th century. It is very unlikely to consider, that the authors overlooked the order Diphyllobothriidea in elaborated seals. The parasitofauna of above-mentioned otariids is probably less known due to small number of studies.

Linstow (1901) mentioned the existence of *Pyramicocephalus phocarum* infecting the genus *Phoca*, however, with any identification of the species. It is difficult to determine which species of the genus *Phoca* served as the host for the above mentioned tapeworm, because both species (*P. vitulina* (Fig. 7.) and *P. largha* (Fig. 6.) are distributed worldwide in the northern hemisphere, and both of them were described to be associated with *Pyramicocephalus phocarum* (Popov 1975; Popov 1982; Delyamure 1984). All available data related to the given hosts and parasite are located in the Pacific Ocean, while the information from Linstow (1901) describes the locality of Iceland. Due to this fact, the locality was kept in the database for possible verifying in the future.

In the southern hemisphere, in area of the Antarctic Ocean, most of individuals of same phocid and otariid species originated from same localities (Fig. 2 – 4, Fig. 24.). It was probably due to existence of research stations, as Arctowski Station localized on the King George Island of the South Shetland Archipelago, where was easier to elaborate fresh-collected material compared to unknown, uninhabited areas of Antarctic (Wojciechowska & Zdzitowiecki 1995).

The diet of *Odobenus rosmarus* generally consists of benthic invertebrates, while they exceptionally harbour cestode plerocercoids after eating fish (Yurakhno 1971). The species *Diphyllobothrium fayi* is strictly host-specific to *Odobenus rosmarus divergens*. It is hard to identify specific host-subspecies of *D. latum* and *D. roemeri* present in *O. rosmarus*, because they were mentioned in studies with unknown locality. Other parasite, *D. cordatum* is (beside the genus *Odobenus*) also known from another 3 genera of phocids and one genus of otariids. The generalist, *D. cordatum*, invades species *Erignathus barbatus*, *Pagophilus groenlandicus*, *Phoca largha*, *Phoca vitulina*, *Eumetopias jubatus* and the above-mentioned *Odobenus rosmarus*. The occurrence of *D. cordatum* is common in phocids, but rare in odobenids. *Adenocephalus pacificus* is family-specific to Otariidae. Another three generalists invade digestive tract, at least, of 4 phocids and 1 otariid. *Diphyllobothrium lanceolatum* infects *E. barbatus*, *P. groenlandicus*, *P. vitulina* and *Pusa hispida* from the family Phocidae. The only one otariid host for *D. lanceolatum* is *E. jubatus*. The generalist *Diplogonoporus tetrapterus* invades members of pinnipeds as for *D. lanceolatum*, including phocids *Cystophora cristata*, *Phoca largha* and one extra otariid member Callorhinus ursinus. *Pyramicocephalus phocarum* parasitizes the same range of pinniped hosts as *D. tetrapterus*, except for *Pagophilus groenlandicus*. Other 29 diphyllobthriids are host-specific to the family Phocidae, while 17 tapeworm species of the order Diphyllobothriidea are identified as strict specialists. *Diphyllovothrium pseudowilsoni* and *D. scoticum* are specialists for *Hydrurga leptonyx*. *Diphyllobothrium archeri* and *Glandicephalus perfoliatus* are specialist for *Leptonychotes weddellii*. *Baylisia baylisi*, *B. supergonoporis* and *D. lobodoni* are specialists for *Lobodon carcinophaga*. *Mirounga angustirostris* is distributed along California coast and harbours cestode of the genus *Diphyllobotohrium*, but the species of the parasite is unknown (Gerber et al. 1993). *Diphyllobothrium cordatum*, *P. phocarum and Diplogonoporus tetrapterus* share the same range of distribution (Fig. 6 – 9) of their phocids hosts. Another common tapeworm of this locality (Fig. 21 – 23) is *Adenocephalus pacificus* (cosmopolite), which is a strict specialist to otariids. Other phocids harbour from 2 to 8 strict specialists. Another 12 species of diphyllobothriidean tapeworm in phocids are generalists. According to Rausch (2005), the host-specifity of cestodes in marine mammals is low, while the results of this study shows that the host-specifity of the order Diphyllobothriidea in pinnipeds is relatively high (51 % of diphyllobothriidean tapeworms are specialists).

