GLOBAL WATER PATHOGEN PROJECT

**PART THREE. SPECIFIC EXCRETED PATHOGENS: ENVIRONMENTAL AND
EPIDEMIOLOGY ASPECTS**

DIPHYLLOBOTHRIIDAE

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Summary

The Diphyllbothriidae family includes zoonotic species of importance in human and animal health that are grouped into genera *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, and *Spirometra*. The species of the first three genera have larval stages (coracidia) that develop in the aquatic environment and have intermediate host copepods (proceroids) and fish (plerocercoids or sparganums) in their life cycles. In addition, all include a definitive host, where they develop into the adult stage (mammals or birds, the latter in only some species of *Dibothriocephalus*). In *Spirometra* spp., the plerocercoid develops in amphibians, snakes, birds and mammals, including humans, and their definitive host corresponds to canids and felids.

Humans become infected by species of so-called broad fish tapeworms (*Dibothriocephalus*, *Diphyllbothrium* and *Adenocephalus*) by eating raw or undercooked fish (freshwater or marine). These parasites can cause severe gastrointestinal symptoms. *Spirometra* spp. can cause severe disease, depending on the final location of larvae in humans. This infection is acquired by drinking water contaminated with infected copepods, ingesting undercooked meat of amphibians, snakes, birds or mammals or applying flesh of these animals on open wounds.

The aquatic environment becomes contaminated with the feces of the definitive host, containing the parasite eggs: *D. latus* in mainly humans and dogs, also other domestic and wild mammals; *D. dendriticus* in fish-eating birds, mainly gulls (Laridae); *A. pacificus* in marine mammals such as sea lions and eared seals and *Diphyllbothrium* in whales and fur seals; and canids and felids in the life cycle of *Spirometra*. The spread of *D. dendriticus*, *A. pacificus*, and *Diphyllbothrium* would be by wild hosts will be greater than the spread by humans.

The spread of parasite eggs derived from human infections by *Dibothriocephalus*, *Diphyllbothrium*, and *Adenocephalus* can partly be controlled by latrine use and treating wastewater. The dissemination of parasites in the family Diphyllbothriidae present in wild or domestic hosts is more difficult to avoid. While wastewater treatment is part of the transmission control options, efficient health education to create awareness of adequate thermal and freezing processes in the preparation of fish dishes will also have an impact. Strict regulations of food prepared with raw fish in restaurants, timely treatment of patients and availability of clean drinking water, in the case of *Spirometra*, could contribute to an improved control of these re-emerging infections.

1.0 Epidemiology of the Disease and Pathogens

1.1 Global Burden of Disease

1.1.1 Global distribution

1.1.1.1 *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, and *Spirometra*

The Diphyllbothriidae family includes the so-called broad tapeworms that are widely distributed all around the world, with some being agents of human infection. They are commonly present in the wildlife among fish, mammal and bird hosts that make an important reservoir (Muller, 2002; Chai et al., 2005). Recently, taxonomy of broad tapeworms included in the family Diphyllbothriidae have been revised, including the followings genera with species of greater importance in health: *Dibothriocephalus* with *D. latus* (syn. *Diphyllbothrium latum*), *D. dendriticus* (syn. *Diphyllbothrium dendriticum*), and *D. nihonkaiensis* (syn. *Diphyllbothrium nihonkaiense*); *Diphyllbothrium* with *Di. stemmacephalum* and *Di. balaenopterae* (syn. *Diplogonoporus balaenopterae*); *Spirometra* and *Adenocephalus* (Waeschenbach et al., 2017).

In the early 1970's the worldwide prevalence of human infection was estimated at 9 million cases (WHO, 1979). Recent data indicate that 20 million people are infected worldwide with *D. latus* (Muller, 2002; Chai et al., 2005; Scholz and Kuchta, 2016). Currently, there are estimated about 270; 1,000; 1,000; and 2,000 human cases of infection by *Di. balaenopterae*, *D. dendriticus*, *Adenocephalus pacificus*, and *D. nihonkaiensis*, respectively (Scholz and Kuchta, 2016). The zoonosis occurs most commonly in countries where the consumption of raw fish is a frequent practice. Flesh (marinated fillets, carpaccio, ceviche, sushi, etc.) of infected fish with plerocercoid larvae could transmit the infection to humans.

The areas of endemicity are Japan, the Baltic countries, Scandinavia, Western and Eastern Russia, North America (Pacific Northwest) and South America. In Japan, especially along the coast of the Sea of Japan, about 100 cases per year have been reported since the 1970s (Oshima and Wakai, 1983). In the coastal areas of the Okhotsk Sea, human prevalence ranges from 1.0 to 3.3% (Lloyd, 1998).

Human infection by *Dibothriocephalus* spp. is decreasing in some historical areas of endemicity as the Baltic or Scandinavian countries (Raisanen and Puska, 1984) and in North America (Dick, 2008; Jenkins et al., 2013). In contrary, due to the increasing popularity of dishes utilizing raw or undercooked fish, human infection is emerging or re-emerging in some European countries, particularly in Italian and French speaking areas of the perialpine lakes (Dupouy-Camet and Peduzzi, 2004; Scholz et al., 2009), and in South America (Torres et al., 1989b, 1993, 2004a; Semenas et al., 2001; Sampaio et al., 2005; Knoff et al., 2011; Kuchta et al., 2015a). In Peru, prevalence of infection by *A. pacificus* is around 2%, but has been as high as 7.5% in some regions (Cabrera, 2003).

Due to the globalization of fish trading and to travels and immigration, sporadic cases occurred in no endemic countries and exotic cases (cases with exotic species) occurred in endemic countries (Yera et al., 2006; Wicht et al., 2007, 2008a, 2008b; Paugam et al., 2009; Knoff et al.,

2011; Semenas, 2014; An et al., 2017). In 2010, 214 million people lived outside their country of origin, these movements of people contributing to the displacement of foodborne parasites to different regions of the world, as evidenced by example in *Dibothriocephalus* spp. in groups of refugees (Robertson et al., 2014). Scholz and Kuchta (2016), gives precise data on the geographical distribution of dibothriocephalosis.

1.1.1.2 Europe

Dibothriocephalus latus has been considered to be the principal species infecting humans in Europe. It is autochthonous in northeastern Europe and subalpine lakes. Microscopic eggs of this species (morphologically diagnosed) were found in coprolites from 4,000 BC in France and Germany (Le Bailly et al., 2005). Human become infected with *D. latus* by eating raw freshwater fish as perch (*Perca fluviatilis*), pike (*Esox lucius*), Arctic char (*Salvelinus alpinus*), burbot (*Lota lota*), and other fish containing plerocercoid larvae of parasite. Recently, molecular identification of samples allowed the diagnosis of sporadic exotic clinical cases locally acquired from imported raw fish or overseas acquired in travelers. In this way *D. nihonkaiensis* cases were reported in France and Switzerland (Yera et al., 2006; Wicht et al., 2008a, 2008b; Paugam et al., 2009), *D. dendriticus* in Switzerland and Czech Republic (De Marval et al., 2013; Kuchta et al., 2013), *A. pacificus* (Kuchta et al., 2014; Pastor-Valle et al., 2014), *Di. balaenopterae* (Clavel et al., 1997; Pastor-Valle et al., 2014) and *D. latus* cases in Spain (Esteban et al., 2014).

1.1.1.3 North America

Dibothriocephalus latus seems to be native in North America and occurred in indigenous people and dogs (Dick, 2008). Most cases have been reported in the Great Lakes region, central Canada (Manitoba), and Alaska. Human become infected with *D. latus* by eating raw freshwater as pikeperch or walleye (*Sander canadensis* and *S. vitreus*). *Dibothriocephalus dendriticus* is found commonly throughout arctic and subarctic regions parasitizing piscivorous birds and mammals. Humans have infrequently been infected with this species in north Canada and Alaska (Kuchta et al., 2013). Humans become infected with *D. dendriticus* by eating raw salmonid including whitefish (*Coregonus* spp.) (Scholz and Kuchta, 2016). Others species *D. dalliae*, *D. lanceolatum*, *D. ursi*, *D. alascensis* (syn. *Diphyllbothrium alascense*) (Rausch and Adams, 2000), and more recently *D. nihonkaiensis* have been responsible for human infections in the Pacific coast of North America (British Columbia, Canada and North-West, USA) (Wicht et al., 2008b; Fang et al., 2015). Recently, Kuchta et al. (2017) reported plerocercoids of *D. nihonkaiensis* in the pink salmon, *Oncorhynchus gorbuscha*, from Resurrection Creek, Alaska.

1.1.1.4 South America

Dibothriocephalus latus was introduced in 20th century in Chile and Argentina where it is endemic in the southern freshwater ecosystems (Neghme et al., 1950; Torres et al.,

2004a, 2012; Semenas and Úbeda, 1997; Semenas et al., 2001; Semenas, 2014; Mercado et al., 2010). In Chile, first human autochthonous case was reported in 1950 by Neghme et al. Around 90 cases of dibothriocephalosis attributed to *D. latus* have been published of patients who have consumed trouts, *Oncorhynchus mykiss* or *Salmo trutta*, both hosts of *D. latus* and *D. dendriticus* in lakes from southern Chile (Torres et al., 1993, 1998, 2004a; Torres et al., 2001; Mercado et al., 2010; Rosas and Weitzel, 2014). Some of the autochthonous cases have been identified by finding the microscopic egg parasites in feces of patient, other by studying adult stage or their proglottids, that can distinguished morphologically between the 3 species of diphyllbothriids identified in Chile, included *D. latus*, *D. dendriticus* and *A. pacificus*, this last in species of marine fish (Torres et al., 1981b; Andersen et al., 1987; Kuchta et al., 2013; Hernández-Ortiz et al., 2015), or by genetic testing (Mercado et al., 2010). Prevalence of infection by *D. latus* fluctuated between 0.2% and 3.4% in humans living near of some lakes in southern Chile (Torres et al., 1989b, 1998, 2004a). Same authors indicated that domestic dogs are an important reservoir of dibothriocephalosis in the region.

In Argentina, to date there have been about 50 cases of human dibothriocephalosis reported, where the first case was identified in a European patient in 1906, but the first autochthonous case was recorded in 1982 in a fisherman frequent in the Andean-Patagonian lakes that had consumed introduced salmonids (Semenas, 2014).

More recently, based in the review and references cited by Knoff et al. (2011), about 70 cases attributed to *D. latus* or other unidentified species have been published in Brazil, including autochthonous and non-autochthonous cases (Eduardo et al., 2005a, 2005b; Tavares et al., 2005; Santos and Faro, 2005; Emmel et al., 2006; Capuano et al., 2007; Mezzari and Wiebbeling, 2008; Llaguno et al., 2008; Knoff et al., 2011). The cases in Brazil have been mainly related to the consumption of raw, smoked or insufficiently cooked imported salmonids. However, the presence of *Diphyllbothrium* plerocercoids in marketed marine fish, such as the pink cusk-eel, *Geniptyrus brasiliensis*, has been reported (Knoff et al., 2008). Recently, *A. pacificus* has been identified in the South American fur seal, *Arctophoca australis* (syn. *Arctocephalus australis*), a definitive host in the coast of Brazil (Jacobus et al., 2016).

Adenocephalus pacificus is the native South American diphyllbothriid. Eggs of this species (morphologically diagnosed) were found in coprolites from 2,700 to 2,850 BC in a coastal site in North-Central Peru (Reinhard and Urban, 2003) and at the site of Tiliviche in Northern Chile from 4,110-1,950 BC (Ferreira et al., 1984). Human infections have been reported with about 1,000 (Flores et al., 2002; Kuchta et al., 2015b), 19 (Sagua et al., 2001; Mercado et al., 2010), and 13 (Gallegos and Brousselle, 1991) human cases from Peru, Chile and Ecuador, respectively. Also, Kuchta et al. (2015b) mentioned a case of a student from Peru who was in Argentina and was infected with a tapeworm identified as *Lueheella* sp. (syn. *Spirometra*) and after revision of specimen revealed characteristics of *A. pacificus*. The origin of the infection in

that patient it is not clear. The parasite displays a relatively low host specificity (found mainly in otariid mammals, and in accidental hosts such as man, dog and jackal. Human become infected with *A. pacificus* by eating raw marine fish as cebiche, tiradito, and chinguirito.

A human case identified as *Diplogonoporus* sp. was recorded in Chile by Wilhelm (1958); however, Figure 1 of the article shows proglottids with lateral and non-ventral and submedian genital pores as with *Diphyllbothrium* (syn. *Diplogonoporus*) (Torres et al., 1977).

Figure 1. Adult of *Dibothriocephalus latus* eliminated by a human patient

1.1.1.5 Asia

Cases of human infection by *D. nihonkaiensis* corresponds mainly to northeastern Asia (Scholz and Kuchta, 2016; Kuchta et al., 2017) and it would be the main cause of dibothriocephalosis in Japan and in Russia (Yamane et al., 1986; Dick et al., 2001; Cai et al., 2017). In Japan, numerous species of *Diphyllbothrium* have been reported (Kuchta et al., 2015a). However, molecular identification showed recently that most of these species were *D. nihonkaiensis*. Arizono et al. (2009b) based on a study of 149 cases, estimated that the mean incidence of infection by *D. nihonkaiensis* in Kyoto was 0.32 cases/100,000 inhabitants per year between 1988 and 2007, while in 2008, 1 case/100,000 inhabitants was detected.

In Korea, using genetic probes around 77 cases of *D. nihonkaiensis* have been identified since 1998 (Jeon et al., 2009; Kim et al., 2014; Shin et al., 2014; Song et al., 2014; Choi et al., 2015; Go et al., 2015; Park et al., 2015).

In China, about 11 cases of *D. latus* were reported between 1927 and 2011, mostly identified by morphological characteristics (Chen et al., 2014). Between 2008 and 2015 about 12 cases of *D. nihonkaiensis* have been reported (Chen et al., 2014; Zhang et al., 2015; Cai et al., 2017). Two of 4 cases reported recently acquired the infection in Canada and Russia (Cai et al., 2017). Also, a pediatric case of *D. latus* was confirmed by molecular analysis and attributed to consumption of imported raw fish from

endemic areas (An et al., 2017).

In Russia, the parasite was reported as *Diphyllbothrium klebanovskii*, a synonym of *D. nihonkaiensis*, before the use of molecular identification (Arizono et al., 2009a). Few human cases, probably due to *A. pacificus*, have been reported in Sakhalin Island (Far East Russia) Kuchta et al., 2015b). *Dibothriocephalus dendriticus* has infrequently been reported in human in the region of Siberia, especially from the surroundings of Lake Baikal (Kuchta et al., 2013).

About 200 cases of human infections due to *Diplogonoporus* tapeworms, a synonymous of *Diphyllbothrium*, have been recorded in Japan (Yamasaki et al., 2012). Also, have been identified a case in Korea (Chung et al., 1995).

Twenty four human cases of *D. stemmacephalum* have been reported in Kanagawa Prefecture and the southwestern part of Japan as well as Okinawa Island and a coastal village in Korea (Yamasaki et al., 2016).

1.1.1.6 Australia and New Zealand

Autochthonous human cases of dibothriocephalosis have not been identified in Australia (Bearup, 1957), but one case by *D. nihonkaiensis* in New Zealand was reported (Yamasaki and Kuramochi, 2009).

1.1.1.7 Africa

In fecal samples of children from Nigeria have been founded eggs morphologically identified as *D. latus* (Ihesiulor et al., 2013), but more studies with adult parasites and molecular testing are required.

Different species of *Dibothriocephalus* have been responsible for few human cases from the India, Indonesia, Malaysia, Middle East, Mongolia, Saudi Arabia, and Taiwan (Alkhalife et al., 2006; Rohela et al., 2006; Devi et al., 2007; Lou et al., 2007; Margono et al., 2007; Myadagsuren et al., 2007). Some cases were probably imported from areas of endemicity (Alkhalife et al., 2006).

1.1.1.8 Sparganosis

Despite being a common infection in definitive hosts, cats and dogs, throughout much of the world, human cases of infection by *Spirometra* plerocercoid larvae or sparganosis occur mostly in East and Southeast Asia. Human sparganosis has cosmopolitan distribution and its endemic area include the East and Southeast Asia (China, Japan, Korea, Thailand, and Vietnam), and the USA, with sporadic cases reported elsewhere in Africa, South America, Europe, and Australia (Rahman et al., 2011; Tappe et al., 2013; Liu et al., 2015; Eberhard et al., 2015; Kuchta et al., 2015a). Imported cases have been described from people after travelling in endemic countries (Botterel and Bourée, 2003).

More than 1,685 cases have been reported worldwide, with more than 80.6% of them from China, 7.9% from Korea, 3.7% from USA and Thailand, 1.5% from Japan and between 0.1-0.5% of the cases from other countries (Argentina, Australia, Czech Republic, Ecuador, France, India, Italy, Paraguay, Sri Lanka, Tanzania, The Republic of Mozambique, Uganda, United Kingdom, and Venezuela) (Liu et al., 2015). In South America cases of human sparganosis also have been reported from Bolivia, Brazil, Colombia, Guiana, and Peru (Oda et al., 2016).

Spirometra erinaceieuropaei is found more frequently in the Old World and *S. mansonoides* occurs mainly in the New World. Recently, the DNA sequences were obtained for the first time from the *Spirometra* spp. from Brazil and were reported two species, *Spirometra* sp. 1 in the ocelot *Leopardus pardalis*, and *Spirometra* sp. 2 in the hoary fox *Lycalopex vetulus* and in the wild tropical rattlesnake (*Crotalus durissus*) (Almeida et al., 2016). These species differed from Asian isolates of *S. erinaceieuropaei* (Almeida et al., 2016). *Spirometra decipiens* is distributed in Brazil, China, and Korea and recently, a morphological study of

two mitochondrial *cox1* and *nad3* genes of the *Spirometra* species from USA were sequenced concluding that also *S. decipiens* is present in that country (Jeon et al., 2016). These same authors suggest that *S. decipiens* was spread from South to North America through their hosts, dogs and cats.

A sister species to *S. erinaceieuropaei* has been responsible for few human cases in Ethiopia and Soudan (Eberhard et al., 2015). Thirty cases of sparganosis have been reported from Africa (Schmid and Watschinger, 1972). Molecular studies are need to determine if this species is related to species of *Spirometra* known to commonly infect, as definitive hosts, to large carnivores in East Africa (for example, *S. theileri* or *S. pretoriensis*).

Human cases of sparganosis by *Sparganum proliferum* have been recorded in North and South America and Asia (Noya et al., 1992). Kuchta et al. (2015a) compiled 16 cases reported from Japan, China, USA, Venezuela, Paraguay, and Czech Republic. Also, two cases were reported in Thailand (Anantaphruti et al., 2011). Some cases of infection in cats, dogs and feral hogs have been identified (Drake et al., 2008).

1.1.2 Symptomatology, morbidity, and mortality

Human dibothriocephalosis can cause important morbidity. In addition to be responsible of intestinal disorders and anemia, which can lead to specialized consultations and expensive complementary analyses; the parasitosis can have a substantial psychological impact on patients and their families, because proglottids of parasite are evacuated over a long period of time. The average cost for the management of a single dibothriocephalosis case has been evaluated to € 400 (Desvois et al., 2001). Human infection becomes patent (passage of proglottids or eggs in stools) after approximately 15 to 30 days after ingestion of plerocercoid (Freeman and Jamieson, 1972; von Bonsdorff, 1977; Curtis and Bylund, 1991; Go et al., 2015) and depend of diphyllbothriid and host species and other factors (Section 1.3). The infection is often asymptomatic (Kamiya and Ooi, 1991; Torres, 2013). People are just alerted of the presence of the tapeworm in the intestine because of the expulsion of chains of proglottids with the stools (Torres, 2013). The adult parasite can also be found accidentally when performing a colonoscopy (Lai and Steinhart, 2007; Stanciu et al., 2009). It is found attached to the mucosa in the small intestine (mostly in the ileum and jejunum). It measures from 1 m to 12 m, but could be up to 25 m in *D. latus* species (Table 1). It could live 10 to 15 years, but an infection lasting about 30 years has been reported (von Bonsdorff, 1977).

Table 1. Geographical distribution and morphological characteristics of adult stages of *Dibothriocephalus* spp., *Adenocephalus pacificus*, and *Diphyllobothrium balaenopterae*

Characteristics	<i>D. latus</i> ^a	<i>D. nihonkaiensis</i> ^b	<i>D. dendriticus</i> ^c	<i>A. pacificus</i> ^d	<i>Di. balaenopterae</i> ^e
Geographical distribution	Asia, Europe, North America, and South America (Chile and Argentina; also some human cases in Brazil)	China, Japan, South Korea, Far East, Russia up to Kamchatka Peninsula, North America; Human sporadic cases in France, Switzerland, and New Zealand.	North Hemisphere: Arctic, subarctic and temperate areas. South America (Southern Chile and Argentina)	South Hemisphere: Australia, Argentina Brazil, Chile, Ecuador, Peru, , Uruguay, South Africa, North hemisphere: Canada, Japan, Russia. United States, (Sporadic cases in Spain)	Human cases in Japan. Sporadic cases in Korea and Spain In marine mammals in South Atlantic Ocean, Arctic and Antarctic, and Pacific Ocean.
Eggs (range of length/wide by μm) in different hosts	48 to 76 by 33 to 59.3 (Humans)	54 to 76 by 35 to 58 (Humans)	49 to 70 by 30 to 52 (Humans) 55 to 69 by 38 to 52 (dogs) 55 to 62 by 35 to 41 (gulls)	48.5 to 60 by 36 to 50 (humans) 41 to 67.5 by 35 to 48 (marine mammals) 49 to 60 by 36 to 45 (dogs)	57 to 81 by 34 to 51 (Humans)
Worm size: Length (in m) by wide in mm	as long as 25 length, 10 to 12 by 8 to 20 (Humans)	as long as 6 m by 5 to 12 (Humans)	2 by 20 (Humans) 0.1 to 1.4 by 9 (dogs) 0.3 to 1 by 5.6 to 10 (gulls)	as long as 2 by 5 to 10 (Humans) 0.8 length (dogs) 1.1 by 10 (marine mammals)	as long as 6.4 by 21 (Humans) 2.8 length (dogs) 7.4 by 10 (marine mammals)
Scolex length by wide in mm	Spatulate, ovalate or cordate. 1.5 to 2.7 by 0.6 to 1.0	Spatulate, 1.2 to 2.8 by 0.7 to 1.5	Spatulate or lanceolate, 0.5 to 2.2 by 0.5 to 0.8	Lanceolate, elongate, or oval, 1.8 to 4.0 by 0.6 to 3.5	Subglobular, 1.3 to 2.5 by 1.3
Neck length by wide in mm	4.1 to 30 by 0.6 to 1.4	6 to 16.8 by 1.2 to 1.3	0.6 to 3.2 by 0.3 to 0.8.	0.7 to 1.1 length.	Not present
Proglottids (PR)	Up to 4000	Up to 2200	Up to 968	Up to 269	Up to 2,648
Size of gravid proglottids (PR): Wide by length in mm	10 to 15 by 2 to 5 (Humans) In experimental infections (1 to 2 months) last PR can be longer than wide.	6.7 to 12 by 1.9 to 2.5 (Humans)	2.4 to 10 by 0.7 to 3.9 (Humans) Distal PR 1.8 to 3.1 by 2.3 to 8.0. Gravid PR with concave laterals margins. Pointed projections in the union of PR.	4 to 6.6 by 4 to 5.5 with tegmental papilla - like protuberances between margin PR and male gonopore	2.8 to 11.2 by 0.4 to 2.1
Distribution of vitelline follicles (VF) and testes (TE)	In lateral fields without confluence anterior to GA, except in some PR longer than wide. Area usually free of VF and TE between neighboring PR	Similar to <i>D. latum</i>	In lateral fields, often with confluence anterior to GA. VF not overlaps the uterine coils. No area free of VF and TE between neighboring PR	In lateral fields, slightly overlapping ends of loops of gravid uterus. TE not confluent at anterior margin of PR. VF sometimes confluent at anterior margin of PR	In two laterals and central field between 2 uteri, excepting areas around uteri and both GA. Sometimes VF confluent at anterior margin of PR
Gonopores in genital atrium (GA) surrounded by papillae	Yes, Ventral in first half of ventral surface	Yes, Ventral, on the middle at 1/3 or 1/4 anterior of the PR	Yes. In anterior third of PR. In PR longer than wide is posterior to the middle line	No, gonopores separates	Yes, with 2 GA, near anterior margin of PR, surrounded by papillae

Characteristics	<i>D. latus</i> ^a	<i>D. nihonkaiensis</i> ^b	<i>D. dendriticus</i> ^c	<i>A. pacificus</i> ^d	<i>Di. balaenopterae</i> ^e
Cirrus sac (CS). Length by wide in μm	Round or oval, with microtriches. 375 to 640 by 245 to 390. Horizontal or oblique in sagittal section	Oval, 430 to 480 by 275 to 390. Open obliquely in the genital atrium	Oblique or oval with microtriches. Somewhat oblique in sagittal section.	Subspherical to pyriform, unarmed. 105 to 325 by 89 to 243 in lateral view	Unarmed, pyriform, 262 to 600 by 250 to 368
Seminal vesicle (SV) Length by wide in μm	172 to 357 by 130 to 233. Dorso-caudal to CS. Visible ventrally in whole mounts (WM). Obtuse angle between CS and SV longitudinal axis	Round to elliptical 300 to 420 by 170 to 300. Dorso-caudal to the CS. Visible ventrally in WM. Sharp angle between CS and SV longitudinal axis	Round shape. Dorso-caudal to CS. Generally not ventrally visible in WM	Oval in ventral view 160 to 377 by 111 to 299 to almost elliptical in lateral view 76 to 223 by 106 to 291 in dorsoventral length. Angle between CS and longitudinal axis (LA) of PR: 46°-94°. Angle between LA of SV and CS: 20°-143°	Ovoid to spherical. 151 to 230 by 110 to 177 extending slightly posterior from proximal end of CS
Shape of ovary	Variable. Dumbbell - like with 2 rounded or elongate lobes	Kidney shaped, without horns	Variable. Dumbbell shape. With 2 flattened elongated or round lobes	Asimetric bilobed, reticulate extended anteriorly adjacent to proximal uterine loops	Bilobed , reticulate extended anteriorly for both sides of the proximal coils
Number of uterine coils to each side/Uterine pore (UP)	4 to 8 extended up to posterior margin of CS. UP to 260-1240 μm , posterior to GA	6 to 7 no extended to the anterior margin of GA. UP to 150 to 300 μm of GA	2 to 9 extended up to CS. UP to 100 μm of GA	4 to 8 extended up to border of male gonopore. UP: 111 to 299 μm from vaginal aperture	3 to 5 loops extended up to the anterior margin of GA. UP to 100 to 200 μm from GA
Microtriches size (in μm)	Up to 4	8 to 9	7 to 10	Gladiate spinitriches (2) and capilliform filitriches	3.8

References: ^aAndersen, 1971, 1975; Andersen et al., 1987; von Bonsdorff, 1977; Dick and Poole, 1985; Kamo, 1999; Kuchta et al., 2015b; Rausch and Hilliard, 1970; Scholz et al., 2009; Torres, 2013; Torres et al., 1989a, 1993; Yamane et al., 1989a; Yazaki et al., 1984; ^bAndo et al., 2001; Arizono et al., 2009b; Choi et al., 2015; Fang et al., 2015; Jeon et al., 2009; Kamo, 1999; Wicht et al., 2008b; Yamane et al., 1986; Yamasaki and Kuramochi, 2009; Yera et al., 2006; ^cAndersen, 1975; Andersen et al., 1987; Dick and Poole, 1985; Figueroa et al., 1979, 1980; Kuchta et al., 2013; Rausch and Hilliard, 1970; Torres et al., 1981b; ^dBaer et al., 1967; Cattán et al., 1977; Escalante and Miranda, 1986; Hernández-Ortiz et al., 2015; Jacobus et al., 2016; Jimenez et al., 2012; Kuchta et al., 2015b; Maejima et al., 1981; Miranda et al., 1968; Rausch et al., 2010; Torres et al., 1983; Tsuboi et al., 1993; ^eArizono et al., 2008; Clavel et al., 1997; Kino et al., 2002; Maejima et al., 1990; Nakamura et al., 1993; Rausch, 1964; Suzuki et al., 1988, Suzuki et al., 1993.

When the infection is symptomatic, the clinical signs are limited and polymorphous: common manifestations may include abdominal discomfort, abdominal pain, diarrhea, transit disorders, constipation, weight loss, asthenia, and vertigo. An extensive field investigation in Finnish Karelia showed that the worm carriers had a very significant higher frequency of fatigue, weakness, diarrhea and numbness of extremities (von Bonsdorff, 1977). A sensation of hunger and "craving for salt" was also associated with worm infection. Uncommon manifestations are headache, allergic

reactions, pain in the tongue exacerbated with food, intestinal obstruction and a case in which *D. latus* was found in the gallbladder at cholecystectomy (von Bonsdorff, 1977). Patients can present low B12 levels, eosinophilia and, rarely, anemia (Nyberg et al., 1961; Kamiya and Ooi, 1991; Vuylsteke et al., 2004; Paugam et al., 2009).

Megaloblastic anemia due to vitamin B12 deficiency has been described in case of prolonged or heavy infection to *D. latus* in malnourished Finnish population after world war II

(von Bonsdorff, 1977), but it is only rarely reported in other cestode infection (Golay and Mariaux, 1995). Approximately 80% of the B12 intake is absorbed by the worm. The mechanism is a parasite-mediated dissociation of the vitamin B12-intrinsic factor complex within the gut lumen, making B12 unavailable to the host. Megaloblastic anemia occurs in 2% or less people infected with *D. latus*; but about 40% of them may have low B12 levels (Nyberg et al., 1961). In Chile, 3 cases of megaloblastic anemia have been reported associated to *D. latus* infection (Osorio et al., 1974; Cristoffanini et al., 1976; Donoso-Scroppo et al., 1986). This deficiency may produce damage to the nervous system, including peripheral neuropathy or central nervous system degenerative lesions (von Bonsdorff, 1977). Unlike *D. latus*, infection with *A. pacificus* is seldom associated with megaloblastic anemia or vitamin B12 deficit and has been reported in only one case (Jimenez et al., 2012). Iron deficiency anemia has rarely been associated with human infection (Medina et al., 2002; Stanciu et al., 2009).

The diagnosis of human infection is based on the identification of proglottids expelled with the stool or found after colonoscopy and identification of eggs after stool examination. In this last case, it is important consider that eggs of *Bothriocephalus* (adult parasites of fish) ingested accidentally by humans, are similar from that of *Dibothriocephalus*, *Diphyllbothrium* or *Adenocephalus*, using light microscopy by inexperienced microscopists. Differences between genera can be established by molecular identification (Yera et al., 2013). Analysis of nuclear genes (28S) allowed the identification of *Bothriocephalus* species. That of mitochondrial genes (*cox1*, NADH dehydrogenase subunit 3 *nd3*) allowed differentiating the most common *Dibothriocephalus*, *Diphyllbothrium* or *Adenocephalus* species involved in human infections (Yera et al., 2008). Analysis of nuclear genes (18S) is necessary to identify other species related to *Dibothriocephalus* or *Diphyllbothrium* (Waeschenbach et al., 2017). Recently, Lestnova et al. (2016) using light and scanning microscopy established differences between eggs of *D. latus*, *D. nihonkaiensis*, *D. dendriticus*, and *A. pacificus*. Also, a pyrosequencing method to differentiate among *D. dendriticus*, *Dibothriocephalus ditremus* (a not zoonotic parasite that live as adult in aquatic birds), *D. latus*, *D. nihonkaiensis*, *Di. stemmacephalum*, *Di. balaenopterae*, *A. pacificus*, *Sp. decipiens*, and *Sp. proliferum* has been described based on the mitochondrial cytochrome c oxidase subunit 1 (*cox1*) gene as a molecular marker (Thanchomnang et al., 2016).

Human sparganosis can be responsible of death when the larvae invade vital organs (Phunmanee et al., 2001; Liu et al., 2015). It causes important morbidity. Severe health consequences can occur (i.e. blindness, immobilization, respiratory disorders, and neurological disorders) and lead to partial or total incapacity. The larvae migration into tissues can cause non-specific discomfort, eosinophilia, inflammation or abscess and others serious complications to vital organs. The larvae could live for up to 20 years in humans (Lee et al., 2010; Presti et al., 2015; Liu et al., 2015) and over 5-25 years in infection by *Sp. proliferum* (Liu et al., 2015). The survival of plerocercoids might be in relation with capacity to modify immune reactions in their

hosts (Lee et al., 2010). The larvae have different locations in humans: intestine wall, scrotum, epididymis, ureter, subcutis, muscles, brain, cerebellum, eye, spinal cord, urinary tract, pleural cavity, pericardium, abdominal viscera, and oral cavity (Beaver and Rolon, 1981; Gutierrez, 2000; Iamaroon et al., 2002; Eom and Kim, 2009; Anantaphruti et al., 2011; Liu et al., 2015). Usually plerocercoids are slightly encapsulated in the tissues but insufficiently to prevent migration; in most cases the capsule is not present (Mueller, 1961).

The incubation period of human sparganosis depends on the route of infection: it is usually 6-11 days to some months, but it might be up to several years in oral infection (Lv et al., 2010). The first symptoms correspond to the migration of the larvae in the tissues. A review on human sparganosis in different countries reported 26% of cerebrospinal, 13% of subcutaneous, 4% of ocular, 1% of visceral, and 0.9% of sparganosis, but in 54% of cases the location of the infection was unknown (Liu et al., 2015). In Thailand, the locations of plerocercoids were the following: 46% subcutaneous, 35% ocular, 14% cerebral and 6% visceral (Anantaphruti et al., 2011). In Africa, 16 of 30 cases were subcutaneous (Schmid and Watschinger, 1972).

In cerebrospinal sparganosis, larvae invade and grow in the spinal canal or in the cerebral hemispheres, and, in some cases, extend to the cerebellum. The neurological symptoms, depending of the location of the parasite, are various as fatigue, limb weakness, confusion, headache, seizure, memory loss, coma, fever, paresthesia (as example, numbness or tingling) and hemiparesis (Kuchta et al., 2015a).

Subcutaneous sparganosis manifests as subcutaneous tumour or cyst. The lesion is slow growing and could be itchy, inflamed or painful. Sometimes, it is migratory or the larva emerges outside the skin (Eberhard et al., 2015). A single nodule is often present, whereas many nodules can arise in other larval cestode infections in humans, as example due to *Taenia solium* (Liu et al., 2015).

Eye is the main invasive location, even if ocular sparganosis is rare. The symptoms depend on the depth of the larva invasion. Extraocular infections (mostly subconjunctival or periorbital) are frequent. At the onset of the disease, patients present palpebral edema or conjunctivitis with eye pain, then kerato-conjunctivitis, ulceration of the cornea. Rarely, the retrobulbar area and the anterior chamber are involved. At this stage, patients had hypopyon, foreign body in the eye which can cause blindness and secondary glaucoma (Ament and Young, 2006). Visceral sparganosis can lead to abdominal pain, intestinal obstruction, peritonitis, pulmonary nodules with or without cavitation or eosinophilic pneumonia (Cheng et al., 2014).

Proliferative sparganosis is caused by *Sp. proliferum* a rare disease that affect particularly to immunocompromised patients (Schauer et al., 2014). The patients present tumour-like, subcutaneous nodules that can spread to other parts of the skin, muscles, brain, lung, and abdomen or a bone tumor-like (Nakamura et al., 1990). Most cases are

lethal (Kuchta et al., 2015a). The plerocercoid larva invades various organs of the body (including bone and spinal cord). It is pleomorphic as it forms continuous branches and buds to produce many progeny in a single site. The progenies can migrate to other tissues and repeat the processes of invasion, proliferation and dissemination. In an immunocompetent patient with molecular identification *Sp. proliferum*, the infection present only subcutaneous manifestations without signs of a proliferative course as dissemination and branching or budding (Schauer et al., 2014).

Specific imaging signs are present in cerebral sparganosis (Hong et al., 2013; Song et al., 2007) and subcutaneous sparganosis (Sakurawa et al., 2007) at CT, MRI and ultrasonography and could allow the diagnosis of the infection. Eosinophilia is possible. Serologic tests (detection of antibodies or antigens from peripheral blood or cerebrospinal fluid) are useful for diagnosis, but available in endemic countries. They are sensitive and enough specific, but cross-reactivity is possible in patients presenting cysticercosis, echinococcosis, paragonimosis, clonorchiosis, and schistosomiasis (Cui et al., 2011). In consequence, diagnosis is based on the histological identification of the larvae in tissues. PCR seems promising for identification of larvae from archival tissues and for species identification.

1.2 Taxonomic Classification of the Agents

1.2.1 Taxonomy and physical description of the agent

1.2.1.1 General aspects

The Diphyllbothriidae family includes zoonotic parasite species of importance in human and animal health which are grouped principally into genera *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, and *Spirometra*. In general, the species of those genera have larval stages which develop in the aquatic environment (coracidia), and have intermediary host copepods (proceroids) and fish (plerocercoids or sparganums) in their life cycles. In addition, these species include a definitive host, where they develop into the adult stage in the small intestine (mammals or birds, the latter, in only some species of *Dibothriocephalus*). Life cycle in *Spirometra* spp. is similar, but, plerocercoid develops in amphibians, snakes, birds and mammals, including humans, and their definitive host corresponds to canids and felids. *Dibothriocephalus* spp. and *Spirometra* spp. occur in freshwater and terrestrial habitats, while *Adenocephalus*, including *A. pacificus*, and *Diphyllbothrium*, including *Di. stemmacephalum* and *Di. balaenopterae*, occur in marine habitats (Waeschenbach et al., 2017).

1.2.1.2 Taxonomic position

The parasites of the family Diphyllbothriidae are included in the phylum Platyhelminthes, subphylum Rhabditophora, class Neophora, subclass Neodermata, infraclass Cestoda, and order Diphyllbothriidea (Ruggiero et al., 2015). Very recently, a new molecular phylogeny of

diphyllbothriideans species has been published (Waeschenbach et al., 2017). It is based on analysis of two nuclear genes (almost completed small subunit ribosomal RNA gene, 18S and partial large small subunit ribosomal RNA gene, 28S) and two mitochondrial ones (partial small subunit ribosomal RNA gene, 16S and cytochrome c oxidase subunit 1 gene, *cox1*). Diphyllbothriideans species are classified into three different families: Cephalochlamyidae, Solenophoridae and Diphyllbothriidae. In this classification, the family Diphyllbothriidae presents 13 valid genera (Waeschenbach et al., 2017). Of these genera, *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, and *Spirometra* are more frequently associated with human disease. The genus *Dibothriocephalus* was resurrected including freshwater and terrestrial species with *D. latus*, *D. nihonkaiensis* and *D. dendriticus* (ex-*Diphyllbothrium latum*, *D. nihonkaiense* or *D. dendriticum*). *Diplogonoporus* is determined as a junior synonym of *Diphyllbothrium*, as suggested by previous molecular data (Yamasaki et al., 2012). *Diphyllbothrium* accommodate parasites from cetaceans including the type species *Di. stemmacephalum* and *Di. balaenopterae*.

Six *Dibothriocephalus* species (*D. latus*, *D. dendriticus*, *D. nihonkaiensis*, *D. alascensis*, *D. dalliae*, and *D. ursi*), 9 *Diphyllbothrium* species (*Di. stemmacephalum*, *Di. elegans*, *Di. cameroni*, *Di. cordatum*, *Di. hians*, *Di. lanceolatum*, *Di. orcini*, *Di. scoticum*, and *Di. balaenopterae*), and the only *Adenocephalus* species (*A. pacificus*) have been reported in humans (Hernandez-Ortiz et al., 2015; Scholz and Kuchta, 2016; Waeschenbach et al., 2017). The species most frequently associated with human infection are *D. latus*, *D. nihonkaiensis*, *D. dendriticus*, *A. pacificus*, and *Di. balaenopterae* (Yamasaki et al., 2012; Scholz and Kuchta, 2016).

Faust et al. (1929) established *Spirometra* as a subgenus of *Diphyllbothrium* considering characteristics of the eggs and uterine coils. However, Mueller (1937) included *Spirometra* as an independent genus. Of the 11 species considered by Wardle and McLeod (1968), Kamo (1999) considers as valid species to *S. erinaceiropaei* (syn. *S. mansoni*, *S. erinacei*), *S. pretoriensis*, *S. theileri* and *S. mansonoides*. Of the *Spirometra* species, the most related to human cases are *S. erinaceiropaei*, *S. mansonoides*, and *S. decipiens*. *Sparganum proliferum* corresponds to a larval stage (plerocercoid), while the adult is unknown. Plerocercoid has been considered be an aberrant form of *S. erinaceiropaei* or *S. mansonoides*. Through the use of genetic testing it was concluded that *Sp. proliferum* is different from *S. erinaceiropaei* and molecular data suggest it is a species of *Spirometra* (Miyadera et al., 2001; Okamoto et al., 2007; Liu et al., 2015; Almeida et al., 2016).

1.2.1.3 Physical description

1.2.1.3.1 Adult stage

Morphological characteristics of *Dibothriocephalus* spp., *A. pacificus*, and *Di. balaenopterae* are indicated in Table 1. Adults can be very long (Figure 1); *D. latus* can reach 25 m in length by 2 cm in width (von Bonsdorff,

1977). At their anterior end they have a scolex with two longitudinal grooves or bothria (Figure 2), ventral and dorsal ones, which adhere to the mucosa of the small intestine. The scolex presents glands whose ducts open in the tegument (Kuperman and Davydov, 1982). A neck posterior (Figures 2-3) to the scolex of variable length gives

rise to a strobila consisting of segments or proglottids (Figure 3), usually wider than they are long. The tegument has finger-like projections or microtriches that increase the surface area 10-fold facilitating an efficient transport of all of the dietary requirements into the parasite (Dalton et al., 2004).

Figure 2. Scolex showing a bothrium (B), and the neck (N) of adult stage of *D. latus*. (Scanning electron microscopy, SEM)

Figure 3. Scolex (S), neck (N) and immature proglottids (I) of adult of *D. latus*. Semichon's acetic carmine stain

The male and female reproductive organs included in each proglottid a set (*Dibothriocephalus*, *Diphyllbothrium*,

Adenocephalus, *Spirometra*), or a paired (*Dibalaenopterae*) of reproductive organs (Bray et al., 1994; Kamo, 1999). Male reproductive organs include testes (Figures 4-5) with their respective efferent ducts that join together in a deferens duct which widens forming an external muscular seminal vesicle (Figure 6) attached to the proximal part of the cirrus sac (Figure 6) including the copula organ (cirrus) that opens in the ventral gonopore (Figure 6). In *Spirometra* spp. the dorsal portion of the cirrus sac represents the seminal vesicle from other genera. The female organs include an ovary, usually lobed (Figure 5), in the posterior region of the proglottid provided

with an oviduct, connected to a structure called the ootype that is surrounded by a Mehlis gland, from which originated the uterus. The ootype communicates with the vagina and vitelline reservoir that stores vitelline cells formed in the vitelline follicles. The vagina opens into a ventral gonopore behind the male gonopore (Figure 6). The uterus has a ventral opening (uterine pore), located behind the female gonopore (Figure 6).

Figure 4: Gravid proglottid of *D. latus*: genital atrium (G), seminal vesicle (SV), uterus (U) testes and vitelline glands (TV). Semichon's acetic carmine stain

Figure 5. Gravid proglottid of *Dibothriocephalus dendriticus*: genital atrium (G), uterus (U) testes and vitelline glands (TV), ovary (O). Semichon's acetic carmine stain

Figure 6. Longitudinal section of gravid proglottid of *D. latus*: male (M) and female (F) gonopores and uterine pore (P) in ventral area, seminal vesicle (SV) dorsal and posterior to cirrus sac (C), and uterus (U) with eggs. Hematoxylin and eosin stain

Proglottids of *Dibothriocephalus* spp. and *Diphyllobothrium* spp. have a common genital atrium that includes male and female gonopores. In *Adenocephalus* and *Spirometra* there is no common genital atrium because female and male gonopores open separately (Bray et al., 1994; Hernández-Ortiz et al., 2015). *Diphyllobothrium balaenopterae* presents 2 genital atria (Bray et al., 1994).

Adenocephalus differs from other Diphyllobothriidae with the presence of protuberance like- papillae separated by semi-circular transverse grooves on the ventral surface, between the male gonopore and the anterior border of the proglottids (Rausch et al., 2010; Hernández-Ortiz et al., 2015). The uterus of *Spirometra* resembles a spiral with 2 to 7 compressed and anteriorly extended coils. In

Dibothriocephalus, *Diphyllobothrium*, and *Adenocephalus* the uterus has several loops which may be parallel and extend laterally or rosette-shaped. *Spirometra* eggs tend to

be pointed at one end (Figure 7) (Bray et al., 1994), while in other genera they are rounded (Figure 8).

Figure 7. Egg of *Spirometra* sp. from feces of domestic cat infected with an adult stage: operculum (O)

Figure 8. Egg of *Dibothriocephalus* sp. from feces of a human patient infected with an adult stage: operculum (O)

Adults of *S. erinaceieuropaei* measure about 1 m long by 1 cm wide and *S. mansonioides* measure up to 1.5 m long by 0.7 cm wide (Mueller, 1974; Bowman et al., 2002). The scolex of *S. mansonioides* measures up to 0.5 mm long (Mueller, 1974) with 2 bothria of 1 mm in length; the U-

shaped uterus terminates in the anterior of the proglottid and presents two sections: anterior heavy outer coils and posterior narrow inner coils joined by a narrow duct (Bowman et al., 2002). In *S. mansonioides* the uterus form is simple, uniform and always presents two anterior turns as

opposed to the uterus of *S. erinaceieuropaei* which lacks uniformity in the number of loops, besides having irregular arrangements and sizes (Mueller, 1974; Scioscia et al., 2014). A uterus with 5-7 coils on each side and a gravid proglottid with a width of 3.1-4.2 mm by 2.1-3.2 long in *S. erinaceieuropaei*; and a uterus with 4-4.5 coils and gravid proglottids with a width of 6.8-10.8 mm by 1.1-1.2 mm long in *S. decipiens* have been reported (Jeon et al., 2015). Both species show differences in the *cox1* gene (Jeon et al., 2015). Okino et al. (2017) obtained a clonal population from a single adult specimen designated as a Kawasaki triploid strain whose partial sequence of mitochondrial cytochrome *c* oxidase subunit 1 gene showed more than 98% similarity with isolates from Australia, China and South Korea. However, gravid proglottids of Kawasaki strain showed 3 to 4 uterine coils, showing similarity with *S. decipiens*. Although the etiological agents of sparganosis in Asia can be divided into two major groups by mtDNA sequences

(Eom et al., 2015; Jeon et al., 2015, 2016), their specific status is highly ambiguous, particularly in parthenogenetic triploid clones and further taxonomic reconsiderations are needed (Okino et al., 2017).

1.2.1.3.2 Eggs and larvae

Eggs of Diphyllbothriidae species are surrounded by a thin proteinaceous vitelline capsule secreted by vitelline cells at the time of fertilization, and a shell of highly resistant protein (sclerotin) formed by a polyphenol/quinone tanning process on the inner surface of the vitelline capsule (Conn and Swiderski, 2008). Eggs have a suture at one end, forming an operculum (Figures 7-9). Internally the eggs have a zygote and vitelline cells. The size of the eggs is variable and depends of various factors, such as the host species, the intensity of infection and the time of exposition (Andersen and Halvorsen, 1978).

Figure 9. Uterus of *D. latus* with eggs: operculum (O). SEM

Table 1 shows the interval of measurements of eggs corresponding to *Dibothriocephalus* spp., *A. pacificus*, and *Di. balaenopterae*. The size (length, width and length/width ratio) and surface characteristics of the egg shell, using light and scanning electron microscopy have made it possible to distinguish some diphyllbothriideans, such as those that commonly infect humans (Lestnova et al., 2016). Eggs of *Spirometra* spp. measure about 60-66 μm in length by 35-36 μm wide (Lee et al., 1990; Bowman et al., 2002; Liu et al., 2015).

Coracidium: During their development two syncytial

embryonic envelopes appear: a) an outer envelope, surrounding the inner embryonic components and larval stage, and b) an inner envelope, forming a ciliated embryophore (Figure 10), surrounding the hexacanth larva (Figure 10). Both structures formed the coracidium (Conn and Swiderski, 2008) that measures about 50 μm . The hexacanth larva has a tegument (Grammeltvedt, 1973) and different cell types (Swiderski and Mackiewicz, 2004) and 3 pairs of hooks (Figure 10) that differ significantly between *D. latus*, *D. nihonkaiensis*, and *D. dendriticus* (Bylund, 1975; Yamane et al., 1989b).

Figure 10. Coracidium of *D. latus*: hooks (H) of hexacanth larva and cilia (C). Thionin stain

Proceroid: It develops from the hexacanth larva that hatches in the gut of the first intermediate host (copepod species component of zooplankton) and after being located in its hemocele (Figure 11) (Janicki and Rosen, 1917; Guttowa, 1961; Torres et al., 2004b, 2007). Infective proceroid for the second intermediate host presents calcareous corpuscles, glands with ducts well developed and a posterior invagination (cercomer) containing the embryonic hooks (Figures 12-13) (Janicki and Rosen, 1917;

Grammeltvedt, 1973; Torres et al., 2004b). The tegument of *D. dendriticus* proceroids presents microtriches up to 1.7 μm long (Malmberg, 1971; Grammeltvedt, 1973). Infective proceroids in *D. latus* measure between 180 and 610 μm long, depending on the host species, exposure time and intensity of infection (Guttowa, 1961; Torres et al., 2004b). In *D. dendriticus* they measure between 288 and 576 μm (Halvorsen, 1966) and in *Spirometra* spp. they reach about 260 μm long by 44-100 μm wide (Liu et al., 2015).

Figure 11. Copepod, *Tumeodiaptomus diabolicus*, infected with proceroids (PR) of *D. latus*

Figure 12. Proceroids of *D. latus* extracted from *T. diabolicus*, showing ducts of glands (D), calcareous corpuscles (C) and cercomer (CE)

Figure 13. Cercomer (CE) of proceroid of *D. latus* showing some of their hooks

Plerocercoid: these develop in the second intermediate host from infecting proceroids in ingested copepods. The

plerocercoids measure 1 mm to several cm in length and present a scolex with 2 bothria (Figure 14). The tegument

includes microtriches (Figure 15). Plerocercoids are located in the mesenteries and in different viscera and muscles of fish. The morphological characteristics of the principal species are indicated in Table 2. These larvae present two types of frontal glands (Green and Golden glands) in the parenchyma of scolex and neck (Gustafsson and Vaihela, 1981; Kuperman and Davydov, 1982). The plerocercoids have a muscular layer separating the parenchyma in an internal or medullar region and an external or cortical region (Figure 16), except, in *Spirometra* spp. where there are groups of muscle fibers unevenly distributed. *Spirometra* plerocercoids measure between 1 mm to 50 cm long (Mueller, 1966; Bo and Xuejian, 2006; Anantaphruti et

al., 2011; Tappe et al., 2013; Lee et al., 2014) and present a scolex with 2 bothria and tegument with microtriches up to 2.4 μm in *S. erinaceieuropaei* (Yamane, 1968). *Sparganum proliferum* plerocercoids can reproduce asexually through a process of branching and budding producing abundant progeny that invade various tissues (Anantaphruti et al., 2011; Tappe et al., 2013; Lee et al., 2014). They measure 38 mm long by 3 mm wide (Noya et al., 1992) and their tegument have irregular branches and buds, lacking bilateral symmetry. The parenchyma presents cavities covered by tegument which appear to contain gelatinous material (Noya et al., 1992; Drake et al., 2008).