Diphyllobothriidean specialists of *Lobodon carcinophaga* are limited in distribution by range of localities of their host (Fig. 3.). *B. baylisi*, *B. supergonoporis*, *Baylisiella tecta* and *D. lobodoni* are distributed near the Balleny Islands and few other localities, while the generalist *D. quadratum* is distributed along the entire Antarcitc (Wojciechowska & Zdzitowiecki 1995; Yurakhno & Maltsev 1997). *Diphyllobothrium scoticum* has in

Hydrurga leptonyx and *G. perfoliatus* in *L. weddellii* the similar range of distribution (Fig. 2.) as *D. quadratum*. *Flexobothrium microovatum* is limited only to one place (St. Lawrence Island, Antarctic) of the *M. leonina* distribution (Fig. 3.) (Rausch 1964). The location (Fig. 5.) of *Monachus schauinslandi* limits area of distribution of its specialists: *D. cameroni*, *D. minutus*, *D. rauschi* near the Midway Atoll in the Pacific Ocean (Rausch 1969). The generalists, *D. cordatum* and *Diplogonoporus tetrapterus*, are distributed exclusively in the northern hemisphere. Specialists of species *D. phocarum* and *Ligula colymbi*, invading endemic species *Pusa caspica* (Fig. 13.), are limited to Caspian Sea (Delyamure et al. 1964). The opposite case is already above-mentioned *Adenocephalus pacificus*, which is widely distributed and its occurence may be limited by distribution of its intermediate hosts.

In order to maintain timeliness of the information, it is useful to repeat the study. In case of *Pusa caspica*, the examination of parasites of this host was done only few times, one mentioning *D. phocarum* and *Ligula colymbi*, and two mentioning unidentified species of the genus *Diphyllobothrium* (Delyamure et al. 1964; Kurochkin 1958).

According to The IUCN Red List of Threatened Species, the Caspian seal is currently classified as endangered species, so the studies of his endoparasites may be difficult (www.iucnredlist.org[4]). In this and many other cases, the coprological examination of faeces is the method of choice. Because the collection of material can be challenging, a great advantage is the existence of rescue, rehabilitation and research centers, such as SRRC in Netherlands or Pacific Marine Mammal Center in California, USA. In these centers it is easier to collect faecal samples than in nature because members (nurses) of the organisation have to be in direct contact with seals and sea lions, if necessary.

5.2. Material from coprology of *Phoca vitulina*

The samples were negative for the cestode parasites. On the other hand, several species of nematodes and trematodes were detected in the samples.

The sedimentation technique is more efficient to prove heavy eggs of trematodes or acathocephalans in the faecal material, while the flotation technique can show presence of lighter elements of sample as larvae, oocysts and eggs of Protozoa, Cestoda and Nematoda.

The reason of no occurrence of protozoa or any diphyllobothridean stages in the faecal samples can be the age of seal patients. The majority of seal patients of my study were juveniles (around 3 months old). Cestodes of the order Diphyllobothriidea could be present in bodies of seal patients, but their demonstrable stages in faeces did not exist yet, due to low age of the seal. It is also possible, that they were not very efficient in feeding at this period. The probability of infection were lower due to lack of food.

From the previous studies, *Phoca vitulina* harbours a wide range of tapeworms of the order Diphyllobothriidea in compare to other pinnipeds. According to elaborated data, *P. vitulina* is a host for *D. cordatum*, *D. ditremum*, *D. hians*, *D. lanceolatum*, *D. schistochilos*, *Diplogonoporus tetrapterus*, *Pyramicocephalus phocarum*, while *D. cordatum* and *D. hians* are probably common for the locality near the Netherlands in seals. Borgsteede (1991) studied 94 seals, which died during the epidemic of the phocine distemper virus. His study revealed, that only 8.5 % of examined seals, had parasites of the order Diphyllobothriidea. The prevalence of these tapeworms increased in direct proportion with the age of examined seals.

The trematode *Ascocotyle septentrionalis,* present in examined samples, is in most publications known as *Phagicola* cf. *septentrionalis* van den Broek, 1967 (Gibson 2001).

Since the identification of parasites was based only on light microscopy and measurements of eggs and larvae, the endoparasites could not be determined to species with certainty. Combination of coprological methods with molecular analyses are noninvasive and perspective for the future study of seals and sea lions, which is the aim of my master thesis.

6. CONCLUSIONS

1) The elaborated data were summarized to gain a view of the host specifity and geographical distribution of the order Diphyllobothriidea invading Pinnipeds.

2) From 33 species of diphyllobothriids, 29 are family-specific to Phocidae, while *Diphyllobothrium cordatum* is host-specific to Pinnipedia. *Adenocephalus* pacificum is family-specific to Otariidae. The species *D. fayi* is strictly host-specific to *Odobenus rosmarus divergens*.

3) Diphyllobothriidean cestodes appear to be low host-specific, with the exception of few species, which are probably strict on related to intermediate hosts.

4) The faecal material from predominantly young seal patients (juveniles) of *Phoca vitulina*, was positive for following endoparasites: *Anisakis* complex, *Parafilaroides* cf. *gymnurus*, *Capillaria delamurei*, belonging to Nematoda and *Ascocotyle septentrionalis* (Trematoda). Tapeworms of the order Diphyllobothriidea were not found.

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