Figure 14. Plerocercoid of *D. dendriticus*. SEM

Figure 15. Microtriches in the tegument of *D. dendriticus* plerocercoid. SEM

Table 2. Morphological characteristics of plerocercoids of *Dibothriocephalus* spp. and *Adenocephalus pacificus*

Characteristics	<i>D. latus</i> ^a	<i>D. nihonkaiensis</i> ^b	<i>D. dendriticus</i> ^c	<i>A. pacificus</i> ^d
Scolex	Normally retracted and not visible. Anterior extremity with transverse dorso-ventral ridge	Evaginated, with dorsal and ventral median grooves like lips	Partially retracted. Interbothridial groove visible. Transverse fissure (frontal pit) at anterior end varies in depth	Extruded with bothriads of 0.5 to 1.4 mm length
Body	Wrinkled. Body widest at the anterior third	Wrinkled with cylindrical external appearance, body widest at the middle.	Wrinkled	Wrinkled. Body widest at the anterior region
Segmentation and primordia	Absent	Absent	Present in large larvae	Absent
Size: length/wide in mm	2 to 50/1 to 1.1	6.2 to 10.7/0.9 to 1.3	9 to 30/1 to 3. Relaxed in water up to 45 or more mm in length	4 to 25/0.5 to 2.4
Microtriches: length in μm	1 to 3	2.5 to 10	3 to 16	3 to 4
Subtegumentary longitudinal muscles (N° of layers)	1	1	1 to 2	NR ^e
Frontal glands	In the scolex. Extend into first third of body	In the scolex region	In the scolex region	NR
Location	Usually free in musculature or body-cavity; also in pike and salmonids encysted.	Encysted or free in the musculature of anadromous salmonids	Usually encysted in viscera or in body cavity or free in musculature.	Encysted in the surface of stomach, intestine, liver or peritoneum of marine fish

^aAndersen et al., 1987; Andersen and Gibson, 1989; von Bonsdorff, 1977; von Bonsdorff and Bylund, 1982; Torres et al., 1983, 1989a; ^bSuzuki et al., 2010; Yamane et al., 1986; ^cAndersen et al., 1987; Andersen and Gibson, 1989; Halvorsen, 1970; Kuchta et al., 2013; Kuhlow, 1953; Torres et al., 1981b; Yamane et al., 1989a; ^dEscalante, 1983; Escalante et al., 1988; Escalante and Miranda, 1986; Tantalean, 1975; Tantalean and Huiza, 1994; ^eNot Reported

Figure 16. Histological cross section of *D. dendriticus* plerocercoid: tegument (T), and cortical (CO) and medullar (ME) regions, separated by a muscular (MU) layer. Hematoxylin and eosin stain

1.3 Transmission

1.3.1 Life cycle, routes of transmission, and reservoirs

Adults of *D. latus* live in the small intestine of humans, domestic dog, domestic cat, foxes and other fish-eating mammals (von Bonsdorff, 1977; Andersen et al., 1987). Parasite eggs, eliminated with the feces of the host, develop a ciliated larva (coracidium) when in contact with fresh water, depending on the temperature. The coracidium will then hatch, influencing this process the wavelength and intensity of light (Grabiec et al., 1963). In the water coracidia can live between 1 to 3 days depending on the temperature and relative depletion of their energy reserves. Following ingestion of the coracidia by copepods, four possible situations may occur: 1) Hatching of hexacanth larva in the gut and the displacement to the hemocoel which develop into the infecting proceroid for planktivorous fish, 2) hatched larvae can be damaged by host enzymes leaving only a few surviving to develop in the hemocoel, 3) hatched larvae die by action of the enzymes of the host and those who come to hemocoel would die by the host response, and 4) the coracidia remain unhatched in

the copepod due to unfavorable conditions.

Table 3 shows host copepod species of *D. latus*. The infective proceroids measure 500-600 μm in length after 15 to 20 days of exposure to laboratory temperature in *Cyclops strenuus* (Janicki and Rosen, 1917), while in *Eudiaptomus transylvanicus* (syn. *Diaptomus coeruleus*) and *Eudiaptomus gracilis* reach 270-500 μm and 240-450 μm in length, respectively at 12 to 15 days at 18-20°C (Guttowa, 1961). On the other hand, proceroids grow to between 180 to 610 μm in length from 11 days of infection at 20 ± 1°C in *Tumeodiaptomus diabolicus* (syn. *Diaptomus diabolicus*) (Torres et al., 2004b). The establishment of infections in humans depends on their eating habits and whether they are associated to the consumption of dishes made with raw fish, cold smoked, treated by salting or undercooked. Of the 29 species of copepods (Table 3), 18 are highly susceptible to *D. latus*, being 15 (83%) of the order Calanoida and only 3 (17%) of the order Cyclopoida. The susceptibility of some species of copepods depends on their age, physiological compatibility, ecological conditions and geographic origin of host and parasite (Guttowa, 1961; Halvorsen, 1966; Bylund, 1969; Sharp et al., 1990; Marcogliese, 1995).

Table 3. Zooplanktonic copepod hosts of *Dibothriocephalus latus* in different countries

Country	Copepod hosts	Reference
Australia	<i>Mesocyclops leuckarti</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
Australia	<i>Gladiferens brevicornis</i> ^{b,c}	Guttowa, 1961; von Bonsdorff, 1977
Australia	<i>Boeckella minuta</i> ^b	Guttowa, 1961; von Bonsdorff, 1977
Australia	<i>Microcyclops varicans</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
Australia, Poland	<i>Eucyclops serrulatus</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
Chile	<i>Tumeodiaptomus diabolicus</i> (= <i>Diaptomus diabolicus</i>) ^b	Torres et al., 2004b, 2007
Chile	<i>Boeckella gracilis</i> ^b	Torres et al., 2004b, 2007
Chile	<i>Mesocyclops araucanus</i> ^{a,c}	Torres et al., 2004b, 2007
Finland, Poland	<i>Cyclops vicinus</i> ^a	Guttowa, 1961; von Bonsdorff, 1977
Finland, Poland	<i>Eudiaptomus zachariasii</i> ^{b,c}	Guttowa, 1961; von Bonsdorff, 1977
Finland, Poland	<i>Cyclops insignis</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
Finland, Germany, Karelia, Norway, Poland, Switzerland	<i>Cyclops strenuus</i> (= <i>C. furcifer</i>) ^a	Guttowa, 1961; von Bonsdorff, 1977
Germany, Poland	<i>Eudiaptomus transylvanicus</i> (= <i>E. coeruleus</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
Germany, Finland, Ireland, Karelia, Norway, Poland, Switzerland	<i>Eudiaptomus gracilis</i> ^b	Hickey and Harris, 1947; Guttowa, 1961; von Bonsdorff, 1977
Karelia	<i>Thermocyclops oithonoides</i> (= <i>Mesocyclops oithonoides</i>) ^{a,b}	Guttowa, 1961; von Bonsdorff, 1977
Karelia, Finland, Germany	<i>Eudiaptomus graciloides</i> ^b	Guttowa, 1961; von Bonsdorff, 1977
Norway	<i>Acanthodiaptomus denticornis</i> ^b <i>Cyclops lacustris</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
Russia	<i>Arctodiaptomus ulomskyi</i> ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Acanthocyclops vernalis</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Eurytemora affinis</i> ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Tropocyclops prasinus</i> ^{a,c}	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Skistodiaptomus oregonensis</i> (= <i>Diaptomus oregonensis</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Skistodiaptomus mississippiensis</i> (= <i>D. mississippiensis</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Leptodiaptomus siciloides</i> (= <i>D. siciloides</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Leptodiaptomus minutus</i> (= <i>D. minutus</i>) ^{b,c}	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Aglaodiaptomus leptopus</i> (= <i>D. piscinae</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Onychodiaptomus sanguineus</i> (= <i>D. sanguineus</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977
USA	<i>Mastigodiaptomus albuquerquensis</i> (= <i>D. albuquerquensis</i>) ^b	Guttowa, 1961; von Bonsdorff, 1977

^aOrder Cyclopoida; ^bOrder Calanoida; ^cSpecies less viable to infection or only accidentally infected

Infections in *E. gracilis* cause a significant drop in consumption of oxygen in the host after 6-10 days of exposure, when there is greater demand for building materials and secretion of metabolites by the parasite which could induce a physiological imbalance in the host (Klekowski and Guttowa, 1968). In mixed infections by *D. latus* and *Proteocephalus* sp. in *E. gracilis* the procercoids can cause death of the host during organogenesis (Guttowa, 1966).

Infected copepods transmit the infection to planktivorous fish. The procercoids reach the gut and migrate to the wall of the stomach or intestines, mesenteries, viscera and muscles. In fish, procercoid develops into plerocercoid. These larvae are found free (Figures 17-18) or encapsulated in the tissues (Figures 19-20), depending on the host species. *Dibothriocephalus latus* present about 17 fish host species distributed in 6 orders (Table 4). Infection in native fish from Chile, such as

Galaxias spp., *Percichthys trucha*, *Odontesthes mauleanum*, and *Basilichthys australis* have only been observed in 2 lakes with a low intensity of infection in contrast to the high

parasitic load of introduced rainbow trout (Torres et al., 1989b, 1998, 2004a, 2012).

Figure 17. Free plerocercoid (P) of *Dibothriocephalus* sp. in the muscular surface of a rainbow trout, *Oncorhynchus mykiss*

Figure 18. Histological section of the liver of a rainbow trout with a free plerocercoid (P) of *Dibothriocephalus* sp. Hematoxylin and eosin stain

Figure 19. Encapsulated plerocercoids (P) of *Dibothriocephalus* sp. in the stomach of a rainbow trout

Figure 20. Histological section of muscle of a rainbow trout with an encapsulated plerocercoid (P) of *Dibothriocephalus* sp. Hematoxylin and eosin stain

Table 4. Fish hosts of plerocercoids of *Dibothriocephalus* spp. and *Adenocephalus pacificus*

Species: <i>Dibothriocephalus latus</i> ^a	
Area	Hosts
Europe (EU)	Gadiformes: burbot, <i>Lota lota</i> Perciformes: perch, <i>Perca fluviatilis</i> ruffe, <i>Gymnocephalus cernuus</i> ; Sander, <i>Sander lucioperca</i>
EU and Asia	Salmoniformes: brown trout, <i>Salmo trutta</i> char, <i>Salvelinus fontinalis</i>
EU and North America	Esociformes: pike, <i>Esox lucius</i>
EU and North America and South America	Salmoniformes: rainbow trout, <i>Oncorhynchus mykiss</i>
North America	Perciformes: yellow perch, <i>Perca flavescens</i> wallege, <i>Sander vitreus</i> sauger, <i>Sander canadensis</i> Salmoniformes: Coho salmon, <i>Oncorhynchus kisutch</i>
South America	Osmeriformes: puye, <i>Galaxias maculatus</i> puye, <i>Galaxias platei</i> Atheriniformes: cauque, <i>Odontesthes mauleanum</i> silverside, <i>Basilichthys australis</i> Perciformes: creole perch, <i>Percichthys trucha</i>
Species: <i>Dibothriocephalus dendriticus</i> ^b	
Asia	Salmoniformes: Baikal omul, <i>Coregonus migratorius</i> Gasterosteiformes: nine-spined stickleback <i>Pungitius pungitius</i> Salmoniformes: common whitefish, <i>Coregonus lavaretus</i> vendace, <i>Coregonus albula</i> lake Constance whitefish, <i>Coregonus gutturosus</i> whitefish, <i>Coregonus pallasii</i> Valaam whitefish, <i>Coregonus widegreni</i> Osmeriformes: grayling, <i>Thymallus thymalus</i> Gasterosteiformes: three-spined stickleback, <i>Gasterosteus aculeatus</i>
EU	Salmoniformes: Arctic char, <i>Salvelinus alpinus</i>
EU and North America	Salmoniformes: rainbow trout, <i>Oncorhynchus mykiss</i> ; brown trout, <i>Salmo trutta</i>
EU and North America and South America	Salmoniformes: Atlantic salmon, <i>Salmo salar</i> lake white fish, <i>Coregonus clupeiformis</i> Osmeriformes: rainbow smelt, <i>Osmerus mordax</i>
North America	Salmoniformes: char, <i>Salvelinus fontinalis</i>
North America and South America	

Species: *Dibothriocephalus latus*^a

South America	Salmoniformes:
	Coho salmon, <i>Oncorhynchus kisutch</i>
	Osmeriformes:
	puye, <i>Galaxias maculatus</i>
	Perciformes:
	<i>Percichthys trucha</i>
	Atheriniformes:
	cauque, <i>Odontesthes mauleanum</i>
	silverside, <i>Basilichthys australis</i>
	Siluriformes:
	tollo, <i>Diplomystes camposensis</i> .

Species: *Dibothriocephalus nihonkaiensis*^c

Asia	Salmoniformes:
	masu salmon, <i>Oncorhynchus masou</i> ;
	pink salmon, <i>Oncorhynchus gorbuscha</i> ,
	chum salmon, <i>Oncorhynchus keta</i>
North America	Salmoniformes:
	<i>Oncorhynchus gorbuscha</i>

Species: *Adenocephalus pacificus*^d

South America (coast of Peru)	Perciformes:
	Peruvian grunt, <i>Anisotremus scapularis</i> .
	Corvine drum, <i>Cilus gilberti</i> .
	common dolphinfish, <i>Coryphaena hippurus</i>
	Peruvian weakfish, <i>Cynoscion analis</i>
	kingfish, <i>Menticirrhus ophicephalus</i>
	Peruvian banded croaker, <i>Paralichthys peruanus</i>
	Lorna drum, <i>Sciaena deliciosa</i>
	Callao drum, <i>Sciaena callaensis</i>
	Pacific mackerel, <i>Scomber japonicus</i>
	Atlantic Spanish mackerel, <i>Scamberomorus maculatus</i>
	Eastern Pacific bonito, <i>Sarda chiliensis</i>
	Palm ruff, <i>Seriola violacea</i>
	paloma pompano, <i>Trachinotus paitensis</i>
	Jack mackerel, <i>Trachurus murphyi</i>
	Peruvian rock seabass, <i>Paralabrax humeralis</i>
	Siluriformes:
	Colombian shark fish, <i>Ariopsis seemanni</i> .
	Peruvian sea catfish, <i>Galeichthys peruvianus</i> .
	Ophidiiformes:
	pink cusk-eel, <i>Genypterus maculatus</i> .
South America (coast of Chile)	Gadiformes:
	Peruvian hake, <i>Merluccius gayi peruanus</i>
	Mugiliformes:
	flathead grey mullet, <i>Mugil cephalus</i>
	Carcharhiniformes:
	sickle fin smooth-hound, <i>Mustelus lunulatus</i>
	Pleuronectiformes:
	fine flounder, <i>Paralichthys adspersus</i>
	Plerocercoids identified as Diphyllbothriidae or <i>Diphyllbothrium</i> sp. ^e
	Perciformes:
	corvina drum, <i>Cilus gilberti</i> .
	Gadiformes:
	South Pacific hake, <i>Merluccius gayi gayi</i>
	Southern hake, <i>Merluccius australis</i> ;
	Southern blue whiting, <i>Micromesistius australis</i> ,
	Ophidiiformes:
	pink cusk-eel, <i>Genypterus maculatus</i> .

^aAndersen and Gibson, 1989; De Vos and Dick, 1989; Halvorsen and Wissler, 1973; Janicki and Rosen, 1917; Kuchta et al., 2015a; Mercado et al., 2010; Revenga, 1993; Scholz et al., 2009; Thanchomnang et al., 2016; Torres et al., 1989b, 1998, 2004a, 2012

^bBylund, 1972; Chubb, 1980; De Vos and Dick, 1989; Gustafsson and Vaihela, 1981; Henricson, 1977; Kuchta et al., 2013; Moravec, 2004; Revenga, 1993; Thanchomnang et al., 2016; Torres, 1990; Torres et al., 1981b, 1989b, 1998, 2004a, 2012; Vik, 1957

^cAndo et al., 2001; Fang et al., 2015; Jeon et al., 2009; Kuchta et al., 2017; Suzuki et al., 2010; Yamane et al., 1986; Yanagida et al., 2010

^dEscalante, 1983; Escalante and Miranda, 1986; Chero et al., 2014a, 2014b, 2014c; Iannaconne and Alvarino, 2009; Kuchta et al., 2015b; Tantalean, 1975; Tantalean and Huiza, 1994

^eGarcías et al., 2001; George-Nascimento and Huet, 1984; George-Nascimento et al., 2011; MacKenzie and Longshaw, 1995; Oliva and Ballón, 2002

Chubb (1980) in their review indicate the following species of fish that were experimentally infected with procercoids: *E. lucius*, *Gymnocephalus cernua*, and *P. fluviatilis*. Similarly, in *E. lucius*, *G. cernua*, *P. fluviatilis*, *Rutilus rutilus* and *O. mykiss* plerocercoids infection have been restored after administering orally. Thus, planktivorous fish can transmit the infection to fish-eating fish that act as paratenic hosts, for example, *E. lucius*, *P. fluviatilis* and *L. lota* in the northern hemisphere (von Bonsdorff and Bylund, 1982) or *O. mykiss* and *P. trucha* in the lakes of southern Chile (Torres et al., 1989b, 1998,

2012) and Argentina (Revenga, 1993).

Seven of the 10 species of copepods susceptible to infection by *D. dendriticus* (Table 5) belong to the order Cyclopoida, all described in Europe and North America. In South America the host copepods are unknown. The procercoids of *Dibothriocephalus spp.* in natural infections of *C. strenuus abyssorum* reduce the reproductive capacity of females, the feeding and the respiration rate as well as causing a reduction in motility, and impaired escape responses, which results in the individual being more vulnerable to predators (Pasternak et al., 1995).

Table 5. Zooplanktonic copepod hosts of *Dibothriocephalus dendriticus* in different countries

Area	Copepods	Reference
Canada	<i>Aglaodiaptomus leptopus</i> (= <i>Diaptomus leptopus</i>) ^b	Wright and Curtis, 2000
Canada	<i>Leptodiaptomus minutus</i> (= <i>Diaptomus minutus</i>) ^b	Wright and Curtis, 2000
Finland		Rahkonen and Valtonen, 1998;
Germany		Kuhlow, 1953;
Norway	<i>Cyclops strenuus</i> ^a	Halvorsen, 1966, Halvorsen, 1967;
United Kingdom		Sharp et al., 1990;
	<i>Eudiaptomus gracilis</i> ^b	
Norway	<i>Cyclops abyssorum</i> ^a	Halvorsen, 1966
	<i>Cyclops lacustris</i> ^a	
	<i>Mesocyclops leuckarti</i> ^a	
Norway		Halvorsen, 1966;
USA	<i>Cyclops scutifer</i> ^a	Johnson, 1975; Meyer and Vik, 1963
USA	<i>Diacyclops bicuspidatus</i> (= <i>Cyclops bicuspidatus</i>) ^a	Meyer and Vik, 1963
USA	<i>Acanthocyclops vernalis</i> (= <i>Cyclops vernalis</i>) ^a	Meyer and Vik, 1963

^aOrder Cyclopoida; ^bOrder Calanoida

Fish-eating birds of 9 families (Accipitridae, Alcidae, Vorvidae, Gaviidae, Pandionidae, Pelecanidae, Podicipedidae, Sternidae, and Laridae) distributed in about 50 species represent the final host of *D. dendriticus* (Kuchta et al., 2013). In South America, *Larus dominicanus* (Chile and Argentina) and *Chroicocephalus maculipennis* (Chile) have been recorded with natural and experimental infections (Figueroa et al., 1979, 1980; Torres et al., 1981a,

1981b; Semenas, 2014). Also, infection is present in humans, dogs, cats, red foxes, bears and other mammals (Rausch and Hilliard, 1970; Andersen et al., 1987; Torres et al., 2004a, Wicht et al., 2008a; Kuchta et al., 2013; Catalano et al., 2015). Parasite eggs in the feces of their host spread in freshwater ecosystems developing coracidia in 5 to 6 days at 25°C. Coracidia require light for hatching (Hilliard, 1960; Grabiec et al., 1963). Coracidia infect

copepods (Table 5) then develop into proceroid in 14 to 20 days measuring 288 to 576 µm long in *C. strenuus* (Halvorsen, 1966).

Copepods transmit the infection to planktivorous fish in which the proceroids migrate to the gastrointestinal wall and then the mesenteries, viscera and muscles where they develop into the plerocercoid stage. Table 4 lists 22 species of fish host for *D. dendriticus* distributed in 6 orders. Thirteen of these species are salmoniforms. The location of *D. dendriticus* in the muscles is less frequent than in *D. latus* (Dick and Poole, 1985; Torres et al., 1998, 2004a, 2012; Rozas et al., 2012). The plerocercoids of *Dibothriocephalus* spp., like other helminths induce granulomatous inflammatory response that encapsulates the parasite in order to isolate and eventually destroy it (Feist and Longshaw, 2008), a situation described by different authors (Gonzalez et al., 1978; Sharp et al., 1989, 1992; Torres et al., 1991, 2002, 2012; Revenga et al., 1995). However this reaction can vary drastically between hosts. Bylund (1972) observed varying degrees of response to *D. dendriticus* in 3 host species: 1) in *Coregonus lavaretus* the plerocercoids encyst in the gastrointestinal wall due to a more efficient response that mitigates their migration to other viscera, 2) the response in *Salmo trutta* is slower allowing penetration of the larva to the gastrointestinal wall and then to different organs where they are encapsulated, and 3) in *Coregonus albula* they are not encapsulated, remaining free in the abdominal cavity and causing significant damage with their migration. The lack of encapsulation of *Dibothriocephalus* spp. resulting in greater necrosis, destruction of blood vessels, and hemorrhaging, all of which were observed in some native fish from Lake Panguipulli in Chile (Torres et al., 2012). Experimentally, *D. dendriticus* plerocercoids in fish held at 14 to 15°C exhibit greater activity and motility from the body cavity to the heart, pericardium and muscles that are found in fish held at 7.5-11°C (Rahkonen and Valtonen, 1998).

The adult stage of *D. nihonkaiensis* lives in the intestine of humans and the brown bear (*Ursus arctos middendorffi*) (Yamane et al., 1986; Kamo, 1999; Yera et al., 2006; Arizono et al., 2009a; Yamasaki et al., 2012; Catalano et al., 2015; Choi et al., 2015). Also, Scholz and Kuchta (2016) mentioned as host to *Ursus arctos piscator*, *U. americanus*, *U. thibetanus*, *Canis lupus*, *C. familiaris*, *Vulpes vulpes*, *Neovison vison* and *Sus scrofa*. *Cyclops strenuus*, a freshwater copepod, is an experimental first intermediate host (Eguchi, 1926), but natural copepod hosts are considered to be not known marine species. There are 3 Salmoniformes anadromous species listed as hosts of plerocercoids (Table 4), and which are unlike *D. latus* that develop in freshwater fish. *Dibothriocephalus nihonkaiensis* plerocercoids only have been detected in the musculature (Yamane et al., 1986; Suzuki et al., 2010).

Adult stage of *A. pacificus* develops in the small intestine of marine mammals: *Otaria byronia* (syn. *Otaria flavescens*), *Arctophoca* (syn. *Arctocephalus*) *philippii*, *A. australis forsteri*, *A. pusillus pusillus*, *A. pusillus doriferus*, *A. gazella*, and *A. tropicalis* in the southern hemisphere, and *Callorhinus ursinus*, *Neophoca cinerea*, and

Eumetopias jubatus in the northern hemisphere; also develop in humans, *Canis familiaris* and *Canis mesomelas* (Kuchta et al., 2015b; Hernández-Ortiz et al., 2015; Jacobus et al., 2016). Parasite eggs are eliminated with the feces of the final host completing their development in seawater over 3 days at 22°C and hatching on the fourth day (Escalante et al., 1987). Their first intermediate hosts are unknown but probably correspond to marine copepods. Twenty two species of marine fish included in 7 orders act as intermediate or paratenic hosts, identified in the coast of Peru (Table 4); in Chile the specific identification of plerocercoids of diphyllbothriids registered in marine fish, included in similar orders, genus or species of the fish host of *A. pacificus*, have not been verified (Table 4). The lack of records of *A. pacificus* plerocercoids in the muscles of fish is particularly striking and could be associated with a low level of infection or lack of application of more sensitive detection techniques in this location (Torres and Puga, 2011).

Diphyllbothrium stemmacephalum develops as adult in dolphins and porpoises (Yamasaki et al., 2016). The principal sources of human infection are still unknown (Scholz and Kuchta, 2016).

Diphyllbothrium balaenopterae develops its adult stage in humans and dogs (Rausch, 1964; Yamasaki et al., 2012), but marine mammals are the principal hosts as Mysticeti *Balaenoptera acutorostrata*, *Balaenoptera borealis*, *Megaptera novaeangliae*, *Balaenoptera musculus*, and *Balaenoptera physalus* (Kamo, 1999). Kamo et al. (1973) infected a marine planktonic copepod (*Oithona nana*) an important potential prey for fish larvae, distributed in tropical and subtropical waters of the Atlantic Ocean, in the Mediterranean Sea and the Pacific and Indian Oceans (Cepeda et al., 2012). Some suspected marine fish intermediate hosts are reported, as *Engraulis japonica* (Clupeiformes / Engraulidae), *Sardinops melanostictus* (Clupeiformes, Clupeidae), *Katsuwonus pelamis* (Perciformes, Scombridae) (Yamasaki et al., 2012), as well as *Thynnus* spp. (Perciformes, Scombridae) and *Trachurus japonicus* (Perciformes, Carangidae) (Kino et al., 2002). Nevertheless, today the true source of human infection is unknown.

Domestic and wild canids and felids are the final hosts of *Spirometra* spp.. Adult parasites living in the intestine of *Canis lupus familiaris*, *Cerdocyon thous*, *Lycalopex fulvipes*, *L. griseus*, *L. gymnocercus*, *L. vetulus*, *Felis silvestris catus*, *Leopardus geoffroyi*, *L. guigna guigna*, *L. pardalis*, *L. tigrinus*, *L. wieddi*, *Panthera onca*, *Puma concolor*, and *P. yagouaroundi* in South America (Oda et al., 2016).

Two cases of infection by the adult stage of *S. erinacei* have been reported in human in Korea (Lee et al., 1984). Also, these authors cited 4 additional cases reported in Japan and 2 cases from China.

The eggs of *Spirometra* spp. are eliminated with the host feces and then develop coracidia in fresh water. Cyclopoid copepods act as the first intermediate hosts, developing proceroids; and frogs, water snakes, birds, and mammals act as the second intermediate/paratenic hosts

being located in different tissues as plerocercoid.

Spirometra erinaceieuropaei adult develops principally in dogs, but is also identified in cats (Mueller, 1974). Copepod species of *Mesocyclops*, *Eucyclops*, and *Paracyclops* act as the first intermediate hosts (Venturini, 1989; Denegri and Reisin, 1993; Bowman et al., 2002). Lee et al. (1990) infected tadpoles of *Pelophylax nigromaculatus* (syn. *Rana nigromaculata*) with *Mesocyclops leuckarti* and *Eucyclops serrulatus* infected with plerocercoids. Also, *S. mansonioides* adult develops in the small intestine of domestic and wild felids (Bowman et al., 2002). Coracidia develop in fresh water and infect copepods of the genus *Cyclops*, such as *C. vernalis* and *C. viridis* (Corkum, 1966; Mueller, 1966). The second intermediate host (amphibians, rodents or water snakes) becomes infected through the ingestion of infected copepods; plerocercoids can also develop in the domestic cat (Bowman et al., 2002).

A revision of *Spirometra* in China reported 7 species of *Rana*, and 1 species of the genus *Microhyla*, *Hyla*, and *Paa*, respectively. Also, the authors indicate some reptilian hosts: 6 species of *Elaphe*, 1 species of *Natrix*, *Naja*, *Zaocys*, *Dinodon*, *Deinagkistrodon*, *Xenochrophis*, and 2 species of *Ptyas* and *Bungrarus*, respectively (Liu et al., 2015)

In Malaysia, *Rana limnocharis* and *R. cancrivora*, and in Australia *Litoria caerulea* have been reported as hosts of *Spirometra* (Liu et al., 2015).

Plerocercoids of *S. erinaceieuropaei* have been registered in the reptilians *Natrix natrix* and *N. tessellate* from Turkey (Yildirimhan et al., 2007).

In USA have been reported infection by plerocercoids in amphibians as *Lithobates catesbeianus* (syn. *Rana catesbeiana*) and *Lithobates clamitans* (syn. *Rana clamitans*) and in 7 species of reptilians, including 4 species of *Nerodia* (syn. *Natrix*), *Thamnophis proximus proximus* (syn. *Thamnophis sauritus proximus*), *Lampropeltis getula holbrooki* and *Coluber constrictor flaviventris*; in mammals infection was detected in *Didelphis virginiana*, *Procyon lotor* and in *Urocyon cinereoargenteus* (Corkum, 1966).

Oda et al. (2016) review and include new records of Amphibia and Reptilia hosts of *Spirometra* from South America, including between Amphibia, 3 species of *Rhinella*, 2 species of *Pristimantis*, 2 species of *Hypsiboas*, 1 species of *Leptodactylus*, *Dermatonotus*, *Aplastodiscus*, and *Scinax*, respectively (Oda et al., 2016). Authors include between reptilians, 3 species of *Erythrolamprus*, *Bothrops*,

Chironius, 2 species of *Xenodon* and *Philodryas*, and 1 species of *Ameiba*, *Amphisbaena*, *Aplastodiscus*, *Drymarchon*, *Clelia*, *Leptophis*, *Lygophis*, *Mastigodryas*, *Salvator*, *Scinax*, *Simophis*, *Philodryas*, *Micrurus*, and *Crotalus*.

In Australia, the existence of two genotypes for *S. erinaceieuropaei* have been reported, with a genotype in the *Litoria caerulea* frog different to that found in the dog, fox, cat, tiger snake and python (Zhu et al., 2002).

Humans acquire the sparganosis by ingestion from the first intermediate host in non-drinking water or swimming in unsafe water (Zhou et al., 2005; Eberhard et al., 2015) or, more likely with raw or undercooked second intermediate or paratenic hosts, as frogs or snakes, and crocodiles (Magnino et al., 2009; Liu et al., 2015). The plerocercoid larvae leave their intermediate or paratenic hosts to enter the human body, which is warmer. Infective larvae are released into the digestive tract and penetrate the intestinal wall to develop proceroid to plerocercoid or become established as the latter stage migrating through the intestinal wall, then into the muscle, the viscera or subcutaneous tissue. Also, in East and Southeast Asia, the zoonosis is frequent due to the use of freshwater frogs and snakes-based plasters on eyes or opened wounds in traditional medicine practices, as example, in China, Thailand, and Vietnam (Acha and Szifres, 2003; Wiwanitkit, 2005; Anantaphruti et al., 2011; Liu et al., 2015). Drinking untreated contaminated water is a major infection route in Thailand and in Korea, especially in women and children in the last case (Wiwanitkit, 2005; Anantaphruti et al., 2011; Liu, 2015)

1.3.2 Incubation and pre-patent periods

The incubation period in *Dibothriocephalus* spp., *A. pacificus* and *Di. balaenopterae* infections is variable and depends on the size of the worms, intensity of infection, nutritional status of the host and degree of sensitivity to antigen parasites. Clinical signs usually are not pathognomonic, however, a frequent clinical sign is the periodic elimination of strobila pieces up to 50 cm long with human stools (Kamo et al., 1986; Torres, 2013) and up to 1 m every 1 or 2 months in dogs (Magath, 1929), a situation which occurs after the pre-patent period and when the parasite is mature. The pre-patent period of *Dibothriocephalus*, *Adenocephalus* and *Spirometra* species is variable in different hosts (Table 6). In the majority of species it fluctuates between 6 and 30 days, except in *A. pacificus*, whose period varies between 20 and 55 days.

Table 6. Pre-patent periods of some Diphylobothriidae species

Species	Host	Pre-patent Period (Days)	Reference
<i>Adenocephalus pacificus</i>	Dogs	20 to 55	Escalante and Miranda, 1986; Escalante and Chico-Ruiz, 1988
<i>Dibothriocephalus latus</i>	Hamsters	16 to 22	Andersen, 1971
<i>Dibothriocephalus latus</i>	Humans	25 to 30	von Bonsdorff, 1977
<i>Dibothriocephalus latus</i>	Dogs	18 to 27	Torres et al., 1989a
<i>Dibothriocephalus latus</i>	Cats	27 to 29	Torres et al., 1989a
<i>Dibothriocephalus dendriticus</i>	Humans	15	Freeman and Jamieson, 1972; Curtis and Bylund, 1991
<i>Dibothriocephalus dendriticus</i>	Hamsters	7 to 13	Andersen, 1971
<i>Dibothriocephalus dendriticus</i>	Dogs	12 to 18	Torres et al., 1981b, 1989a
<i>Dibothriocephalus dendriticus</i>	Gulls, <i>Larus dominicanus</i>	12	Torres et al., 1981b, 1989a
<i>Dibothriocephalus dendriticus</i>	Gulls, <i>Chroicocephalus maculipennis</i>	13	Torres et al., 1981a
<i>Dibothriocephalus dendriticus</i>	Gulls, <i>L. argentatus</i>	6	Sharp et al., 1990
<i>Dibothriocephalus nihonkaiensis</i>	Hamsters	20	Yamane et al., 1986
<i>Spirometra erinaceieuropaei</i>	Dogs	15 to 30	Venturini, 1989; Denegri and Reisin, 1993
<i>Spirometra erinaceieuropaei</i>	Dogs	16 to 18	Lee et al., 1990
<i>Spirometra erinaceieuropaei</i>	Cats	15	Lee et al., 1990
<i>Spirometra erinaceieuropaei</i>	Cats	8 to 10	Ooi et al., 2000
<i>Spirometra erinaceieuropaei</i>	Cats	12	Lin et al., 2010
<i>Spirometra mansonoides</i>	Cats	12 to 13	Mueller, 1959

1.3.3 Period of communicability

The adult of *D. latus* have a great capacity for egg production. A patient can eliminate about 122,000 eggs/g

feces with a total of 20 to 40 million eggs daily (von Bonsdorff and Bylund, 1982). Between 50,000 and 400,000 eggs/g of feces were eliminated daily over a 4 month period in an experimental human infection with 5 worms (Kamo et al., 1986). Adults of *D. latus* can live 10 to 15 years in the

human intestine, although there have been cases of infections lasting up to 30 years (Bonsdorff, 1977). In Lake Panguipulli in Chile, prevalence in humans (2.8%) and dogs (4.5%) in the Choshuenco locality and 1.8% in the dogs at the Panguipulli locality in part favoring a high prevalence of infection (83% to 93%) in *O. mykiss*, living in that lake (Torres et al., 2004a, 2012; Torres and Puga, 2011). Control of infection in humans and domestic animals can consequently reduce infection of fish which are food source for the local population and constitute a tourist attraction for sport fishing.

Dibothriocephalus dendriticus lives around 6 months in the gull, *Larus argentatus*; a humoral response has been demonstrated during infection (Sharp et al., 1990), which would be effective in the removal of the parasite but would not cause a protective immunity against reinfection. However, infection in a human case could remain for 2 years (Wicht et al., 2008a).

Spirometra erinaceieuropaei eliminate about 70,000 eggs/g feces after 15 days of infection (Lee et al., 1990). Ooi et al. (2000) estimated in 428,000 eggs/g of feces per day and a total of 14,416,000 eggs per day in a single worm infection in a cat. *Spirometra mansonoides* in cats can live for about 3.5 years (Mueller, 1974).

1.4 Population and Individual Control Measures

1.4.1 Treatment options

Dibothriocephalus species are sensitive to niclosamide (2 g in adult or 50 mg/Kg in children older than 2 years-old, on an empty stomach in two doses an hour apart) (Bylund et al., 1977). The tablets are taken with very few water during a hydric diet (Bourée et al., 2012). The drug inhibits the oxidative phosphorylation of the parasite mitochondria leading to the diminution of energetic capacities, an acid lactic accumulation and the death of the worm (Weinbach and Garbus, 1969). The drug inhibits also the defensive mechanisms of the parasite against the host intestinal proteases. Niclosamide is not available in many countries but is the preferred drug because it is not absorbed in the intestine (Flisser and Willingham, 2013). In general, the drug is well tolerated and no toxic effects have been reported in therapeutic doses besides it has no contraindications and can be used during gestation (Flisser and Willingham, 2013). Niclosamide is also efficient on other cestodes as *Taenia solium*, *Taenia saginata* and *Hymenolepis nana*. Niclosamide is effective, but its administration is sometimes difficult to respect; which may be responsible for therapeutic failure (Bourée et al., 2012).

Dibothriocephalus species are sensitive to praziquantel (25 mg/Kg/day in one dose) (Bylund et al., 1977). This dose is usually used to cure *D. latus* and *D. nihonkaiensis* infections, lower dose 10 mg/Kg/day are effective against *A. pacificus* infection (Scholz et al., 2009). The tablets are taken at the end of a meal. The drug is active on the parasite calcium channels, leading to an increase of the cellular membrane permeability and the muscular paralysis of the worm. It could inhibit the absorption of the adenosine by the parasite, which could not synthesize

purine de novo. Also, praziquantel is efficient on adult (*Taenia* spp.) and larva (cysticercus) of cestodes, as trematodes (*Schistosoma* spp.) and flukes (except, *Fasciola hepatica*). Praziquantel is well tolerated but should be avoided during pregnancy, breastfeeding and in the first trimester of pregnancy and in children younger than 2 years-old as well as in patients with cirrhosis and hepatic problems (Flisser and Willingham, 2013). Side effects are frequent but transitory and consist in nausea, headaches, dizziness, drowsiness and itch (Chai, 2013). The administration of each drug should be accompanied posteriorly (1-2 h) by a saline purge (Torres, 2013). This prevents the stay of the worm in the intestine avoiding a prolonged action of the enzymes of the host, which contributes to damage and alter it, which can make difficult the search of the scolex in the feces after the treatment, aspect necessary to verify the cure parasitological (Torres, 2013). Moreover, a vitamin B12 supplementation can be initiated in case of depletion. Whatever the treatment, its efficiency has to be controlled one (*D. latus*) or two months (*A. pacificus*) after by stool examination, according pre-patent period of parasites (Table 6).

Treatment of sparganosis is surgical. All the nodular masses should be removed to avoid recurrence if the scolex of larvae is let in tissues. If surgery is not possible, praziquantel (120 mg/Kg/day in three doses in adult and 150 mg/Kg/day in three doses in children) is given for 2 days (Liu et al., 2015). In cerebral sparganosis, chemotherapy has been associated to surgery. Moreover, combined dexamethasone and praziquantel allows a better outcome in patients (Hong et al., 2013). Mebendazole has been given after surgical treatment of subcutaneous sparganosis (Tung et al., 2005). In a case of pulmonary sparganosis, two cure of praziquantel (600 mg/day for 5 days) were efficient (Cheng et al., 2014). In proliferative sparganosis, mebendazole and praziquantel (40 mg/Kg/day three times for 2 weeks) did not show efficiency (Moulinier, 1982). The efficiency of the treatment and the absence of recurrence has to be controlled by serology. Antigen or antibody titers should decrease then disappear in case of complete recovery.

Most of the cases of human infection by *Sp. proliferum* are lethal due to the widespread dissemination and their continuous branching of asexually multiplying plerocercoids. Attempts of surgical removal are usually unsuccessful (Kuchta et al., 2015a).

1.4.2 Hygiene measures

Hygiene measures aim to avoid human contamination. It will be difficult to stop the life cycle of the parasites as species of Diphylobothriidae are widely present in wildlife.

Treatment of people and domestic animals, principally dogs, known to be infected is a first step in prevention as it would decrease contamination of water.

As people could be asymptomatic wearers, the use of sanitary facilities and sewage treatment plants would reduce water contamination too. The impact of water treatment is limited because excreta from wild and

domestic mammals (infected with *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, or *Spirometra* spp.) and birds (infected with *D. dendriticus*) are important source of aquatic environment contamination and are difficult to control.

Eating well-cooked fish prevent infection. Some authors recommend cooking fish at least for ≥ 10 min at 55°C kills the plerocercoid larvae (Dick et al., 2001). European Food safety Authority (EFSA, 2011) for parasites other than trematodes specifies a heat treatment with a core temperature of 60°C or more for 1 min. Reaching such a core temperature depends on the thickness and composition of the product, in fillets that a 3 cm thick should be heated for 10 min (EFSA, 2011). The U.S. Food and Drug Administration (FDA, 2011) guidelines for cooking fish should suffice to inactivate plerocercoids. The guidelines for fish are as follows: cook the fish to an internal temperature of 145°F (62.7°C) for 15 s; to 155°F (68.3°C) for comminuted fish, such as fish cakes, and 165°F (73.8°C) for stuffed fish.

Freezing fish before consumption prevent the infection, too. Some authors considered freezing at least at -10°C during 24 h kills the plerocercoid larvae (Raether and Hanel, 2003). EFSA (2011) recommends specifically for *Dibothriocephalus* plerocercoids the application of -18°C in all parts of the product for at least 24 h. Also, EFSA considered for different zoonotic parasites other than trematodes, the freezing treatment must lower the temperature in all part of the product to either -20°C or lower for not less than 24 h, or to -35°C or lower for not less than 15 h. FDA (2011) recommend the freezing and storing at an ambient temperature of -20°C or below for 7 days (total time); or freezing at an ambient temperature of -35°C or below until solid and storing at an ambient temperature of -35°C or below for 15 h; or freezing at an ambient temperature of -35°C or below until solid and storing at an ambient temperature of -20°C or below for 24 h. In some countries (e.g. France and Switzerland), the legislation requires that any fish intended to be consumed raw must be frozen at a temperature lower than -20°C , during at least 24 h. These standards have the advantage of also protecting from others zoonosis transmitted by fish, such as anisakidosis. In general, these measures are not done by individual consumers: In practice, fish have been conserved in the usual freezers during one week.

Salting fish reduce plerocercoid larvae infectivity. Fish should be in 12% NaCl (Raether and Hanel, 2003). The time requires killing the larvae, from several days to weeks, depend on the thickness of the fish and the volume of salt. Smoking and pickling are not effective against the plerocercoids larvae. Information campaigns for the consumers could be useful as they must make them aware with the dangers of raw fish consumption.

The plerocercoids of *D. latus* survive in the tissues of dead fish for up to 10 days in river water (Feachem et al., 1983). The *D. latus* plerocercoids survive 1 to 7 days, 72 h, and 10 min at -10°C , 4°C and 50°C , respectively (Peduzzi and Boucher-Rodoni, 2001). Plerocercoids in fish of 9 Kg can be effectively killed at -6°C for 7 days, in fish of 2 Kg in

6 days at -6°C , and in those of 0.7 kg at -6°C in 3 days. At -18°C the larvae die in 4 or 2 days in fish of 2 Kg or 0.5 Kg, respectively (Feachem et al., 1983).

Only in 8 cases the adult stage of *Spirometra* has been reported in humans (Lee et al. 1984) in contrast to the 1.685 cases of sparganosis (Liu et al., 2015). By this, treatment of infected people will have no impact in preventing transmission as human is an impasse. In contrary, treatment of the farm definitive mammal hosts (cats and dogs) would decrease water contamination (Liu et al. 2015). In the cities of Yunfu and Zhaoqing the prevalence of infection by *S. erinaceieuropaei* was of 47.2% and 12.6%, respectively in dogs. Infection in the cats was of 64.4% and 35.9% in Yunfu and Zhaoqing, respectively (Hong et al., 2016). Treatment of wild definitive mammal hosts (others canids and felids) is difficult.

The use of drinking water or the filtration of non-drinking water prevents infection; most sparganosis cases reported from Thailand it is by the consumption of drinking contaminated water with infected copepods (Wiwanitkit, 2005; Anantaphruti et al., 2011). Installation of water fountains or areas with open water for public use should be encouraged in regions of endemicity (Nithiuthai et al., 2004).

Eating well-cooked frogs or aquatic snakes prevent the infection (Anantaphruti et al. 2011; Liu et al. 2015). Hong et al (2016) determined through a survey that the consumption of cooking meat with frying and/or porridge and/or soup would be the most probable means of transmission of sparganosis by frogs in Guangzhou, China. These authors found prevalences of 51.2% and 35.1% in wild *R. tigrina rugulosa* and *R. limnocharis*, however, sparganosis was not found in farmed frogs. Cooking with 55°C during at least 5 min kills the plerocercoid larvae (Dick, 2008). There is no appropriate data on the effect of freezing on amphibians or reptiles meat except an Australian regulation for the export of fish or their products indicating for crocodiles exposed or suspected of having sparganosis to place, after processing, at -12°C or cooler at the thermal center for a minimum of 5 days (Magnino et al 2009). Public regulation of hunting or sale of wild animals, particularly frogs and snakes, could prevent human infection (Li et al., 2011; Liu et al., 2015).

Avoiding traditional medical practices, using poultices made of frogs or snakes on skin or eye, prevent infection (Anantaphruti et al., 2011; Li et al., 2011; Liu et al., 2015). Information campaigns for people in endemic countries on the dangers of raw frogs or snakes consumption or use as poultices and of non-drinking water would help to the prevention and health care of sparganosis.

2.0 Environmental Occurrence and Persistence

2.1 Detection Methods

Detecting copepods infection by microscopic examination may be performed by directly observing the proceroids in hemocele (Fig. 11), by extraction by mild

compression between slide and cover slips (Guttowa, 1963; Torres et al., 2004b, 2007) or dissection, when the wall thickness makes it difficult to observe the larvae (Guttowa, 1963; Sharp et al., 1990).

The infection in copepods has been determined in their aquatic environments (Guttowa, 1963; Johnson, 1975) (Table 7) and its detection depends in part, on the sampling system included seasonal period, collection sites and the development status and sex of the host (Guttowa, 1965; Chubb, 1980). Additionally, morphological descriptions of

procercoids have not been evaluated criteria for specific differentiation. The procedure most often used to determine the susceptibility of copepods has been through its experimental infection with coracidia obtained by in vitro incubation of eggs of *D. latus*, *D. dendriticus*, *D. nihonkaiensis*, *Di. balaenopterae*, and *Spirometra* spp. (Janicki and Rosen, 1917; Eguchi, 1926; Essex and Magath, 1931; Bearup, 1957; Guttowa, 1961; Halvorsen, 1966; Kamo et al., 1973; Venturini, 1989; Lee et al., 1990; Denegri and Reisin, 1993; Wright and Curtis, 2000; Torres et al., 2007; Lin et al., 2010).

Table 7. Natural infection in copepods by *Dibothriocephalus* spp.

Country	Parasite	Host	Period	Sector A Prevalence % (# of Samples)	Sector B Prevalence % (# of Samples)	Reference
Finland, Lake Karpero	<i>D. latus</i>	<i>Thermocyclops oithonoides</i>	June	23.3% 7/30	20.6% 6/29	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Thermocyclops oithonoides</i>	June	18.5% 5/27	17.6% 3/17	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Thermocyclops oithonoides</i>	July	12.5% 1/8	22.2% 2/9	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Thermocyclops oithonoides</i>	July	12.5% 2/17	14.2% 2/14	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Cyclops strenuus</i>	June	51.7% 43/83	52.3% 11/21	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Cyclops strenuus</i>	June	51.5% 16/31	37.5% 12/32	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Cyclops strenuus</i>	July	33.3% 3/9	27.2% 3/11	Guttowa, 1963
Finland, Lake Karpero	<i>D. latus</i>	<i>Cyclops strenuus</i>	July	18.0% 2/11	0 0/2	Guttowa, 1963
USA, in waters near Cobb Fish Hatchery, Enfield, Maine	<i>D. dendriticus</i>	<i>Cyclops scutifer</i>	Mid-July	1.7% 1/200	NR ^a	Johnson, 1975

^aNR: Not Reported

The plerocercoids measure 1 mm to several cm long and their detection on the surface of muscles and in the viscera is feasible by naked eye observation (Figs. 17, 19). However those of small size or located in the thickness of muscles or organs must be detected using a

stereomicroscope. The presence of plerocercoids in muscles is of primary importance in human transmission of infections. Its detection is performed by visual inspection, slicing, candling, or pressing (EFSA, 2011; FDA, 2011). The pressing technique in plaques has proved more efficiently

than candling in the detection of *D. latus* and *D. dendriticus* (Torres and Puga, 2011). Digestion is more expensive in terms of reagents than candling and glass compression techniques. Plerocercoids of *D. latus* and *D. dendriticus* in *O. mykiss* are more abundant in the ventral than dorsal musculature (Torres and Puga, 2011).

2.2 Data on Occurrence

2.2.1 Occurrence in the environment

Diphyllbothriideans eggs are sensitive to environmental conditions and to develop must achieve adequate water courses such as rivers or lakes where meet the biotic and abiotic characteristics required for their life cycle. As the temperature rises from 8 to 30°C, the development of *D. latus* coracidia increases in fresh water; at 8-10°C in 39-42 days, at 15°C in 18-20 days, at 18-20°C in 8-9 days, at 25°C in 7 days, and at 30°C in 6 days. However, hatching rate decreases (Guttowa, 1961). The eggs remain viable for years at temperatures near 0°C; but they can do not survive at 35°C (Guttowa, 1961). Eggs in water depths greater than 20 m do not hatch and those that found in feces contaminating soil or ice die by desiccation after 3 days in winter (Feachem et al., 1983). *Dibothriocephalus latus* eggs remain viable in soil for 2 months and in water for 5 months (Khromenkova et al., 1995). The oxygen concentration can influence their development through their aerobic metabolism. The best biotopes are shallow littorals with water temperatures of 15-20°C, with vegetation favoring the development of copepods and fish (von Bonsdorff and Bylund, 1982). The minimum concentration of oxygen in the water for the hatching of coracidium of *D. latus* is 1.4 mg/L at 24°C. The eggs can survive at low concentrations of oxygen for months and develop coracidia when the optimum concentration is reestablished (von Bonsdorff, 1977).

Coracidia (Fig. 10) can survive for 72 h at 8-10°C, 48-72 h at 15°C, 48 h at 18-20°C, 24-36 h at 25°C, and 12 to 24 h at 30°C, respectively (Guttowa, 1961). The larvae survive 4 days in 0.4 0/00 saline (von Bonsdorff and Bylund, 1982) and could tolerate the brackish water of the coastal areas, estuaries and innermost bays where the salinity is 0.2-0.4 0/00 (Bonsdorff and Bylund, 1982). The capacity to develop coracidia from *D. latus* eggs from human infection reaches between 74.3% and 95% (Essex and Magath, 1931; Guttowa, 1959). In dog infections that capacity reaches 45% to 52% (Tarassow, 1934; Guttowa, 1959).

Hatching coracidia of *D. dendriticus* live 12-72 h at room temperature in fresh water (Sharp et al., 1990). At 10°C the eggs develop coracidia in 25 days reaching their peak at 41 days, with full development of the population of eggs at about 2.5 months; at 29 days the eggs start to hatch with a peak at 65 days (Wright and Curtis, 2000). These same authors obtained larvae at 5 days at 20°C with a peak at 9 days with the full development of the population of eggs taking 25 days. Coracidia may develop slowly at 4°C, hatching between 11 and 30 months, favoring the life cycle in those lakes that range from 8 to 14°C during the ice free period in northern North America and Northern Europe as

well as in Arctic Lakes with maximum temperatures not exceeding 4°C (Wright and Curtis, 2000). *Dibothriocephalus dendriticus* eggs die within 6-8 days when kept at 37°C (Kuhlow, 1953). Their eggs are developed at 7°C in 48 days, at 20 °C in 10 to 11 days, and at 23°C in 8 days, respectively (Chubb, 1980).

Dibothriocephalus nihonkaiensis develops experimentally in a freshwater copepod (Eguchi, 1926). *Adenocephalus pacificus* coracidia hatch in sea water after incubation of the eggs at 22°C for 3 days (Escalante et al., 1987).

Spirometra mansonoides eggs die when exposed to 37°C for 6 to 8 days, but at 4°C they can survive for 5 years, although their viability gradually decreases (Mueller, 1961, 1974). Eggs develops experimentally coracidia that hatch after 7 days at 25°C and infecting proceroids develop between 11 and 12 days at 20-25°C in *Paracyclops fimbriatus*, reaching a mean of 210 µm long by 120 µm wide (Venturini, 1989; Denegri and Reisin, 1993). The coracidia develop in 10 days at 25 to 27°C (Mueller, 1959) or in 10 to 14 days at 23°C (Mueller, 1974). In *Cyclops vernalis*, infective proceroids develop between 9 and 14 days at 24 to 27°C (Mueller, 1966).

In areas of the Kama reservoir in Russia where raw sewage is eliminated untreated the infection levels of *D. latus* in fish are fairly high, especially in areas where chemical residues are also eliminated in water (Artamoshin and Khodakova, 1976). The wastewater effluent from the town of Abakan in Russia contained 0.6-4.1 *D. latus* eggs/L and effluent from a septic tank of a hospital contained 52.1 eggs /L. The ground around the septic tank contained 1,830 eggs /Kg and in the areas surrounding the fishing sites, 95% of *E. lucius* was infected (Gerasimov, 1987). Eggs of *D. latus* were found in 5% of fresh vegetables in the markets of Nasiriyah and Suq-AL-Shuyukh, Iraq (Al-Mozan et al., 2015). Additionally, they were reported as an indicator on the use of untreated sewage in the watering and fertilizing of farm land (Al-Mozan et al., 2015). Also 3.2% of lettuces acquired in supermarkets in the city of Dourados-MS, Brazil were contaminated with eggs of *Dibothriocephalus* sp. (Correa et al., 2012). Mixed human and farm effluent from three state farms contained 300-1,600 helminth eggs per m³, including *Dibothriocephalus*, *Ascaris*, *Enterobius*, *Trichuris*, and *Taenia* (Arkhipova, 1977). The passage of effluent through a treatment plant only halved the egg count (Arkhipova, 1977). Eggs of diphyllbothriidean were found in most of the samples of sludge collected from night-soil disposal plants in Kochi Prefecture, Japan (Takenouchi et al., 1980). In recreational areas in the lower Don region of Russia, most of the sites had either no sanitation or inadequate facilities resulting in heavy contamination of the soil with ova identified as *Dibothriocephalus* and other helminths where about 43.1% of soil samples contained 17 to 206 ova /Kg (Khromenkova, 1993).

Data on occurrence of eggs of Diphyllbothriidae in the environment do not really reflect the epidemiological situation of the infection. On the opposite, the prevalence of plerocercoids in receptive fish is an indication of the presence of the infection in a given region.

2.2.2 Occurrence in copepods, fish, and others intermediate and paratenic hosts

Most copepod host records correspond to experimental studies in *D. latus* (Table 3) and *D. dendriticus* (Table 5). Studies on occurrence of infection by procercoids in copepods under natural conditions are scarce (Table 7). It has been estimated that for the development of *D. latus* a minimum density of copepods (1,000/m³) is necessary (von Bonsdorff, 1977). In the Don River, Russia, where 8 to 13 human cases of *D. latus* were recorded annually and 0.3% of predatory fish were infected; the densities of 7 copepod host species ranged from 200 to 2,000/m³ in an area where parasite eggs were released into the river with effluent from households, boats and ships (Khromenkova et al., 1995).

In *M. leuckarti* in the Luohe City in China a prevalence of 3.5% by *S. erinaceieuropaei* with an intensity of 3 to 5 procercoids was found (Lin et al., 2010). Nine *Mesocyclops* species have been recorded as hosts of *Spirometra* procercoids in China (Liu et al., 2015). In Korea, *M. leuckarti* and *E. serrulatus* as hosts of *S. erinaceieuropaei*

have been identified (Lee et al., 1990).

Data on the occurrence, including prevalence, mean intensity and/or mean abundance of infection in fish, for *D. latus* and *D. dendriticus* in lakes and rivers of different countries are given for distinct hosts in Table 8. In South America, the highest prevalence and intensity or abundance of infection by *D. latus* are recorded principally in introduced salmonids such as *O. mykiss* and *S. trutta* in Chile and *O. mykiss* and *Salvelinus fontinalis* in Argentina. *Dibothriocephalus latus* and *D. dendriticus* infection has been recorded in native fish from 1989 in 2 of 21 lakes investigated in southern Chile (Torres et al., 1989b, 1990, 1991, 1992, 1998, 2012). Similar observations have been made on Lake Moreno in Argentina (Table 8). The frequency of finding plerocercoids of *D. latus* and *D. dendriticus* ranges from 55.8% to 73.3% and 9.1% to 43% respectively, in the muscles of *O. mykiss* in Lake Panguipulli, Chile (Torres et al., 2004a, 2012; Torres and Puga, 2011). Examination of 200 (Nicolaud et al., 2005) and 960 (Dupouy-Camet et al., 2015) fillets of *P. fluviatilis* from Lake Léman showed an overall prevalence of 7% (4-10%) and 0.9% (0.3-1.5%), respectively.

Table 8. Prevalence, mean intensity and/or mean abundance of infection by *Dibothriocephalus latus* and *D. dendriticus* in different fish hosts and countries

Area	Parasite	Fish Host	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Mean Abundance ^b	Reference
Argentina, Lake Moreno	<i>D. latus</i>	<i>Oncorhynchus mykiss</i>	28.1% (32/114)	NR ^c	1.4	Revenga, 1993
Argentina, Lake Moreno	<i>D. latus</i>	<i>Salvelinus fontinalis</i>	9.1% (1/11)	NR	0.1	Revenga, 1993
Argentina, Lake Moreno	<i>D. latus</i>	<i>Percichthys trucha</i>	18.8% (6/32)	NR	0.3	Revenga, 1993
Argentina, Lake Moreno	<i>D. dendriticus</i>	<i>O. mykiss</i>	57.9% (66/114)	NR	7.2	Revenga, 1993
Argentina, Lake Moreno	<i>D. dendriticus</i>	<i>S. fontinalis</i>	27.3% (3/11)	NR	0.5	Revenga, 1993
Canada, Great Central Lake	<i>D. dendriticus</i>	<i>O. nerka</i>	93.2% (55/59)	4.0	NR	Ching, 1988
Canada, Lake Igalugaajurului	<i>D. dendriticus</i>	<i>S. alpinus</i>	41.8% (117/280)	73.0	NR	Gallagher and Dick, 2010
Chile, Lake Caburgua	<i>D. latus</i>	<i>O. mykiss</i>	3.7% (1/27)	3.0	NR	Torres et al., 1991
Chile, Lake Caburgua	<i>D. dendriticus</i>	<i>O. mykiss</i>	44.4% (12/27)	3.3	NR	Torres et al., 1991
Chile, Lake Calafquén	<i>D. latus</i>	<i>O. mykiss</i>	4.3 % (1/18)	4.0	NR	Torres et al., 1991
Chile, Lake Calafquén	<i>D. dendriticus</i>	<i>O. mykiss</i>	27.8% (5/18)	2.2	NR	Torres et al., 1991
Chile, Lake Colico	<i>D. latus</i>	<i>O. mykiss</i>	28.6% (6/21)	6.8	NR	Torres et al., 1991
Chile, Lake Colico	<i>D. latus</i>	<i>S. trutta</i>	75% (3/4)	34.0	NR	Torres et al., 1991
Chile, Lake Colico	<i>D. dendriticus</i>	<i>O. mykiss</i>	47.6% (10/21)	2.6	NR	Torres et al., 1991

Area	Parasite	Fish Host	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Mean Abundance ^b	Reference
Chile, Lake Colico	<i>D. dendriticus</i>	<i>S. trutta</i>	75% (3/4)	26.7	NR	Torres et al., 1991
Chile, Lake Cucao	<i>D. dendriticus</i>	<i>O. mykiss</i>	5.1% (4/79)	1.1	NR	Rozas et al., 2012
Chile, Lake Huillinco	<i>D. dendriticus</i>	<i>O. mykiss</i>	3.6% (5/138)	1.1	NR	Rozas et al., 2012
Chile, Lake Llanquihue	<i>D. dendriticus</i>	<i>O. mykiss</i>	18.8 % (6/32)	6.2	NR	Torres et al., 1991
Chile, Lake Maihue	<i>D. latus</i>	<i>S. trutta</i>	50.0% (1/2)	37.0	NR	Torres et al., 1991
Chile, Lake Maihue	<i>D. dendriticus</i>	<i>O. mykiss</i>	3.0% (1/33)	1.0	NR	Torres et al., 1991
Chile, Lake Maihue	<i>D. dendriticus</i>	<i>S. trutta</i>	50.0% (1/2)	10.0	NR	Torres et al., 1991
Chile, Lake Natri	<i>D. dendriticus</i>	<i>O. mykiss</i>	4.7% (5/107)	1.1	NR	Rozas et al., 2012
Chile, Lake Panguipulli	<i>D. latus</i>	<i>O. mykiss</i>	78.0 % (32/41)	48.4	NR	Torres et al., 1991
Chile, Lake Panguipulli	<i>D. latus</i>	<i>O. mykiss</i>	83.3% (25/30)	87.1	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. latus</i>	<i>Basilichthys australis</i>	7.1% (1/14)	1.0	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. latus</i>	<i>Odontesthes mauleanum</i>	69.2% (9/13)	4.6	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. latus</i>	<i>Percichthys trucha</i>	52.2 (12/23)	3.3	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. latus</i>	<i>Galaxias maculatus</i>	2.1 (1/47)	1.0	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. latus</i>	<i>O. mykiss</i>	92% (71/77)	33.0	30.4	Torres and Puga, 2011
Chile, Lake Panguipulli	<i>D. latus</i>	<i>O. mykiss</i>	93% (26/28)	35.5	32.9	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. latus</i>	<i>B. australis</i>	10.4% (5/48)	7.2	0.8	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. latus</i>	<i>O. mauleanum</i>	43.2% (19/44)	5.8	2.5	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. latus</i>	<i>P. trutta</i>	44.0 % (11/25)	4.4	1.9	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mykiss</i>	65.9% (27/41)	7.0	NR	Torres et al., 1991
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mykiss</i>	76.7% (23/30)	5.7	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>B. australis</i>	14.3 (2/14)	1.0	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mauleanum</i>	23.1 (3/13)	1.0	NR	Torres et al., 2004a
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mykiss</i>	48.1% (37/77)	5.9	2.8	Torres and Puga, 2011
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mykiss</i>	57.1% (16/28)	5.0	2.9	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>B. australis</i>	4.5 % (2/44)	NR	0.05	Torres et al., 2012
Chile, Lake Panguipulli	<i>D. dendriticus</i>	<i>O. mauleanum</i>	2.1% (1/48)	NR	0.04	Torres et al., 2012
Chile, Lake Puyehue	<i>D. latus</i>	<i>O. mykiss</i>	20% (6/30)	1.2	NR	Torres et al., 1991
Chile, Lake Puyehue	<i>D. dendriticus</i>	<i>O. mykiss</i>	60.0% (18/30)	3.2	NR	Torres et al., 1991

Area	Parasite	Fish Host	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Mean Abundance ^b	Reference
Chile, Lake Ranco	<i>D. latus</i>	<i>O. mykiss</i>	6.7% (2/30)	8.0	NR	Torres et al., 1991
Chile, Lake Ranco	<i>D. dendriticus</i>	<i>O. mykiss</i>	16.7% (5/30)	4.6	NR	Torres et al., 1991
Chile, Lake Riñihue	<i>D. latus</i>	<i>O. mykiss</i>	44.9% (31/69)	9.2	NR	Torres et al., 1991
Chile, Lake Riñihue	<i>D. latus</i>	<i>O. mykiss</i>	35.7% (50/140)	5.2	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. latus</i>	<i>G. maculatus</i>	0.5% (1/184)	1.0	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. latus</i>	<i>P. trucha</i>	11.1% (7/63)	1.3	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. dendriticus</i>	<i>O. mykiss</i>	53.6% (37/69)	11.1	NR	Torres et al., 1991
Chile, Lake Riñihue	<i>D. dendriticus</i>	<i>O. mykiss</i>	17.9 (25/140)	2.4	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. dendriticus</i>	<i>G. maculatus</i>	0.5% (1/184)	1.0	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. dendriticus</i>	<i>B. australis</i>	0.5% (1/192)	1.0	NR	Torres et al., 1998
Chile, Lake Riñihue	<i>D. dendriticus</i>	<i>P. trucha</i>	3.2% (2/63)	2.0	NR	Torres et al., 1998
Chile, Lake San Antonio	<i>D. dendriticus</i>	<i>O. mykiss</i>	66.7% (4/6)	1.1	NR	Rozas et al., 2012
Chile, Lake Villarrica	<i>D. latus</i>	<i>O. mykiss</i>	6.9% (2/29)	1.5	NR	Torres et al., 1991
Chile, Lake Villarrica	<i>D. dendriticus</i>	<i>O. mykiss</i>	44.8% (13/29)	6.1	NR	Torres et al., 1991
Chile, Lake Villarrica	<i>D. dendriticus</i>	<i>S. trutta</i>	33.3% (1/3)	2.0	NR	Torres et al., 1991
Chile, Lake Rupanco	<i>D. dendriticus</i>	<i>O. mykiss</i>	7.4% (2/27)	1.5	NR	Torres et al., 1991
Chile, Lake Tarahuín	<i>D. dendriticus</i>	<i>O. mykiss</i>	50.9% (81/159)	1.1	NR	Rozas et al., 2012
Chile, Lake Todos los Santos	<i>D. dendriticus</i>	<i>O. mykiss</i>	14.3% (2/14)	2.0	NR	Torres et al., 1991
Finland, Lake Inari	<i>D. dendriticus</i>	<i>S. trutta m. lacustris</i>	90% (109/121)	NR	52.9	Rahkonen and Koski, 1997
Finland, Lake Inari	<i>D. dendriticus</i>	<i>S. trutta m. lacustris</i>	83.0 (73/88)	NR	50.2	Rahkonen and Koski, 1997
France, Lake Léman	<i>D. latus</i>	<i>Perca fluviatilis</i>	29.2% (7/24)	NR	NR	Dupouy-Camet et al., 2015
France, Lake Léman	<i>D. latus</i>	<i>Esox lucius</i>	100% (6/6)	NR	NR	Dupouy-Camet et al., 2015
France, Lake Léman	<i>D. latus</i>	<i>Lota lota</i>	28.6% (2/7)	NR	NR	Dupouy-Camet et al., 2015
France, Lake Léman	<i>D. latus</i>	<i>P. fluviatilis</i> (fillets)	0.9% (9/960)	NR	NR	Dupouy-Camet et al., 2015
France, Lake Léman	<i>D. latus</i>	<i>P. fluviatilis</i> (fillets)	7% (14/200)	NR	NR	Nicolaud et al., 2005
Germany, Baltic Sea (coast)	<i>D. dendriticus</i>	<i>S. trutta</i>	2.9% (1/35)	1.0	0.1	Unger and Palm, 2016
Germany, Freshwater streams	<i>D. dendriticus</i>	<i>S. trutta</i>	5.9% (1/17)	4.0	0.2	Unger and Palm, 2016
Italy, Lake Como	<i>D. latus</i>	<i>P. fluviatilis</i>	25.4% (108/426)	1.2	0.31	Gustinelli et al., 2016

Area	Parasite	Fish Host	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Mean Abundance ^b	Reference
Italy, Lake Como	<i>D. latus</i>	<i>E. lucius</i>	84.2% (16/19)	28.3	23.8	Gustinelli et al., 2016
Italy, Lake Como	<i>D. latus</i>	<i>L. lota</i>	3.6% (2/55)	1.5	0.05	Gustinelli et al., 2016
Italy, Lake Iseo	<i>D. latus</i>	<i>P. fluviatilis</i>	7.6% (35/428)	1.3	0.10	Gustinelli et al., 2016
Italy, Lake Iseo	<i>D. latus</i>	<i>E. lucius</i>	71.4% (5/7)	16.4	11.7	Gustinelli et al., 2016
Italy, Lake Iseo	<i>D. latus</i>	<i>L. lota</i>	3.8% (1/26)	3.0	0.12	Gustinelli et al., 2016
Italy, Lake Lario	<i>D. latus</i>	<i>P. fluviatilis</i>	30.0% (609/183)	1.3	NR	Wicht et al., 2009
Italy, Lake Maggiore	<i>D. latus</i>	<i>P. fluviatilis</i>	6.6% (42/635)	1.1	0.07	Gustinelli et al., 2016
Italy, Lake Maggiore	<i>D. latus</i>	<i>E. lucius</i>	100% (1/1)	1.0	1.0	Gustinelli et al., 2016
Italy, Lake Orta	<i>D. latus</i>	<i>P. fluviatilis</i>	33.3% (5/15)	NR	NR	Peduzzi and Boucher-Rodoni, 2001
Italy/Switzerland, Lake Maggiore	<i>D. latus</i>	<i>P. fluviatilis</i>	7.8% (24/309)	NR	NR	Peduzzi and Boucher-Rodoni, 2001
Norway, Lakes Storvatn and Forsanvatn	<i>D. latus</i>	<i>S. trutta</i>	10.2% (22/214)	2.2	0.2	Kuhn et al., 2016
Norway, Lakes Rekvatn and Fjellfrosvatn	<i>D. latus</i>	<i>S. trutta</i>	45.7% (52/113)	5.7	2.6	Kuhn et al., 2016
Norway, Lakes Makkvatn, Skilvatn, Sagelvatn, and Fefjerdevatn	<i>D. latus</i>	<i>S. trutta</i>	45.6% (122/268)	39.4	17.9	Kuhn et al., 2016
Sweden, Lake Bjellojaure	<i>D. dendriticus</i>	<i>Salvelinus alpinus</i>	83.2% (544/654)	8.8	NR	Henricson, 1977
Switzerland, Lake Bienne	<i>D. latus</i>	<i>P. fluviatilis</i>	3.7% (3/81)	NR	NR	Golay and Mariaux, 1995
Switzerland, Lake Bienne	<i>D. latus</i>	<i>E. lucius</i>	14.3% (1/7)	NR	NR	Golay and Mariaux, 1995
Switzerland, Lake Morat	<i>D. latus</i>	<i>P. fluviatilis</i>	5.3% (1/19)	NR	NR	Golay and Mariaux, 1995
Switzerland, Lake Morat	<i>D. latus</i>	<i>E. lucius</i>	12.5% (1/8)	NR	NR	Golay and Mariaux, 1995
United Kingdom (Scotland), Loch Awe	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>O. mykiss</i>	75% (3/4)	5.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Rannoch	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>O. mykiss</i>	42.8% (3/7)	20.6	NR	Dorucu et al., 1995
United Kingdom (Scotland), Lake of Mentheith	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>O. mykiss</i>	33.3% (7/21)	NR	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Lomond	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	25% (8/2)	2.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Lomond	<i>D. dendriticus</i>	<i>Coregonus lavaretus</i>	38.5% (26/10)	NR	NR	Dezfuli et al., 2007

Area	Parasite	Fish Host	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Mean Abundance ^b	Reference
United Kingdom (Scotland), Loch Maragan	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	45.7% (16/35)	27.1	NR	Dorucu et al., 1995
United Kingdom (Scotland), Carbeth Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	10% (2/20)	1.5	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Awe	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	100% (20/20)	23.8	NR	Dorucu et al., 1995
United Kingdom (Scotland), Dunalastair Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	60.0% (9/15)	18.7	NR	Dorucu et al., 1995
United Kingdom (Scotland), Cochno Loch	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	33.3% (1/3)	NR	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Leven	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	33.3% (1/3)	4.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Hill Loch	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	57.0% (4/7)	68.2	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Rannoch	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	57.8% (11/19)	8.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Bruncrooks	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	25% (1/4)	1.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Talla Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	26.6% (4/15)	4.2	NR	Dorucu et al., 1995
United Kingdom (Scotland), Loch Venachar	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	50.0% (1/2)	2.0	NR	Dorucu et al., 1995
United Kingdom (Scotland), Carron Valley Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	13.6% (3/22)	2.3	NR	Dorucu et al., 1995
United Kingdom (Scotland), Whiteader Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	15.7% (3/19)	11.3	NR	Dorucu et al., 1995
United Kingdom (Scotland), Fruid Reservoir	<i>D. dendriticus</i> and <i>D. ditremus</i>	<i>S. trutta</i>	22.2% (2/9)	1.0	NR	Dorucu et al., 1995

^aMean of parasites by infected fish; ^bMean of parasites by examined fish; ^cNot Reported

Table 9 show the prevalence (3.5-23.5%) and mean intensity (1-3.3 plerocercoids/infected fish) of infection with

A. pacificus in the coast of Peru. Plerocercoids have not been found in the muscles of different marine fish species.

Table 9. Prevalence and mean intensity of infection by *Adenocephalus pacificus* in some fish from the coast of Peru

Fish	Prevalence Percentage (# of Samples)	Mean Intensity ^a	Reference
<i>Cilus gilberti</i>	23.3 (24/103)	3.3	Chero et al., 2014c
<i>Coryphaena hippurus</i>	11.7 (33/282)	1.1	Escalante et al., 1988
<i>Cynoscion analis</i>	4.7 (2/43)	1.0	Escalante, 1983
<i>Ariopsis seemanni</i>	12.5 (4/32)	1.5	Escalante and Miranda, 1986
<i>Genypterus maculatus</i>	16.7 (1/6)	1.0	Escalante, 1983
<i>Sciaena deliciosa</i>	6.6 (25/381)	NR ^b	Tantalean, 1975
<i>Sciaena deliciosa</i>	14.3 (16/112)	1.1	Escalante and Miranda, 1986
<i>Sciaena deliciosa</i>	17.1 (6/35)	1.3	Chero et al., 2014a
<i>Sciaena callaensis</i>	23.5 (4/17)	1.8	Escalante, 1983
<i>Trachinotus paitensis</i>	75.0 (12/16)	1.5	Escalante and Miranda, 1986
<i>Trachurus symmetricus murphy</i>	5.7 (4/70)	1.0	Escalante et al., 1988
<i>Paralabrax humeralis</i>	3.5 (369/13)	1.4	Ianaconne and Alvarino, 2009
<i>Merluccius gayi peruanus</i>	6.3 (2/32)	1.0	Escalante, 1983
<i>Merluccius gayi peruanus</i>	4.8 (3/62)	1.0	Chero et al., 2014b
<i>Merluccius gayi peruanus</i>	12.9 (21/163)	1.3	Escalante et al., 1988
<i>Paralichthys adspersus</i>	7.4 2/27	1.0	Escalante and Miranda, 1986

^aMean of parasites by infected fish; ^bNot Reported

In Japan, *Oncorhynchus keta*, *O. masou* and *O. gorboscha* presented prevalence of 51.1%, 10.8% and 18.5% and mean intensities of 3.9, 3.7, and 1 plerocercoids/infected host with *D. nihonkaiensis*, respectively (Suzuki et al., 2010). In Hokkaido, juvenile of *O. masou* do not show any signs of infection before migrating to the sea. However, adults return with prevalence up to 50%, suggesting that these fish could acquire the infection during their migration to the ocean (Awakura et al., 1985; Suzuki et al., 2010; Yanagida et al., 2010).

Fish infection can have economic impact in aquaculture and in the commercialization, export and import of fishes. The presence of parasites in feral or farmed fish would be a high risk for human health, but this risk could be more significant if the plerocercoids were found in the muscle of the fish (Torres et al., 2010, 2012). This last and the higher prevalence and intensity of infection by species of diphyllbothriid of major importance in health are more frequently in feral fish (Torres and Puga, 2011; Torres et al., 2002, Torres et al., 2004a, 2010, 2012). Extensive trade of fresh or chilled fish has led to the description of exotic infections in human living in non-endemic countries (Pastor-Valle et al., 2014) or in endemic countries by non-autochthonous species (Yera et al., 2006; Wicht, 2007; Wicht et al., 2008b; Paugam et al., 2009). It results an impact on the epidemiology of diphyllbothriosis, with a risk of dissemination of the parasitosis (Kuchta et al., 2014).

Dibothriocephalus dendriticus and *D. ditremus*, both lives as an adult in piscivorous birds and have been identified in farmed *O. mykiss* in Scotland with prevalence of 1.5% to 11.7% of the hatch trout from 3 raceways (Wootten and Smith, 1979). In a fish farm on the Lake Särkijärvi in Finland, *D. dendriticus* with prevalence of 2-86% and 1-83% and mean intensity of 1-1.5 and 3.1 plerocercoids in *S. trutta m. lacustris* and *S. trutta m. trutta* were identified, respectively (Rahkonen et al., 1996). *Dibothriocephalus ditremus* was found in visceral organs, musculature and pericardial cavity in stocks of *S. salar* reared in freshwater cage systems in lakes of Ireland (Rodger, 1991). Prevalence of 4.8% to 6.6% and mean intensity with 1 to 3 plerocercoids of *D. dendriticus* was reported in viscera of *O. mykiss* cultured in freshwater cages from 2 farms in the Kola Peninsula, Russia (Karasev et al., 1997). Plerocercoids of *D. dendriticus* caused an epizootic in 10-18 cm long hatchery-reared *O. mykiss* in Southern Newfoundland, Canada; causing 7,500 fish died (Khan, 2009). *Dibothriocephalus dendriticus* was found in viscera of *O. mykiss* with a prevalence of 6.7% and mean intensity of 1 plerocercoid in a freshwater farm at Lake Tarahuin on Chiloe Island, Chile (Torres et al., 2010). Also, plerocercoids identified as *Dibothriocephalus sp.* was reported in a trout, *O. mykiss*, cultivated in the Comau fiord, Chile (Torres et al., 2002).

In China has been reported prevalence of 2.9-91.2% and mean intensities of 1-8.6 plerocercoids/infected hosts in the frogs *Rana tigrina*, *R. limnocharis*, *R. nigromaculata*, *R. temporaria chensinensis*, *R. plancy*, *R. guentheri*, *R. cancrivora*, *Microhyla ornate*, *Hyla arborea*, and *Paa*

yunnanensis (Liu et al., 2015). Also, the authors indicate prevalence of 1.3-100% and mean intensities of 1.8-77.1 larvae/ infected host in the snakes *Elaphe plumbea*, *E. carinata*, *E. taeniura*, *E. radiata*, *E. mandarinus*, *E. carinata*, *Natrix tigrini lateralis*, *Naja naja atra*, *Zaocys dhumnades*, *Dinodon rufozonatum*, *Ptyas korros*, *P. mucosux*, *Bungrarus multicinctus*, *B. albolabris*, *Deinagkistrodon acutus*, and *Xenochrophis piscator*. In Malaysia is reported *R. limnocharis* and *R. cancrivora* with prevalences of 8.8%-21.5%, respectively, and in Australia *Litoria caerulea* with 4.9% of prevalence (Liu et al., 2015). The occurrence of plerocercoids in lizards and snakes is most likely due the ingestion of infected anurans than by acquisition by infected copepods with proceroids (Oda et al., 2016).

In South America, *Leptodactylus latrans* (syn. *Letodactylus ocellatus*) in Uruguay showed 48.6% of prevalence with intensities of up to 22 plerocercoids (Dei-Cas et al., 1976). Tadpoles of *Rhinella arenarum* (syn. *Bufo arenarum*) in experimental infections in Argentina presented intensities of up to 14 plerocercoids (Venturini, 1989) and in *Pseudopaludicola falcipes* and *Odontophrynus americanus* maximum intensities were of 36 and 7 plerocercoids, respectively (Denegri and Reisin, 1993). In Louisiana, *L. catesbeianus* and *L. clamitans* showed prevalence of 1.7% and 4.9%, respectively; also, in 7 species of reptiles, including 4 species of *Natrix* plus *T. proximus proximus*, *L. getula holbrookii* and *C. constrictor flaviventris* were reported prevalence of 4.8% to 72.7%; in mammals as *D. virginiana* and *P. lotor* prevalence was of 60% and 44.8%, respectively; infection was also present in a specimen of *U. cinereoargenteus* (Corkum, 1966).

Also, sparganosis can have economic impact in high endemic areas related to the induced disabilities.

2.3 Persistence (Survival)

2.3.1 Methods for assessing survival

The viability of eggs of *Dibothriocephalus*, *Diphyllbothrium*, *Adenocephalus*, and *Spirometra* can be measured by their incubation at optimum temperatures, to obtain the stage of coracidium. The survival of the infective plerocercoids is essentially determined by their capacity of movement; however, their infectivity capacity can be assessed in some cases by experimental infection in laboratory animals. For example, *A. pacificus* plerocercoids live for 8 to 28 h in ceviche juice, however its infectivity in dogs is limited to 2 h in that liquid (Escalante and Chico-Ruiz, 1988). The criteria recommended by the ESFA (2011) and FDA (2011) European Food Safety Authority or the U.S. Food and Drug Administration for killing plerocercoid stages in fish to prevent human infection are presented in section 1.4.2: Hygiene measures.

3.0 Reducing Environmental Contamination

3.1 Sanitation Technologies

Data on reductions of *Dibothriocephalus latus* by sanitation management are very scarce and some those

given here correspond to von Bonsdorff's book "Diphylobothriasis in man" (von Bonsdorff, 1977).

The elimination of eggs from waste waters is essential to avoid the spread of infection to aquatic environments in endemic regions. The eggs are sensitive to environmental conditions and die at temperatures below 5°C in the case of *D. latus* (von Bonsdorff and Bylund, 1982). The eggs of *D. dendriticus* also can develop slightly at 4°C (Wright and Curtis, 2000). Eggs isolated from faeces die at 10 min in a solution of 1.8 mg Cl/L but in stools 10 h in 2.5-4.0% solution of chloride of lime is required, although this treatment does not ensure absolute safety (von Bonsdorff, 1977). Also, boiling water is effective to kill *D. latus* in feces eggs in containers using ratio of 3:1 of water to feces (von Bonsdorff, 1977). Different chemical agents are effective in killing the eggs of *D. latus* as example alkali, acids, chloroform, ether, alcohol, formaldehyde (von Bonsdorff, 1977, von Bonsdorff and Bylund, 1982).

Dibothriocephalus eggs die in garbage piles where they are exposed to putrefaction and are sensitive to dehydration and will lose their viability in excrements deposited in dry and well aerated latrines (von Bonsdorff, 1977).

Very efficient purification plants are required to eliminate eggs of *Dibothriocephalus*. Carefully constructed and properly dimensioned purification plants of "three step type" with biological and chemical purification are capable of retaining tapeworm eggs up to 95%. Overloading reduces the purifying effect. Adding filtering through a sand filter or, better, a membrane will give a 100% purifying effect which is certainly obtained by the large plants (von Bonsdorff, 1977). The efficacy of small sewage plants in residential institutions in Russia in decontaminating waste water from helminth eggs, including *D. latus*, concluded that mechanical processing was 76.4% to 89.4% effective, and that surface filtration beds were fully effective but that the efficacy of drip bio filters was poor (Talapa, 1978).

In Kaliningrad, Russia, the sewage used to irrigate a market garden co-operative following 24 h of mechanical processing, contained on arrival at the irrigation field an average of 8.6 helminth eggs/L, mainly *Dibothriocephalus*, *Ascaris* and *Trichuris*. The soil of areas under overhead irrigation was found to be heavily contaminated but plots where underground irrigation was used were free from ova (Romanenko et al., 1976).

Removing eggs of *D. latus* in waste water has been made possible by flocculation with aluminum sulfate at 90 to 120 g/m³, at least by 60 min (Doschi, 1972).

From several gums, hydroxypropyl triammonium chloride guar gum (HPTAC-guar) was selected as the most adequate coagulant-flocculant for the class of municipal wastewater obtaining a diminution of 99% of helminth eggs of *D. latus*, *Ascaris lumbricoides*, *H. nana* and *Toxocara canis* (Zamudio-Perez et al., 2013). Furthermore, the reduction of chemical oxygen demand (COD) and turbidity removals was 46% and 39%, respectively (Zamudio-Perez et

al., 2013). These same authors obtained removals of 100% for helminth eggs using the natural gum mixed with Ca (OH)₂, and 47% for COD and 30% for turbidity.

If reduction of *Dibothriocephalus* eggs in water could be efficient in limiting transmission of the parasitosis, reduction of the first intermediate host in water would give the same result on both dibothriocephalosis and sparganosis. *Cyclops* spp. in the raw water could be completely removed by synergistic effect of chlorine dioxide pre-oxidation followed by coagulation process at chlorine dioxide dosage of 0.9 mg/L. The sort and amount of organic substance in the treated water by chlorine dioxide pre-oxidation were evidently less than that of pre-chlorination and the mutagenicity of drinking water treated by pre-dosing chlorine dioxide was substantially reduced compared with prechlorination (Lin et al., 2007).

It is proven that the quality of the sludge mixed with grass clippings at a ratio of 6:1 volume/volume after having passed a windrow composting process for 8 weeks can be classified as class A biosolids as the levels of remaining fecal coliforms were <3 most probable number per g dry solid and all human parasites were destroyed (Sreesai et al., 2013).

Dibothriocephalus latus eggs may also be rendered innocuous by treatment with chlorine. A 2.5-4% solution of chloride of lime kills the eggs in feces within 10 h (von Bonsdorff, 1977)

Cyclops spp. of zooplankton propagated excessively in eutrophic water body and could not be effectively inactivated by the conventional disinfections process like chlorination due to its stronger resistance to oxidation (Lin et al., 2007). It was found that chlorine dioxide possessed better inactivation effect than chlorine. *Cyclops* can be completely inactivated after 30 min of contact time by low dosage of chlorine dioxide (1.0 mg/L) (Zhao et al., 2007). The size of *Cyclops* and its life activity are important influencing factors by oxidants (Zhang et al., 2009).

Up to 56% of the eggs of *D. latus* were inactivated at 222 nm of water surface radiation dose (up to 5 mJ/cm²), but the eggs of *Opisthorchis felineus* were only inactivated up to 85% (Lipatov et al., 2016). These differences are attributed to some features of eggs shells and showed the importance of considering the morphological and chemical composition of the eggs that can vary between groups or species of helminth parasites, when the criteria of water treatment is evaluated (Lipatov et al., 2016).

Data on control of dibothriocephalosis have focused on the treatment of wastewater containing parasite egg disseminated by the definitive host through their feces. Nevertheless, simple individual or collective means could be efficient in reducing the number of parasites in water. Elimination of parasites after medical and veterinary treatment should be burned, incinerated or buried in the soil.

Traffic on lakes has also been an efficient way to transmit eggs. Nowadays, the situation is under control as

most boats are compelled to keep their black waters on board but this will not apply on small unroofed boats (Khromenkova, 1995). Sanitary treatment of fish for the control of the source of human infection is revised in Part I, Section 1.4.4 Hygiene measures).

It seems that prevalence of human dibothriocephalosis and sparganosis over time depended of human habits: how foods were stored or consumed, if drinking water and latrines were used (Le Bailly et al., 2007; Piers, 2015).

References

- EFSA (2011). Scientific opinion on assessment of epidemiological data in relation to the health risks resulting from the presence of parasites in wild caught fish from fishing grounds in the Baltic Sea. EFSA J.. 9(7), pp. 2320.
- FDA (2011). Food and Drugs Administration. Guidance on controlling parasite hazards for seafood processors: FDA fish and fishery products hazards and controls guidance-Fourth edition. Chapter 5: Parasites.
- WHO (1979). Parasitic zoonoses. Report of a WHO expert committee with the participation of FAO. World Health Organization technical report series. pp. 1-107.
- Acha, PN and Szifres, B (2003). Zoonosis y enfermedades transmisibles comunes al hombre y a los animales. Washington: Organización Panamericana de la Salud
- Al-Mozan, HDK, Mankhi, AMG, Khundhair, IH, Al-Rukabi, N, Naser, AMH and Mousa, HM (2015). Epidemiological of parasites that contaminated fresh vegetables. World Journal of Pharmaceutical Research. 4, pp. 349-359.
- Alkhalife, IS, Hassan, RR, Abdel-Hameed, AA and Al-Khayal, LA (2006). Diphyllbothriasis in Saudi Arabia. Saudi Med J. 27, pp. 1901-1904.
- Almeida, GGuilherme, Coscarelli, D, Melo, MNorma, Melo, ALane and Pinto, HALves (2016). Molecular identification of *Spirometra* spp. (Cestoda: Diphyllbothriidae) in some wild animals from Brazil.. Parasitology international. 65, pp. 428-431. doi: 10.1016/j.parint.2016.05.014.
- Ament, CC and Young, LH (2006). Ocular manifestations of helminthic infections: onchocerciasis, cysticercosis, toxocariasis, and diffuse unilateral subacute neuroretinitis. Int Ophthalmol Clin. 46, pp. 1-10.
- An, YC, Sung, CC, Wang, CC, HC, L, Chen, KY, Ku, FM et al. (2017). Molecular identification of *Diphyllbothrium latum* from a pediatric case in Taiwan. Korean Journal of Parasitology. 55, pp. 425-428.
- Anantaphruti, M, Nawa, Y and Vanvanitchai, Y (2011). Human sparganosis in Thailand: An overview. Acta Trop. 118, pp. 171-176. doi: 10.1016/j.actatropica.2011.03.011.
- Andersen, K (1975). Studies of the helminth fauna of Norway. XXXIV. The morphological stability of *Diphyllbothrium* Cobbold. A comparison of adult *D. dendriticum* (Nitzsch), *D. latum* (L.) and *D. ditremum* (Creplin) development in different hosts. Nor J Zool. 23, pp. 45-53.
- Andersen, K (1971). Studies of the helminth fauna of Norway XVII: Morphological comparison of *Dipyllobothrium dendriticum* Nitzsch 1824, *D. norvegicum* Vik 1957 and *D. latum* (Linne 1758) (Cestoda: Pseudophyllidea). Norwegian Journal of Zoology. 19, pp. 21-36.
- Andersen, K, Ching, HL and Vik, R (1987). A review of freshwater species of *Diphyllbothrium* with redescrptions and the distribution of *D. dendriticum* (Nitzsch, 1824) and *D. ditremum* (Creplin, 1825) development in different hosts.. Can J Zool. 65, pp. 2216-2228. doi: 10.1139/z87-336.
- Andersen, K and Halvorsen, O (1978). Egg size and form as taxonomic criteria in *Diphyllbothrium* (Cestoda, Pseudophyllidea). Parasitol. 76, pp. 229-240.
- Andersen, KI and Gibson, DI (1989). A key to three species of larval *Diphyllbothrium* Cobbold, 1858 (Cestoda: Pseudophyllidea) occurring in European and North American freshwater fishes. Systematic Parasitology. 13, pp. 3-9.
- Ando, K, Ishikura, K, Nakakugi, T, Shimono, Y, Tamai, T, Sugawa, M et al. (2001). Five cases of *Diphyllbothrium nihonkaiense* infection with discovery of plerocercoids from an infective source, *Oncorhynchus masou ishikawae*. J Parasitol. 87, pp. 96-100. doi: 10.1645/0022-3395(2001)087[0096:FCODNI]2.0.CO;2.
- Arizono, N, Shedko, M, Yamada, M, Uchikawa, R, Tegoshi, T, Takeda, T et al. (2009). Mitochondrial DNA divergence in

populations of the tapeworm *Diphylobothrium nihonkaiense* and its phylogenetic relationship with *Diphylobothrium klebanovskii*. *Parasitology International*. 58, pp. 22-28.

Arizono, N, Fukumoto, S, Tademoto, S, Yamada, M, Uchikawa, R, Tegoshi, T et al. (2008). Diplogonoporiasis in Japan: Genetic analysis of five clinical isolates. *Parasitology International*. 57, pp. 212-216.

Arizono, N, Yamada, M, Fukumi, N-U and Ohnishi, K (2009). *Diphylobothriasis* Associated with Eating Raw Pacific Salmon. *Emerging Infectious Diseases*. 15, pp. 866-870. doi: 10.3201/eid1506.090132.

Arkhipova, NS (1977). Role of parasite irrigation with sewage effluent in the epizootiology of cattle cysticercosis. *Fyulleten Vsesoyuznogo Instituta Gel'mintologii*. 21, pp. 10-16.

Artamoshin, AS and Khodakova, VI (1976). The effect of sewage on the rate of infection of yearling fish by *Diphylobothrium latum* plerocercoids in the Kama reservoir. *Gidrobiologicheskii Zhurnal*. 12, pp. 89-91.

Awakura, T, Sakaguchi, S and Hara, T (1985). Studies on parasites of masu salmon, *Oncorhynchus masou*-observations on the seasonal occurrence of *Diphylobothrium latum* plerocercoid. *Scientific reports of the Hokkaido Fish Hatchery*. 40, pp. 57-67.

Baer, JG, Miranda, H, Fernandez, W and Medina, J (1967). Human diphylobothriasis in Peru. *Zeitschrift Parasiten*. 28, pp. 277-289. doi: 10.1007/BF00260267.

Bearup, AJ (1957). Experimental vectors of the first larval stage of *Dibothriocephalus latus* (Cestoda) in Australia. *The Australian Journal Experimental Biology and Medical Science*. 35, pp. 187-192.

Beaver, PC and Rolon, FA (1981). Proliferating larval cestode in a man in Paraguay: a case report and review. *American Journal of Tropical Medicine and Hygiene*. 30, pp. 625-637.

Bo, G and Xuejian, W (2006). Neuroimaging and pathological findings in a child with cerebral sparganosis: Case report. *J Neurosurg*. 105, pp. 470-472.

Botterel, F and Bouree, P (2003). Ocular sparganosis: a case report. *J Travel Med*. 10, pp. 245-246.

Bouree, P, Dahane, N, Resende, P, Bisaro, F and Ensaf, A (2012). Diagnostic des cestodes adultes. *Rev Fr Labo*. 440, pp. 67-73.

Bowman, DD, M, HCharles, S, LDavid and Barr, SC (2002). *Feline Clinical Parasitology*. Iowa, USApp. 469 PP.

Bray, RA, Jones, A and Andersen, KI (1994). Order Pseudophyllidea Carus, 1863. Keys to the cestode parasites of Vertebrates. Wallingford UKpp. 202-247.

Bylund, G (1975). The taxonomic significance of embryonic hooks in four european *Diphylobothrium* species (Cestoda, *Diphylobothriidae*). *Acta Zoologica Fennica*. 142, pp. 1-22.

Bylund, G (1972). Pathogenic effects of a diphylobothriid plerocercoid on its hosts. *Comm Biol*. 58, pp. 1-11.

Bylund, G (1969). Experimental undersokning av *Diphylobothrium dendriticum* and *D. norvegicum* fran norra Finland. *Parasitol Inst Soc Scient Fenn Tiedoksianto - Information*. 10, pp. 3-17.

Bylund, G, Bang, B and Wilgren, K (1977). Tests with a new compound (Praziquantel) against *Diphylobothrium latum*. *Journal of Helminthology*. 51, pp. 115-119.

Cabrera, R (2003). *Helmintos intestinales en el Perú: análisis de la prevalencia (1981-2001)*. Lima, Perúpp. 113.

Cai, YC, Chen, SH, Yamasaki, H, Chen, JX, Lu, Y, Zhang, YN et al. (2017). Four human cases of *Diphylobothrium nihonkaiense* (Eucestoda: *Diphylobothriidae*) in China with a brief review of Chinese cases. *Korean Journal of Parasitology*. 55, pp. 319-325.

Capuano, D, Okino, M, H, M and Torres, D (2007). *Difilobotriase: relato de caso no municipio de Ribeirao Preto, SP, Brasil..*

RBCA. 39, pp. 163-164.

Catalano, S., Lejeune, M., Tizzani, P., Verocai, G.G., Schwantje, H., Nelson, C. et al. (2015). Helminths of grizzly bears (*Ursus arctos*) and American black bears (*Ursus americanus*) in Alberta and British Columbia, Canada. *Canadian Journal of Zoology*. 93, pp. 765-772. doi: 10.1139/cjz-2015-0063.

Cattan, PE, Atias, A, Babero, BB and Torres, D (1977). Helminthofauna de Chile: V. Primer hallazgo de *Diphylobothrium pacificum* (Nybelin 1931) Margolis 1956, en lobos marinos de la costa chilena. *Revista Ibérica de Parasitología*. 37, pp. 285-290.

Cepeda, GD, Blanco-Barcial, L, Bucklin, A, Berón, CM and Viñas, MD (2012). Molecular systematic of three species of Oithona (Copepoda, Cyclopoida) from the Atlantic Ocean: Comparative Analysis Using 28S rDNA. *Plos One*. 7, pp. e35861. doi: 10.1371/Journal.pone.0035861.

Chai, JY, K Murrell, D and Lymbery, AJ (2005). Fish-borne parasitic zoonoses: status and issues. *Int J Parasitol*. 35, pp. 1233-1254.

Chai, JY (2013). Praziquantel treatment trematode and cestode infections: an update. *Infect Chemother*. 45, pp. 32-43.

Chen, W, Zhang, J, Lu, G, Yuan, Z, Wua, Q, Li, J et al. (2014). Development of an immunochromatographic lateral flow device for rapid diagnosis of *Vibrio cholerae* O1 serotype Ogawa. *Clin Biochem*. 47, pp. 448-454.

Cheng, K, Gao, B, Liu, J and Xu, J (2014). Pulmonary sparganosis mansonii: a case report from a non-endemic region. *J Thorac Dis*. 6, pp. 120-124.

Chero, J, Cruces, C, Iannacone, J, Sáez, G, Alvarino, L, Rodríguez, C et al. (2014). Indices parasitológicos de la merluza Peruana *Merluccius gayi peruanus* (Ginsburg, 1954) (Perciformes:Merlucciidae) adquiridos del terminal pesquero de Ventanilla, Callao, Perú. *Neotrop Helminthol*. 8, pp. 59-76.

Chero, J, Iannacone, J, Cruces, C, Sáez, G and Alvarino, L (2014). Comunidad de metazoos parásitos de la corvina *Cilus gilberti* (Abbott, 1899) (Perciformes: Sciaenidae) en la zona costera de Chorrillos, Lima, Perú. *Neotrop Helminthol*. 8, pp. 163-182.

Chero, J, Sáez, G, Iannacone, J and Aquino, W (2014). Aspectos ecológicos de los helmintos parásitos de lorna *Sciaena deliciosa* (Tschudi, 1846) (Perciformes: Sciaenidae) adquiridos del terminal pesquero de Ventanilla, Callao, Perú. *Neotrop Helminthol*. 8, pp. 59-76.

Ching, HL (1988). The distribution of plerocercoids of *Diphylobothrium dendriticum* (Nitzsch) in sockeye salmon (*Oncorhynchus nerku*) smolts from Great Central Lake, British Columbia. *Canadian Journal of Zoology*. 129, pp. 19-21.

Choi, S, Cho, J, Jung, B-K, Kim, D-G, Jeon, S, Jeon, H-K et al. (2015). *Diphylobothrium nihonkaiense*: wide egg size variation in 32 molecularly confirmed adult specimens from Korea. *Parasitology Research*. 114, pp. 2129-2134. doi: 10.1007/s00436-015-4401-7.

Chubb, JC (1980). Seasonal occurrence of Helminths in freshwater fishes Part (III).\ Larval Cestoda and Nematoda. *Adv Parasitol*. 18, pp. 1-120.

Chung, DI, Kong, HH, Moon, CH, Choi, DM, Kim, TH, Lee, DW et al. (1995). The first human case of *Diplogonoporus balaenopterae* (Cestoda: Diphylobothriidae) infection in Korea. *Korean Journal of Parasitology*. 33, pp. 225-230.

Clavel, A, Bargues, MDolores, Castillo, FJavier, Rubio, Mdel Carmen and Mas-Coma, S (1997). *Diplogonoporiasis* presumably introduced into Spain: first confirmed case of human infection acquired outside the far East. *American Journal of Tropical Medicine and Hygiene*. 57, pp. 317-320.

Conn, D and Swiderski, Z (2008). A standardised terminology of the embryonic envelopes and associated developmental stages of tapeworms (Platyhelminthes: Cestoda). *Folia Parasitol*. 55, pp. 42-52.

Corkum, K (1966). Sparganosis in some vertebrates of Louisiana and observations on a Human infection. *The Journal of*

Parasitology. 52, pp. 444-448.

Corrêa, J R, Santos, PCdos and (2012). Qualidade higienico - sanitária de alface (*Lactuca sativa*) comercializada em supermercados em dourados - MS. *Scientia Agraria*. 13, pp. 53-58.

Cristoffanini, A, Ibarra, H, Vega, I, Martinez, A and Bertoglio, J (1976). Anemia megaloblástica por *Diphylobothrium latum*. *Revista Medica de Chile*. 104, pp. 921-924.

Cui, J, Li, N, Wang, ZQ, Jiang, P and Lin, XM (2011). Serodiagnosis of experimental sparganum infections of mice and human sparganosis by ELISA using ES antigens of *Spirometra mansonii* spargana. *Parasitol Res*. 108, pp. 1551-1556.

Curtis, MA and Bylund, G (1991). *Diphylobothriasis*: fish tapeworm disease in the circumpolar north. *Arctic Med Res*. 50, pp. 18-25.

Dalton, JP, Skelly, P and Halton, DW (2004). Role of the tegument and gut in nutrient uptake by parasitic Platyhelminths. *Can J Zool*. 82, pp. 211-232.

De Marval, F, Gottstein, B, Weber, M and Wicht, B (2013). Imported diphylobothriasis in Switzerland: molecular methods to define a clinical case of *Diphylobothrium* infection as *Diphylobothrium dendriticum*. *Euro Surveill*. 18, pp. pii: 20355.

De Vos, T and Dick, TA (1989). Differentiation between *Diphylobothrium dendriticum* and *D. latum* using isozymes restriction profiles and ribosomal gene probes. *Systematic Parasitology*. 13, pp. 161-166.

Dei-Cas, E, Rodriguez, N, Botto, C and Osimani, JJosé (1976). Larvas plerocercoides de *Spirometra* (*Dibothriocephalidae*) en el hombre y en animales silvestres de Uruguay. *Revista Instituto de Medicina Tropical de Sao Paulo*. 18, pp. 165-172.

Denegri, GM and Reisin, IL (1993). The complete biological cycle of *Diphylobothrium erinaceieuropei* (Cestoidea, Pseudophyllidea) under experimental conditions. *Helminthol*. 30, pp. 177-179.

Desvois, L, Gregory, A, Ancelle, T and Dupouy-Camet, J (2001). Enquête sur l'incidence de la bothriocéphalose en Haute-Savoie (1993-2000). *Bull Epidem Hebd*. 45, pp. 211-213.

Devi, CS, Shashikala, C, Srinivasan, S, Murmu, UC, Barman, P and Kanungo, R (2007). A rare case of diphylobothriasis from Pondicherry, South India. *Indian J Med Microbiol*. 25, pp. 152-154.

Dezfuli, B.S, Pironi, F., Simoni, E., Shinn, A.P and Giari, L. (2007). Selected pathological, immunohistochemical and ultrastructural changes associated with an infection by *Diphylobothrium dendriticum* (Nitzsch, 1824) (Cestoda) plerocercoids in *Coregonus lavaretus* (L.) (Coregonidae). *Journal of Fish Diseases*. 30, pp. 471-482. doi: 10.1111/j.1365-2761.2007.00833.x.

Dick, TA, Nelson, PA and Choudhury, A (2001). *Diphylobothriasis*: update on human cases, foci, patterns and sources of human infections and future considerations. *Southeast Asian J Trop Med Public Health*. 32, pp. 59-76.

Dick, TA and Poole, BC (1985). Identification of *Diphylobothrium dendriticum* and *Diphylobothrium latum* from some freshwater fishes of central Canada. *Can J Zool*. 63, pp. 196-201. doi: 10.1139/z85-030.

Dick, T.A. (2008). *Diphylobothriasis*: the *Diphylobothrium latum* human infection conundrum and reconciliation with a worldwide zoonosis. *Food-Borne Parasitic Zoonoses: Fish and Plant-Borne Parasites*. (Murrel, K.D. and Fried, B., ed.). London pp. 151-184.

Donoso-Scroppo, M, Raposo, L, Reyes, H, Godorecci, S and Castillo, G (1986). Megaloblastic anemia secondary to infection by *Diphylobothrium latum*. *Rev Med Chil*. 114, pp. 1171-1174.

Dorucu, M, Crompton, DWT, Huntingford, FA and Walters, DE (1995). The ecology of endoparasitic helminth infections of brown trout (*Salmo trutta*) and rainbow trout (*Oncorhynchus mykiss*) in Scotland. *Folia Parasitologica*. 42, pp. 29-35.

Doschi, R (1972). Studies on the sedimentation of eggs of the fish tapeworm *Diphylobothrium latum* using the flocculating agent aluminium sulphate. Thesis. Ludwig Maximilians Universitat. pp. 70 p.

- Drake, DA, Carreño, AD, Blagburn, BL, Little, S, West, MD, Hendrix, CM et al. (2008). Proliferative sparganosis in a dog. *J Am Vet Med Assoc.* 233, pp. 1756-1760.
- Dupouy-Camet, J and Peduzzi, R (2004). Current situation of human diphyllobothriasis in Europe. *Euro Surveillance.* 9, pp. 31-35.
- Dupouy-Camet, J, Haidar, M, Dei-cas, E, Espinat, L, Benmostefa, A, Guillard, J et al. (2013). Prévalence de l'infestation par *Diphyllobothrium latum* de différents poissons des lacs Léman, du Bourget et d'Annecy et évaluation de l'incidence des cas humains auprès des laboratoires d'analyse médicale de la région. *Bulletin epidemiologique sante animale et alimentation.* 67, pp. 2-5.
- Eberhard, ML, Thiele, EA, Yembo, GE, Yibi, MS, Cama, VA and Ruiz-Tiben, E (2015). Thirty-seven human cases of Sparganosis from Ethiopia and South Sudan caused by *Spirometra* spp.. *Am J Trop Med Hyg.* 93, pp. 350-355.
- Eduardo, M, Sampaio, J, Goncalves, E, Castilho, V, Randi, A, Thiago, C et al. (2005). *Diphyllobothrium* spp.: um parasita emergente em Sao Paulo, associado ao consumo de peixe cr-sushis e sashimis, Sao Paulo, Marco de 2005. *Bol Epidemiológico Paulista, Sao Paulo.* 2, pp. 1-5.
- Eduardo, M, Sampaio, J, Susuki, E, César, M, Goncalves, E, Castilho, V et al. (2005). Investigacao epidemiologica do surto de difilobotriase, Sao paulo, maio de 2005. *Bol Epidem Paulista, Sao Paulo.* 17, pp. 1-12.
- Eguchi, S (1926). Studies on *Dibothriocephalus latus*, with special reference to its life history in Japan. *Byorigaku Kiyo.* 3, pp. 1-66.
- Emmel, VE, Inamine, E, Secchi, C, Brodt, TCZ, Amaro, MCO, Cantarelli, VV et al. (2006). *Diphyllobothrium latum*: relato de caso no Brasil. *Rev Soc Bras Med Trop.* 39, pp. 82-84.
- Eom, K., Choe, S, Kim, K and Jeon, HK (2015). Mitochondrial genome sequences of *Spirometra erinaceieuropaei* and *S. decipiens* (Cestoidea: Diphyllobothriidae). *Korean Journal of Parasitology.* 53, pp. 455-463.
- Eom, K and Kim, T (2009). Migration of cerebral sparganosis to the ipsilateral cerebellar hemisphere. *Acta Parasitol.* 54, pp. 276-280. doi: 10.2478/s11686-009-0035-1.
- Escalante, H (1983). Larvas plerocercoides de *Diphyllobothriidae* Lühe, 1910: Hallazgo en peces marinos de consumo humano en la costa peruana. *Bol Chile Parasitol.* 38, pp. 50-52.
- Escalante, H and Chico-Ruiz, J (1988). Viabilidad e infectividad de las larvas plerocercoides de *Diphyllobothrium pacificum* expuestas al "cebiche". *Rebiol.* 8, pp. 1-8.
- Escalante, H, Jara, C and Chico-Ruiz, J (1988). Nuevos hallazgos de larvas plerocercoides de *Diphyllobothrium pacificum* en peces marinos del Perú. *Rebiol.* 8, pp. 7-16.
- Escalante, H, Jara, CA and Chico-Ruiz, J (1987). Obtencion de coracidios de *Diphyllobothrium pacificum* bajo condiciones experimentales. *Decimo Congreso Latinoamericano de Microbiologia. Trujillo, Perúpp.* Pp 68.
- Escalante, H and Miranda, H (1986). *Diphyllobothrium pacificum*: Hallazgo de larvas plerocercoides en peces marinos del Perú y desarrollo de formas adultas del parásito en *Canis familiaris*. *Bol Chile Parasitol.* 41, pp. 7-13.
- Essex, HE and Magath, TB (1931). A comparison of the viability of ova of the broad fish tapeworm, *Diphyllobothrium latum*, from man and dogs: its bearing on the spread of infestation with this parasite. *Am J Hyg.* 24, pp. 698-704.
- Esteban, JG, Muñoz-Antoli, C, Borrás, M, Colomina, J and Toledo, R (2014). Human infection by a "fish tapeworm", *Diphyllobothrium latum*, in a non-endemic country. *Infection.* 42, pp. 191-194.
- Fang, FC, Billman, ZP, Wallis, CK, Abbott, AN, Olson, JC, S, D et al. (2015). Human *Diphyllobothrium nihonkaiense* infection in Washington State. *Journal of Clinical Microbiology.* 53, pp. 1355-1357.
- Faust, EC, Campbell, HE and Kellog, CR (1929). Morphological and biological studies on the species of *Diphyllobothrium* in

China. Am J Hyg. 9, pp. 560-583.

Feachem, RG, Bradley, DJ, Garelick, H and D. Mara, D (1983). Sanitation and Disease: health Aspects of Excreta and Wastewater Management. World Bank Studies in Water Supply and Sanitation 3. pp. 501. doi: 10.1016/0035-9203(84)90011-7.

Feist, S and Longshaw, M (2008). Histopathology of fish parasite infections- importance for populations. J Fish Biol. 73, pp. 2143-2160.

Ferreira, LF, Araújo, AJG, Confalonieri, UEC and Nuñez, L (1984). The finding of eggs of *Diphyllbothrium* in human coprolites (4, 100-1,950 B.C.) from northern Chile. Memorias do Instituto Oswaldo Cruz. 79, pp. 175-180.

Figuerola, L, Torres, P, Franjola, R and Schlatter, R (1980). Investigaciones sobre Pseudophyllidea (Carus, 1813) en el sur de Chile. VI. Infección por *Diphyllbothrium* (Cobbold) en *Larus maculipennis* Lichtenstein en el lago Calafquén. Bol Chil Parasitol. 35, pp. 71-73.

Figuerola, L, Torres, P, Schlatter, R, Asenjo, F, Franjola, R and Contreras, B (1979). Investigaciones sobre Pseudophyllidea (Carus, 1813) en el sur de Chile III: Estudio sobre *Diphyllbothrium* sp. en aves del Lago Calafquén (39° 32' S 72° 09' W). Bol Chil Parasitol. 34, pp. 13-20.

Flisser, A and Willingham, AL (2013). Teniasis por *Taenia solium*, *Taenia saginata* y *Taenia asiatica*. Parasitología Humana. (Apt, W, ed.). pp. 184-194.

Flores, JM, Vidaurre, MT, Rivera, ML and Rosales, MC (2002). *Diphyllbothrium pacificum* en niños del Perú. Diagnostico. 41, pp. 161-164.

Freeman, RS and Jameison, J (1972). Parasites of Eskimos at Igloodik and Hall Beach, Northwest Territories. Proceedings of the 3rd International Congress on Circumpolar Health. (Shaphard, RJ and Ioth, S, ed.). Torontopp. 306-315.

Gallagher, C.P and Dick, T.A (2010). Trophic structure of a landlocked Arctic char *Salvelinus alpinus* population from southern Baffin Island, Canada. Ecology of Freshwater Fish. 19, pp. 39-50. doi: 10.1111/j.1600-0633.2009.00387.x.

Gallegos, R and Brousselle, C (1991). Parasitoses intestinales chez les habitants d'un archipel Equatorien. Bull Soc Fr Parasitol. 9, pp. 219-223.

Garcías, F, Mendoza, R and George-Nascimento, M (2001). Variación entre años de las infracomunidades de parásitos metazoos de la corvina *Cilus gilberti* (Pisces: Scaniae) en Chile. Revista Chilena de Historia Natural. 74, pp. 833-840.

George-Nascimento, M and Huet, B (1984). Una aproximación ecológica al estudio del parasitismo en el congrio negro *Genipterus maculatus* (Tschudi) (Pisces: Ophidiidae). Biología Pesquera. 13, pp. 23-30.

George-Nascimento, M, Moscoso, D, Niklitschek, E and Gonzalez, K (2011). Variación geográfica de las comunidades de parásitos de la merluza de tres aletas *Micromesistius australis* al sur de Sudamérica. Revista de Biología Marina y Oceanografía. 46, pp. 53-58.

Gerasimov, I.V. (1987). Establishing the routes of contamination of the water of the Krasnoyarsk reservoir by *Diphyllbothrium latum* ova as a basis for elaborating prophylactic measures. Meditsinskaya Parazitologiya i Parazitarnye Bolezni. 5, pp. 72-75.

Go, YB, Lee, EH, Cho, J, Choi, S and Chai, JY (2015). *Diphyllbothrium nihonkaiense* infections in a family. Korean Journal of Parasitology. 53, pp. 109-112.

Golay, M and Mariaux, J (1995). Situation de *Diphyllbothrium latum* L., 1758 (Cestoda: Pseudophyllidea) dans quatre lacs du plateau Suisse. Bulletin de la Société Neuchateloise des Sciences Naturelles. 118, pp. 79-86.

González, H, Torres, P, Figuerola, L, Contreras, B and Franjola, R (1978). Researches on Pseudophyllidea (Carus, 1813) in the South of Chile. II Hepatic and splenic: Pathology by plerocercoids infections of *Diphyllbothrium* spp. in *Salmo gairdnerii* Richardson, 1836 of Calafquen Lake.. Indian Journal of Parasitology. 2, pp. 127-129.

- Grabiec, S, Guttowa, A and Michajlow, Q (1963). Effect of light stimulus on hatching of coracidia of *Diphyllbothrium latum* (L). *Acta Parasitol Pol.* 11, pp. 229-238.
- Grammeltvedt, A (1973). Differentiation of the tegument and associated structures in *Diphyllbothrium dendriticum* Nitsch (1824) (Cestoda: Pseudophyllidea). An electron microscopical study.. *Int J Parasitol.* 3, pp. 321-327. doi: 10.1016/0020-7519(73)90110-0.
- Gustafsson, M and Vaihela, B (1981). Two types of frontal glands in *Diphyllbothrium dendriticum* (Cestoda, Pseudophyllidea) and their fate during the maturation of the worm. *Z Parasitendkd.* 66, pp. 145-154.
- Gustinelli, A, Menconi, V, Prearo, M, Caffara, M, Righetti, M, Scanzio, T et al. (2016). Prevalence of *Diphyllbothrium latum* (Cestoda: Diphyllbothriidae) plerocercoids in fish species from four Italian lakes and risk for the consumers.. *International journal of food microbiology.* 235, pp. 109-12. doi: 10.1016/j.ijfoodmicro.2016.06.033.
- Gutierrez, Y (2000). *Diagnostic pathology of parasitic infections with clinical correlations.* Oxford, United States
- Guttowa, A (1966). Experimental coinfection of Copepoda natural infected *Protocephalus* sp. with the larvae of *Diphyllbothrium latum*. *Acta Parasitol Pol.* 14, pp. 339-404.
- Guttowa, A (1965). Copepoda as intermediate hosts of *Diphyllbothrium latum* (L) in European foci of infection. *Bulletin du Museum National d'Histoire Naturelle.* 37, pp. 528-532.
- Guttowa, A (1963). Natural focus of infection of plankton crustaceans with procercoids of *Diphyllbothrium latum* L. in Finland. *Acta Parasitologica Polonica.* 11, pp. 145-153.
- Guttowa, A (1961). Experimental investigations on the systems, procercoids of *Diphyllbothrium latum* (L.) - Copepoda. *Acta Parasitol Pol.* 9, pp. 371-408.
- Guttowa, A (1959). The dog as the host of *Diphyllbothrium latum* (L.). *Wiadomosci Parazytologiczne.* 5, pp. 585-590.
- Halvorsen, O (1966). Studies on the helminth fauna of Norway. VIII an experimental investigation of copepods as first intermediate hosts for *Diphyllbothrium norvegicum* vik. *Nytt Mag Zool.* 13, pp. 83-117.
- Halvorsen, O and Wissler, K (1973). Studies on the helminth fauna of Norway XXVIII: An eperimental study of the ability of *Diphyllbothrium latum* (L.), *D. dendriticum* (Nitzsch), and *D. ditremum* (Creplin) (Cestoda, Pseudophyllidea) to infect paratenic hosts. *Norwegian Journal of Zoology.* 21, pp. 201-210.
- Halvorsen, O (1970). Studies on the Helminth Fauna of Norway XV: On the taxonomy and biology of plerocercoids of *Diphyllbothrium* Cobbold, 1858 (Cestoda, Pseudophyllidea) from North-western Europe. *Nytt Magasin for Zoologi.* 18, pp. 113-174.
- Halvorsen, O (1967). The occurrence of procercoids of *Diphyllbothrium norvegicum* Vik in the abdomen of copepod hosts. *Nytt Magasin for Zoologi.* 14, pp. 142-146.
- Henricson, J (1977). The abundance and distribution of *Diphyllbothrium dendriticum* (Nitzsch) and *D. ditremum* (Creplin) in the Char *Salvelinus alpinus* (L.) in Sweden. *Journal Of Fish Biology.* 11, pp. 231-248. doi: 10.1111/j.1095-8649.1977.tb04116.x.
- Hernández-Ortiz, J, Scholz, T, Brabec, J, Kuzmina, T and Kuchta, R (2015). High morphological plasticity and global geographical distribution of the Pacific broad tapeworm *Adenocephalus pacificus* (syn. *Diphyllbothrium pacificum*): molecular and morphological survey.. *Acta Tropica.* 149, pp. 168-178. doi: 10.1016/j.actatropica.2015.05.017.
- Hickey, M and Harris, J (1947). Progress of the *Diphyllbothrium* epizootic at Poulaphouca reservoir, Co. Wicklow, Ireland. *Journal of Helminthology.* 22, pp. 13-28.
- Hilliard, DK (1960). Studies on the helminth fauna of Alaska. XXXVIII. The taxonomic significance of eggs and coracidia of some diphyllbothriid cestodes. *J Parasitol.* 46, pp. 703-716.

- Hong, D, Feng, J, Liu, H, Li, X, Gong, L, Yang, Z et al. (2016). Prevalence of *Spirometra mansoni* in dogs, cats, and frogs and its medical relevance in Guangzhou, China. *Int J Infect Dis.* 53, pp. 41–45. doi: 10.1016/j.ijid.2016.10.013. Epub 2016 Oct 21.
- Hong, D, Xie, H, Zhu, M, Wan, H, Xu, R and Wu, Y (2013). Cerebral sparganosis in mainland Chinese patients. *Clin Neurosci.* 20, pp. 1514–1519.
- Iamaroon, A, Sukontason, K and Sukontason, K (2002). Sparganosis: a rare case of the oral cavity. *Journal of Oral Pathology and Medicine.* 31, pp. 558–560. doi: 10.1034/j.1600-0714.2002.00051.x.
- Iannaccone, J and Alvarino, L (2009). Dinamica poblacional de la diversidad parasitaria de la "cabrilla" *Paralabrax humeralis* (Teleostei: Serranidae) en Chorrillos, Lima, Peru. *Neotropical Helminthology.* 3, pp. 73–88.
- Ihesiulor, GU, Kashibu, E, Azeez-Akande, O and Imoru, M (2013). Helminths of the gastrointestinal tract among children in Kano, Northern Nigeria. *Asian Journal of Biological and Life Sciences.* 2, pp. 122–126.
- Jacobus, K, Marigo, J, Ph, D, Gastal, SBainy, Akemi, S and Tseng, F (2016). Identification of Respiratory and Gastrointestinal Parasites of Three Species of Pinnipeds (*Arctocephalus Australis* , *Arctocephalus Gazella* , and *Otaria Flavescens*) in Southern Brazil. *Journal of Zoo and Wildlife Medicine.* 47, pp. 132–140. doi: 10.1638/2015-0090.1.
- Janicki, C and Rosen, F (1917). Le cycle evolutif du *Dibothriocephalus latus* L.. *Bull Soc Vaud Sci Nat.* 42, pp. 19–53.
- Jenkins, EJ, Castrodale, LJ, de Rosemond, SJ, Dixon, BR, Elmore, SA, Gesy, KM et al. (2013). Tradition and transition: parasitic zoonoses of people and animals in Alaska, northern Canada, and Greenland. *Adv Parasitol.* 82, pp. 33–204.
- Jeon, HK, Park, H, Lee, D, Choe, S, Sohn, WM and Eom, KS (2016). Molecular detection of *Spirometra decipiens* in the United States. *Korean Journal of Parasitology.* 54, pp. 503–507. doi: org/10.3347/kjp.2016.54.4.503.
- Jeon, H-K, Kim, K-H, Huh, S, Chai, J-Y, Min, D-Y, Rim, H-J et al. (2009). Morphologic and genetic identification of *Diphyllbothrium nihonkaiense* in Korea.. *Korean J Parasitol.* 47, pp. 369–375. doi: 10.3347/kjp.2009.47.4.369.
- Jeon, H-K, Park, H, Lee, D, Choe, S, Kim, K-H, Huh, S et al. (2015). Human infections with *Spirometra decipiens*. Plerocercoids identified by morphologic and genetic analyses in Korea. *Korean J Parasitol.* 53, pp. 299–305. doi: 10.3347/kjp.2015.53.3.299.
- Jimenez, JA, Rodríguez, S, Gamboa, R, Rodriguez, L and Garcia, HH (2012). *Diphyllbothrium pacificum* infection in seldom associated with megaloblastic anemia. *American Journal of Tropical Medicine and Hygiene.* 87, pp. 897–901.
- Johnson, S (1975). First Intermediate Host of *Diphyllbothrium sebae* (Cestoda: Pseudophyllidea) in Nature. *The Journal of Parasitology.* 61, pp. 74.
- Kamiya, M and Ooi, HK (1991). Current status of Food-borne parasitic zoonoses in Japan. *Southeast Asian J Trop Med Public Health.* 22, pp. 48–53.
- Kamo, H, Iwata, S, Htasushika, R and Maejima, J (1973). Experimental studies on the life cycle of *Diplogonoporus grandis* II. Experimental infection of marine copepods with coracidia. *Japanese Journal of Parasitology.* 22, pp. 79–89.
- Kamo, H (1999). Guide to identification of *Diphyllbothriid* Cestodes. Tokyo Japan pp. 146.
- Kamo, H, Yazaki, S, Fukomoto, S, Maejima, J and Kawasaki, H (1986). Evidence of egg-discharging periodicity in experimental human infection with *Diphyllbothrium latum* (Linné, 1758). *Japanese Journal of Parasitology.* 35, pp. 53–57.
- Karasev, AB, Mitenev, VK and Kalinina, NR (1997). Parasite fauna of cage-reared rainbow trout *Oncorhynchus mykiss* (Walbaum). Research in freshwater farms (Kola Peninsula, Russia). *Bulletin of the European Association of Fish Pathologists.* 17.
- Khan, RA (2009). Parasites causing disease in wild and cultured fish in Newfoundland. *Icel Agric Sci.* 22, pp. 29–35.

- Khromenkova, EP (1993). Routes whereby helminth ova become disseminated in the recreational areas and installations of the lower Don region. *Meditinskaya Parazitologiya i Parazitarnye Bolezni*. 3, pp. 29-30.
- Khromenkova, EP, Ermolova, RS, Basmadzhan, YUA and Zinchenko, LN (1995). Study of conditions for the possible formation of foci of diphylobothriasis on the lower reaches of the Don. *Meditinskaya Parazitologiya i Parazitarnye Bolezni*. 2, pp. 35-39.
- Kim, HJ, Eom, KS and Seo, M (2014). Three cases of *Diphylobothrium nihonkaiense* infection in Korea. *Korean Journal of Parasitology*. 52, pp. 673-676.
- Kino, H, Hori, W, Kobayashi, H, Nakamura, N and Nagasawa, K (2002). A mass occurrence of human infection with *Diplogonoporus grandis* (Cestoda: Diphylobothriidae) in Shizuoka Prefecture, central Japan. *Parasitology International*. 51, pp. 73-79.
- Klekowski, R and Guttowa, A (1968). Respiration of *Eudiptomus gracilis* infected with *Diphylobothrium latum*. *Exp Parasitol*. 22, pp. 279-287.
- Knoff, M, S Clemente, DSao, De Andrada, C, De Lima, FC, Padovani, R, Da Fonseca, MC et al. (2008). Cestóideos Pseudophyllidea parasitos de congrio-rosa, *Genypterus brasiliensis* Regan, 1903 comercializados no estado do Rio de Janeiro, Brasil. *R bras Ci Vet*. 15, pp. 28-32. doi: org/10.4322/rbov.2014.192.
- Knoff, M, R Pinto, M, S Clemente, Cde Sao, Fonseca, MCGoncalve and D Gomes, C (2011). *Diphylobothrium latum* and *Diphylobothrium* sp. as the agents of diphylobothriasis in Brazil: morphological analysis and of two new case reports. *Revista Brasileira de Medicina Veterinaria*. 33, pp. 159-164.
- Kuchta, R, Esteban, J-G, Brabec, J and Scholz, T (2014). Misidentification of *Diphylobothrium* species related to global fish trade, Europe. *Emerg Infect Dis*. 20, pp. 1955-1957.
- Kuchta, R, Oros, M, Ferguson, J and Scholz, T (2017). *Diphylobothrium nihonkaiense* tapeworm larvae in salmon from North America. *Emerg Infect Dis*. 23, pp. 351-353.
- Kuchta, R, Brabec, J, Kubáčková, P and Scholz, T (2013). Tapeworm *Diphylobothrium dendriticum* (Cestoda) Neglected or Emerging Human Parasite?. *Plos Neglect Trop D*. 7, pp. e2535. doi: 10.1371/journal.pntd.0002535.
- Kuchta, R, Scholz, T, Brabec, J and Narduzzi-Wicht, B (2015). *Diphylobothrium*, *Diplogonoporus* and *Spirometra*. *Biology of Foodborne Parasites*. Section III. Important foodborne helminths. (Xiao, L, Ryan, U and Feng, F, ed.). Boca Raton, United Statespp. 299-325.
- Kuchta, R, Serrano-Martinez, M and Scholz, T (2015). Pacific Broad Tapeworm *Adenocephalus pacificus* as a Causative Agent of Globally Reemerging Diphylobothriosis. *Emerg Infect Dis*. 21, pp. 1697-1703. doi: 10.3201/eid2110.150516.
- Kuhlow, F (1953). Über die Entwicklung und anatomie von *Diphylobothrium dendriticum* Nitzsch 1824. *Z f Parasitenkunde*. 16, pp. 1-35.
- Kuhn, JA, Frainer, A, Knudsen, R, Kristoffersen, R and Amundsen, P-A (2016). Effects of fish species composition on *Diphylobothrium* spp. infections in brown trout - is three-spined stickleback a key species?. *Journal of Fish Diseases*. pp. 1-11. doi: 10.1111/jfd.12467.
- Kuperman, BI and Davydov, VG (1982). The fine structure of frontal glands in adult cestodes. *Int J Parasitol*. 12, pp. 285-293. doi: 10.1016/0020-7519(82)90031-5.
- Lal, S and Steinhart, A-H (2007). *Diphylobothrium latum*: a case of an incidental finding.. *World journal of gastroenterology*. 13, pp. 1875-1876.
- Le Bailly, M, Leuzinger, U, Schlichtherle, H and Bouchet, F (2007). Crise économique au Néolithique à la transition Pfyn-Horgen (3400 BC): contribution de la paléoparasitologie. *Anthropozoologica*. 42, pp. 175-185.
- Le Bailly, M., Leuzinger, U., Schlichtherle, H. and Bouchet, F. (2005). *Diphylobothrium*: Neolithic parasite?. *Journal of*

Parasitology. 91(4), pp. 957-959.

Lee, KJ, Myung, NH and Park, HW (2010). A case of sparganosis in the leg. Korean Journal of Parasitology. 48, pp. 309-312.

Lee, SH, Chai, JY and BS, S (1984). Two cases of human infection by adult of *Spirometra erinacei*. Korean Journal of Parasitology. 22, pp. 66-71.

Lee, SH, We, JS, Sohn, WM, Hong, ST and Chai, JY (1990). Experimental life history of *Spirometra erinacei*. Korean Journal of Parasitology. 28, pp. 161-173.

Lee, Y-I, Seo, M and Park, H-W (2014). Recurred Sparganosis 1 Year after Surgical Removal of a Sparganum in a Korean Woman. Korean J Parasitol. 52, pp. 75-78. doi: 10.3347/kjp.2014.52.1.75.

Lestinova, K, Soldanova, M, Scholz, T and Kuchta, R (2016). Eggs as a Suitable Tool for Species Diagnosis of Causative Agents of Human Diphyllbothriosis (Cestoda). PLoS Negl Trop Dis. 10, pp. 1-15. doi: 10.1371/journal.pntd.0004721.

Li, MW, Song, HQ, Li, C, Lin, HY, Xie, WT, RQ, L et al. (2011). Sparganosis in mainland China. International Journal of Infectious Diseases. 15, pp. 154-156. doi: org/10.1016/j.ijid.2010.10.001.

Lin, T, Cui, FY and Kiu, DM (2007). Biological control experiment of excess propagation of Cyclops for drinking water security. J Environ Sci. 19, pp. 290-294.

Lin, XM, Liu, CJ, Zhang, HW, Zheng, LY, Yan, QY, He, LJ et al. (2010). Epidemiological investigation on sparganosis mansonii and animal experiments. Chinese Journal of Parasitology Parasitic Diseases. 28, pp. 132-134.

Lipatov, EI, Sosnin, EA and Avdeev, SM (2016). The inactivation of helminth eggs with the narrow-bandwidth radiation of excimer lamps. Russian Journal of Parasitology. 35, pp. 107-113.

Liu, Q, Li, M-W, Wang, Z-D, Zhao, G-H and Zhu, X-Q (2015). Human sparganosis a neglected food borne zoonosis. Lancet Infectious Diseases. 15, pp. 1226-1235.

Llaguno, M, Cortez-Escalante, J, Waikagul, J, Faleiros, A, Chagas, F and Castro, C (2008). *Diphyllbothrium latum* infection in a non-endemic country: case report. Rev Soc Bras Med Trop. 41, pp. 301-303.

Lloyd, S (1998). Other cestode infections: hymenolepiosis, diphyllbothriosis, coenurosis, and other adult and larval cestodes. Zoonoses. (Palmer, SR, Soulsby, L and Simpson, DIH, ed.). London, United Kingdom. 651-663.

Lou, HY, Tsai, PC, Chang, CC, Lin, YH, Liao, CW, Kao, TC et al. (2007). A case human of diphyllbothriasis in northern Taiwan after eating raw fish fillets. J Microbiol Immunol Infect. 40, pp. 452-456.

Lv, S, Zhang, Y, Steinmann, P, Zhou, XN and Utzinger, J (2010). Helminth infections of the central nervous system occurring in Southeast Asia and the Far East. Adv Parasitol. 72, pp. 351-408.

Mackenzie, K and Longshaw, M (1995). Parasites of the hakes *Merluccius australis* and *M. hubbsi* in the waters around the Falkland islands, southern Chile, and Argentina, with an assessment of their potential value as biological tags. Canadian Journal of Fisheries and Aquatic Sciences. 52, pp. 213-224.

Maejima, J, Yazaki, S, Fukumoto, S, Hiraga, M and Kamo, H (1981). Morphological observation of *Diphyllbothrium pacificum* (Nybelin, 1931) Margolis, 1956 from fur seals, *Callorhynchus ursinus* in Japan. Yonago Acta Medica. 25, pp. 69-79.

Maejima, J, Yazaki, S, Fukumoto, S and Miyahara, M (1990). Ten cases of *Dilagonoporiasis* in Sanin and Kyushu Districts. Japanese Journal of Parasitology. 39, pp. 198-203.

Magath, TB (1929). Experimental studies on *Diphyllbothrium latum*. The American Journal of Tropical Medicine. 9, pp. 17-48.

Magnino, S, Colin, P, Dei-Cas, E, Madsen, M, McLauchlin, J, Nöckler, K et al. (2009). Biological risks associated with

consumption of reptile products. *Int J Food Microbiol.* 134, pp. 163-175.

Malmberg, G (1971). On the proceroid protonephridial systems on three *Diphyllbothrium* species (Cestoda, Pseudophyllidea) and Janicki cercomer theory. *Zool Scripta.* 1, pp. 43-56.

Marcogliese, DJ (1995). The role of zooplankton in the transmission of helminth parasites to fish. *Reviews in Fish Biology and Fisheries.* 5, pp. 336-371.

Margono, SS, Sutjahyono, RW, Kurniawan, A, Nakao, M, Mulyani, T, Wandra, T et al. (2007). *Diphyllbothriasis* and *sparganosis* in Indonesia. *Trop Med Health.* 35, pp. 301-305.

Medina, JP, Tantalean, M and Cano, M (2002). *Diphyllbothrium pacificum* en niños del Perú. *Diagnostico.* 41, pp. 161-164.

Mercado, R, Yamasaki, H, Kato, M, Muñoz, V, Sagua, H, Torres, P et al. (2010). Molecular identification of the *Diphyllbothrium* species causing *diphyllbothriasis* in Chilean patients. *Parasitology Research.* 106, pp. 995-1000.

Meyer, M and Vik, R (1963). The life cycle of *Diphyllbothrium sebago* (Ward, 1910). *The Journal of Parasitology.* 49, pp. 962-968.

Mezzari, A and Wiebbelling, A (2008). *Diphyllbothriasis* in Southern Brazil. *Clin Microbiol Newsl.* 30, pp. 28-29.

Miranda, H, Fernandez, W and Ibañez, N (1968). *Diphyllbothriasis* investigación de *Diphyllbothrium pacificum* (Nybelin, 1931) Margolis, 1956, en *Otaria byronia* (Sin *Otaria flavescens*) y en peces marinos. *Archivos Peruanos de Patología Clínica.* 22, pp. 9-24.

Mitchell, P.D. (2015). Sanitation, latrines and intestinal parasites in past populations. Ashgate Publishing Ltd. pp. 290.

Miyadera, H, Kokaze, A, Kuramochi, T, Kita, K, Machinami, R, Noya, O et al. (2001). Phylogenetic identification of *Sparganum proliferum* as a pseudophyllidean cestode by the sequence analyses on mitochondrial COI and nuclear *sdhB* genes.. *Parasitology International.* 50, pp. 93-104. doi: 10.1016/S1383-5769(01)00071-X.

Moravec, F (2004). Metazoan parasites of salmonid fishes of Europe. Praha, Czech Republic

Moulinier, R, Martinez, E, Torres, J, Noya, O, de Noya, BA and Reyes, O (1982). Human proliferative *sparganosis* in Venezuela, report of a case. *Am J Trop Med Hyg.* 31, pp. 358-363.

Mueller, JF (1961). The laboratory propagation of *Spirometra mansonoides* as an experimental tool. IV Experimental inversion of the primary axis in the developing egg. *Exp Parasitol.* 11, pp. 311-318.

Mueller, JF (1937). A repartition of the genus *Diphyllbothrium*. *Journal of Parasitology.* 23, pp. 308-310.

Mueller, JF (1974). The biology of *Spirometra*. *J Parasitol.* 60, pp. 3-14.

Mueller, JF (1966). The laboratory propagation of *Spirometra mansonoides* (Mueller, 1935) as an experimental tool. VII. Improved techniques and additional notes on the biology of the cestode. *J Parasitol.* 52, pp. 437-443. doi: 10.2307/3276304.

Mueller, JF (1959). The laboratory propagation of *Spirometra mansonoides* as an experimental tool. I. Collecting, incubation and hatching of the eggs. *J Parasitol.* 45, pp. 353-361.

Muller, R. (2002). Worms and human disease. Wallingford doi: 10.1079/9780851995168.0000.

Myadagsuren, N, Davaajav, A, Wandra, T, Sandar, T, Ichinkhorloo, P, Yamasaki, H et al. (2007). *Taeniasis* in Mongolia, 2002-2006. *Am J Trop Med Hyg.* 77, pp. 342-346.

Nakamura, N, Nakayama, J, Shimazu, T, Kojima, S and Uchikawa, K (1993). First two cases of *Diplogonoporiasis Grandis* from Nagano Prefecture, Japan. *Japanese Journal of Parasitology.* 42, pp. 434-437.

- Nakamura, T, Hara, M, Matsuoka, M, Kawabata, M and Tsuji, M (1990). Human proliferative sparganosis. A new Japanese case. *Am J Trop Med Hyg.* 94, pp. 224-228.
- Neghme, A, Donckaster, R and Silva, R (1950). *Diphyllbothrium latum* en Chile. *Revista Medica de Chile.* 78, pp. 410-411.
- Nicolaud, J, Yera, H and Dupouy-Camet, J (2005). Prévalence de l'infestation par *Diphyllbothrium latum*, L., 1758 chez les perches (*Perca fluviatilis*) du Lac Léman. *Parasite.* 12, pp. 362-364.
- Nithiuthai, S, Anantaphruti, MT, Waikagul, J and Gajadhar, A (2004). Waterborne zoonotic helminthiasis. *Vet Parasitol.* 126, pp. 167-193.
- Noya, O, de Noya, BA, Arrechdera, H and Torres, J (1992). *Sparganum proliferum*: an overview of its structure and ultrastructure. *Int J Parasitol.* 22, pp. 631-640.
- Nyberg, W, Grasbeck, R, Saarni, M and Von Bonsdorff, B (1961). Serum vitamin B12 levels and incidence of tapeworm anemia in a population heavily infected with *Diphyllbothrium latum*. *Am J Trop Med Hyg.* 9, pp. 606-612.
- Oda, FH, Borteiro, C, da Graca, RJ, Tavares, LER, Crampet, A, Guerra, V et al. (2016). Parasitism by larval tapeworms genus *Spirometra* in South American amphibians and reptiles: new records from Brazil and Uruguay, and a review of current knowledge in the region. *Acta Tropica.* 164.
- Okamoto, M, Iseto, C, Shibahara, T, Sato, MO, Wandra, T, Craig, PS et al. (2007). Intraspecific variation of *Spirometra erinaceieuropaei* and phylogenetic relationship between *Spirometra* and *Diphyllbothrium* inferred from mitochondrial (CO1) gene sequences. *Parasitology International.* 56, pp. 235-238. doi: 10.1016/j.parint.2007.03.003.
- Okino, T, Ushirogawa, H, Matoba, K, Nishimatsu, S and Saito, M (2017). Establishment of the complete life cycle of *Spirometra* (Cestoda: Diphyllbothriidae) in the laboratory using a newly isolated triploid clone. *Parasitology international.* 66, pp. 116-118.
- Oliva, ME and Ballón, I (2002). Metazoan parasites of the Chilean hake *Merluccius gayi gayi* as a tool for stock discrimination. *Fish Res.* 56, pp. 313-320.
- Ooi, H, Chang, S, Huang, C, Kawakami, Y and Uchida, A (2000). Survey of *Spirometra erinaceieuropaei* in frogs in Taiwan and its experimental infection in cats. *J Helminthol.* 74, pp. 173-176. doi: 10.1017/S0022149X0000024X.
- Oshima, T and Wakai, R (1983). Epidemiology of *Diphyllbothrium latum* infection in Japan, with special reference to infection of cherry salmon. *Jpn J Antibiot.* 36, pp. 566-572.
- Osorio, G, Daiber, A, Donckaster, R, Ubilla, M, Con, I, Anguita, T et al. (1974). Anemia megaloblástica grave por *Diphyllbothrium latum*. *Revista Medica de Chile.* 102, pp. 700-703.
- Park, SH, Jeon, HK and Kim, JB (2015). Four additional cases of *Diphyllbothrium nihonkaiense* infection confirmed by analysis of COX1 gene in Korea. *Korean Journal of Parasitology.* 53, pp. 105-108.
- Pasternak, AF, Huntingford, FA and Crompton, DWT (1995). Changes in metabolism and behaviour of the freshwater copepod *Cyclops strenuus abyssorum* infected with *Diphyllbothrium* spp. *Parasitol.* 110, pp. 395-399. doi: 10.1017/S0031182000064738.
- Pastor-Valle, J, González, LM, Martín-Clemente, JP, Merino, FJ, Gottstein, B and Garate, T (2014). Molecular diagnosis of diphyllbothriosis in Spain, most presumably acquired via imported fish, or sojourn abroad. *New Microbes New Infect.* 2, pp. 1-6.
- Paugam, A, Yera, H, Poirier, P, Lebuissou, A and Dupouy-Camet, J (2009). Bothriocéphalose à *Diphyllbothrium nihonkaiense* : un nouveau risque lié à la consommation de saumon. *La Presse Médicale.* 38, pp. 675-677. doi: 10.1016/j.lpm.2008.06.017.
- Peduzzi, R. and Boucher-Rodoni, R. (2001). Resurgence of human bothrioccephalosis (*Diphyllbothrium latum*) in the subalpine lake region. *Journal of Limnology.* 60, pp. 41-44. doi: 10.4081/jlimnol.2001.41.

- Phunmanee, A, Boonsawat, W, Indharapoka, B, Tuntisirin, C and Kularbkeaw, J (2001). Pulmonary sparganosis: a case report with five years follow-up. *J Med Assoc Thai*. 84, pp. 130-135.
- Presti, A, Aguirre, D, Andrés, P, Daoud, L, Fortes, J and Muñiz, J (2015). Cerebral sparganosis: case report and review of the European cases. *Acta Neurochir*. 157, pp. 1339-1343. doi: 10.1007/s00701-015-2466-9.
- Raether, W and Hanel, H (2003). Epidemiology, clinical manifestations and diagnosis of zoonotic cestode infections: an update. *Parasitol Res*. 91, pp. 412-438.
- Rahkonen, R and Valtonen, E (1998). Role of water temperature on the size, migration activity and pathogenicity of *Diphyllbothrium dendriticum* (Cestoda) plerocercoids in brown trout *Salmo trutta m. lacustris* (L.). *Ann Zool Fennici*. 35, pp. 107-113. doi: 10.1016/S0020-7519(97)00123-9.
- Rahkonen, R. and Koski, P. (1997). Occurrence of cestode larvae in brown trout after stocking in a large regulated lake in northern Finland. *Diseases of Aquatic Organisms*. 31, pp. 55-63. doi: 10.3354/dao031055.
- Rahkonen, R, Aalto, J, Koski, P, Särkkä, J and Juntunen, K (1996). Cestode larvae *Diphyllbothrium dendriticum* as a cause of heart disease leading to mortality in hatchery-reared sea trout and brown trout. *Diseases of Aquatic Organisms*. 25, pp. 15-22. doi: 10.3354/dao025015.
- Rahman, M, Lee, E-G and Bae, Y-A (2011). Two-dimensional immunoblot analysis of antigenic proteins of *Spirometra* plerocercoid recognized by human patient sera.. *Parasitol Int*. 60, pp. 139-143. doi: 10.1016/j.parint.2011.01.002.
- Raisanen, S and Puska, P (1984). Fish tapeworm, a disappearing health problem in Finland. *Scand J Soc Med*. 12, pp. 3-5.
- Rausch, RL (1964). Studies on the Helminth fauna of Alaska XLI. Observations on Cestodes or the genus *Diplogonoporus* Lönnberg, 1892. *Can J Zool*. 42, pp. 1049-1069. doi: 10.1139/z64-103.
- Rausch, RL and Adams, AM (2000). Natural transfer of helminths of marine origin to freshwater fishes, with observations on the *Diphyllbothrium alascense*. *J Parasitol*. 86, pp. 319-327.
- Rausch, R and Hilliard, D (1970). Studies on the helminth fauna of Alaska. XLIX. The occurrence of *Diphyllbothrium latum* (Linnaeus, 1758) (Cestoda: Diphyllbothriidae) in Alaska, with notes on other species. *Can J Zoolog*. 48, pp. 1201-1219. doi: 10.1139/z70-210.
- Rausch, R, Adams, A and Margolis, L (2010). Identity of *Diphyllbothrium* spp. (Cestoda: Diphyllbothriidae) from sea lions and people along the Pacific Coast of South America. *J Parasitol*. 96, pp. 359-365. doi: 10.1645/GE-2257.1.
- Reinhard, KJ and Urban, O (2003). Diagnosing ancient diphyllbothriasis from Chinchorro mummies. *Memorias do Instituto Oswaldo Cruz*. 98, pp. 191-193.
- Revenga, JE (1993). *Diphyllbothrium dendriticum* and *Diphyllbothrium latum* in fishes from southern Argentina: association, abundance, distribution, pathological effects, and risk of human infection. *The Journal of Parasitology*. 79, pp. 379-383.
- Revenga, J, Perfumo, CJ, Úbeda, CA and Semenas, LG (1995). Difilobotriasis en salmónidos introducidos en el Parque y la Reserva Nacional Nahuel Huapi. Argentina: Patología de las lesiones producidas por *Diphyllbothrium* spp.. *Archivos de Medicina Veterinaria*. 27, pp. 115-122.
- Robertson, L, Sprong, H, Ortega, Y, Giessen, JRF and Fayer, R (2014). Impacts of globalisation on foodborne parasites. *Trends in parasitology*. 30, pp. 37-52.
- Rodger, HD (1991). *Diphyllbothrium* sp. infections in freshwater reared Atlantic salmon (*Salmo salar* L.). *Aquaculture*. 95, pp. 7-14.
- Rohela, M, Jamaiah, I, Goh, KL and Nissapatorn, V (2006). A second case of diphyllbothriasis in Malaysia. *Southeast Asian J Trop Med Public Health*. 37, pp. 896-898.

- Romanenko, NA, Bobyshev, NM, Yachmenev, NI, Smirnova, ZM and Voinova, VP (1976). The role of agricultural irrigated areas in the spread of helminth infections in the Kaliningrad region. *Voprosy meditsinskoi gel'mintologii (nauchnye trudy)*. Moscow. Tsentral'nyi Institut Usovershenstvovaniya Vrachey. pp. 55-57.
- Rosas, R and Weitzel, T (2014). *Diphyllbothrium latum*. *Revista Chilena de Infectologia*. 31, pp. 211-212.
- Rozas, M., Bohle, H., Sandoval, a., Ildefonso, R., Navarrete, a. and Bustos, P. (2012). First Molecular Identification of *Diphyllbothrium dendriticum* Plerocercoids from Feral Rainbow Trout (*Oncorhynchus mykiss*) in Chile. *Journal of Parasitology*. 98, pp. 1220-1226. doi: 10.1645/JP-GE-3097.1.
- Ruggiero, MA, Gordon, DP, Orrell, TM and Bailly, N (2015). A higher level classification of all living organisms. *Plos One*. 10, pp. e0119248. doi: 10.1371.
- Sagua, H, Neira, I, Araya, J and González, J (2001). Nuevos casos de infección humana por *Diphyllbothrium pacificum* (Nybelin, 1931) Margolis, 1956 en Chile y su probable relación con el fenómeno de El Niño, 1975-2000. *Bol Chil Parasitol*. 56, pp. 22-25.
- Sakurawa, S, Kawanabe, T, Yagasaki, A, Shimizu, A and Shimada, S (2007). Case of subcutaneous sparganosis: use of imaging in definitive preoperative diagnosis. *J Dermatol*. 34, pp. 654-657.
- Sampaio, JLM, de Andrade, VP, M Lucas, daConceio, Fung, L, Gagliardi, SMB, Santos, SRP et al. (2005). *Diphyllbothriasis*, Brazil. *Emerging Infectious Diseases*. 11, pp. 1598-1560.
- Santos, F and Faro, L (2005). The first confirmed case of *Diphyllbothrium latum* in Brazil.. *Mem Inst Oswaldo Cruz, Río de Janeiro*. 100, pp. 585-586.
- Schauer, F, Poppert, S, Technau-Hafsi, K, Mockenhaupt, M, Muntau, B, Hacker, G et al. (2014). Travel-acquired subcutaneous *Sparganum proliferum* infection diagnosed by molecular methods.. *Br J Dermatol*. 170, pp. 741-743. doi: 10.1111/bjd.12679.
- Schmid, H and Watschinger, H (1972). Sparganosis in the Masailand. *Acta Tropica*. 29, pp. 218-230.
- Scholz, T, Garcia, HH, Kuchta, R and Wicht, B (2009). Update on the human broad tapeworm (genus *Diphyllbothrium*), including clinical relevance. *Clin Microbiol Rev*. 22, pp. 146-160. doi: 10.1128/CMR.00033-08.
- Scholz, T and Kuchta, R (2016). Fish-borne, zoonotic cestodes (*Diphyllbothrium* and relatives) in cold climates: A never - ending story of neglected and (re)- emergent parasites. *Food Waterborne Parasitol*. 4, doi: <http://dx.doi.org/10.1016/j.fawpar.2016.07.002>.
- Scioscia, N, Petrigh, R, Beldomenico, P and Denegri, G (2014). The Pampas fox (*Lycalopex gymnocercus*) as new definitive host for *Spirometra erinacei* (Cestoda:Diphyllbothriidae). *Acta Trop*. 133, pp. 78-82. doi: 10.1016/j.actatropica.2014.02.006.
- Semenas, L (2014). Zoonosis de origen acuático: la región de los bosques patagónicos. *Temas de Zoonosis VI*. (J Farjat, B, Enria, D, Martino, P, Rosenzvit, M and Seijo, A, ed.).Buenos Aires, Argentinapp. 295-303.
- Semenas, L, Kreiter, A and Urbanski, J (2001). New cases of human diphyllbothriosis in Patagonia, Argentine. *Revista de Saúde Pública*. 35, pp. 214-216.
- Semenas, L and Úbeda, C (1997). Difilobotriasis humana en la Patagonia. *Revista de Saúde Pública*. 31, pp. 302-307.
- Sharp, G, Pike, A and Secombes, C (1992). Sequential development of the immune response in rainbow trout *Oncorhynchus mykiss* (Walbaum, 1792) to experimental plerocercoid infections of *Diphyllbothrium dendriticum* (Nitzsch, 1824). *Parasitol*. 104, pp. 169-178. doi: 10.1017/S0031182000060911.
- Sharp, GJ, Pike, AW and Secombes, CJ (1989). The immune response of wild rainbow trout, *Salmo gairdneri* Richardson, to naturally acquired plerocercoid infections of *Diphyllbothrium dendriticum* (Nitzsch, 1824). *J Fish Biol*. 35, pp. 781-794.

doi: 10.1111/j.1095-8649.1989.tb03029.x.

Sharp, G.JE, Secombes, C.J and Pike, A.W (1990). The laboratory maintenance of *Diphylobothrium dendriticum* (Nitzsch, 1824). *Parasitology*. 101, pp. 153-161.

Shin, HKyung, Roh, J-H, Oh, J-W, Ryu, J-S, Goo, Y-K, Chung, D-I et al. (2014). Extracorporeal Worm Extraction of *Diphylobothrium nihonkaiense* with Amidotrizoic Acid in a Child. *The Korean Journal of Parasitology*. 52, pp. 677-680. doi: 10.3347/kjp.2014.52.6.677.

Song, S-M, Yang, H-W, Jung, MKyu, Heo, J, Cho, CMin, Goo, Y-K et al. (2014). Two human cases of *Diphylobothrium nihonkaiense* infection in Korea. *The Korean Journal of Parasitology*. 52, pp. 197-199. doi: 10.3347/kjp.2014.52.2.197.

Song, T., Wang, W.-S., Zhou, B.-R., Mai, W.-W., Li, Z.-Z., Guo, H.-C. et al. (2007). CT and MR Characteristics of Cerebral Sparganosis. *American Journal of Neuroradiology*. 28, pp. 1700-1705. doi: 10.3174/ajnr.A0659.

Sreesai, S, Peapueng, P, Tippayamongkonkun, T and Sthiannopkao, S (2013). Assessment of a potential agricultural application of Bangkok-digested sewage sludge and finished compost products. *Waste Manag Res*. 9, pp. 925-936.

Stanciu, C, Trifan, A, Singeap, A-M, Sfarti, C, Cojocariu, C and Luca, M (2009). *Diphylobothrium latum* identified by capsule endoscopy-an unusual cause of iron-deficiency anemia.. *Journal of gastrointestinal and liver diseases : JGLD*. 18, pp. 142.

Suzuki, J, Murata, R, Sadamasu, K and Araki, J (2010). Detection and identification of *Diphylobothrium nihonkaiense* plerocercoids from wild Pacific salmon (*Oncorhynchus* spp.) in Japan.. *Journal of Helminthology*. 84, pp. 434-440. doi: 10.1017/s0022149x10000155.

Suzuki, N, Imamura, K, Kumazawa, H, Okamura, Y and Naragawa, Y (1988). Additional eight cases of diplogonoporiasis in Kochi prefecture. *Japanese Journal of Parasitology*. 16, pp. 285-291.

Suzuki, N, Imamura, K and Kumazawa, H (1993). Diplogonoporiasis in Kochi Prefecture between 1991-1992. *Japanese Journal of Parasitology*. 42, pp. 123-127.

Swiderski, Z and Mackiewicz, JS (2004). Ultrastructural studies on the cellular organisation of the coracidium of the cestode *Bothriocephalus clavibothrium* Ariola, 1899 (Pseudophyllidea, Bothriocephalidae). *Acta Parasitol*. 49, pp. 116-139.

Takenouchi, H, Katoaka, I, Yamamoto, S and Nagasaki, S (1980). Detection of parasite eggs in surplus sludges from night-soil disposal plants. Distribution survey of parasite eggs in Kochi Prefecture. *Japanese Journal of Parasitology*. 4, pp. 319-324.

Talapa, IA (1978). A study of the efficacy of small plants in decontaminating sewage waters from helminth ova. *Gigienicheskie Aspekty Okhrany Okruzhayushchei Sredy*. 6, pp. 101.

Tantalean, M (1975). Hallazgo de larvas plerocercoides de *Diphylobothriidae* Lühe, 1910 (Cestoda) en peces del mar peruano. *Bol Chile Parasitol*. 30, pp. 18-20.

Tantalean, M and Huiza, A (1994). Sinopsis de los parasitos de peces marinos de la costa peruana. *Biotempo*. 53, pp. 53-101.

Tappe, D, Berger, L, Haeupler, A, Muntau, B, Raez, P, Harder, Y et al. (2013). Case report: Molecular diagnosis of subcutaneous *Spirometra erinaceieuropaei* Sparganosis in a japanese Immigrant. *Am J Trop Med Hyg*. 88, pp. 198-202. doi: 10.4269/ajtmh.2012.12-0406.

Tarassow, W (1934). Das schwein und der Hund als endgültige Träger des *Diphylobothrium latum*. *Arch Schiffs-u. Tropenhyg*. 38, pp. 156-159.

Tavares, LER, Luque, JL and Bomfim, TCBD (2005). Human diphylobothriasis: reports from Rio de Janeiro, Brazil. *Rev Bras Parasitol*. 14, pp. 85-87.

- Thanchomnang, T, Tantrawatpan, C, Intapan, PM, Sanpool, O, Lulitanond, V, Tourtip, S et al. (2016). Rapid identification of nine species of diphyllbothriidean tapeworms by pyrosequencing. *Scientific Reports*. 6, pp. 37228. doi: 10.1038/srep37228.
- Torres, MARISA, Perez, CARLOS, Galdamez, ERCIRA, Gabor, MELVIN, Miranda, CLAUDIO, Cofre, XIMENA et al. (2001). Teniosis: Serie clínica en 35 pacientes. *Parasitología al día*. 25, pp. 55-59. doi: 10.4067/S0716-07202001000100010.
- Torres, P (1990). Primeros registros de endohelminthos parásitos en el salmon coho, *Oncorhynchus kisutch* (Walbaum), introducidos en Chile. *Archivos de Medicina Veterinaria*. 22, pp. 105-107.
- Torres, P (2013). Difilobotriasis. *Parasitología Humana*. (Apt, W, ed.). First edition pp. 202-213.
- Torres, P, Contreras, A, Cubillos, V, Gesche, W, Montefusco, A, Rebolledo, C et al. (1992). Parasitismo en peces, aves piscívoras y comunidades ribereñas de los lagos Yelcho y Tagua Tagua, Xa Región de Chile. *Arch Med Vet*. 24, pp. 77-92.
- Torres, P, Contreras, B, Figueroa, L, Franjola, R, González, H and Martin, R (1977). Investigaciones sobre Pseudophyllidea (Carus 1813) en el sur de Chile. I estudio preliminar sobre infección por plerocercoides de *Diphyllbothrium* sp. en *Salmo gairdneri* Richardson 1836 del lago calafquén (39°32'S, 78°09'O), Chile. *Boletín Chileno de Parasitología*. 32, pp. 73-80.
- Torres, P, Cubillos, V, Gesche, W, Rebolledo, C, Montefusco, A, Miranda, JC et al. (1991). Difilobotriasis en salmónidos introducidos en lagos del sur de Chile. Aspectos patológicos, relación con infección humana, animales domésticos y aves piscívoras. *Arch Med Vet*. 23, pp. 165-183.
- Torres, P, Cuevas, C, Tang, M, Barra, M, Franjola, R, Navarrete, N et al. (2004). Introduced and native fishes as infection foci of *Diphyllbothrium* spp. and humans and dogs from two localities at Lake Panguipulli in southern Chile. *Comp Parasitol*. 71, pp. 111-117.
- Torres, P, Figueroa, L and Franjola, R (1983). Pseudophyllidea in south of Chile. IX Plerocercoids in trouts from five lakes and new cases of *Diphyllbothrium latum* in man and *D. pacificum* in a dog. *International Journal for Zoonoses*. 10, pp. 15-21.
- Torres, P, Figueroa, L and Franjola, R (1981). Investigaciones sobre Pseudophyllidea (Carus, 1813) en el sur de Chile. VIII Desarrollo experimental de *Diphyllbothrium dendriticum* Nitzsch en *Larus maculipennis* Lichtenstein. *Boletín Chileno de Parasitología*. 36, pp. 74-75.
- Torres, P, Franjola, R, Figueroa, L, Schlatter, R, González, H, Contreras, B et al. (1981). Researches on Pseudophyllidea (Carus, 1813) in the south of Chile. IV: Occurrence of *Diphyllbothrium dendriticum* (Nitzsch). *J Helminthol*. 55, pp. 173-188.
- Torres, P, Franjola, R, Weitz, JC and Morales, G (1993). Registro de nuevos casos de difilobotriasis humana en Chile (1981-1992), incluido un caso de infección múltiple por *Diphyllbothrium latum*. *Boletín Chileno de Parasitología*. 48, pp. 39-43.
- Torres, P, Gesche, W, Montefusco, A, Miranda, JC, Dietz, P and Huijse, R (1998). Diphyllbothriosis humana y en peces del lago Riñihue, Chile: Efecto de la actividad educativa, distribución estacional y relación con sexo, talla y dieta de los peces.. *Arch Med Vet*. 30, pp. 31-45.
- Torres, P, Lopez, JC, Cubillos, V, Lobos, C and Silva, R (2002). Visceral diphyllbothriosis in a cultured rainbow trout, *Oncorhynchus mykiss* (Walbaum), in Chile. *Journal of Fish Diseases*. 25, pp. 375-379.
- Torres, P and Puga, S (2011). Comparative efficacy of candling and glass plate compression for detection of diphyllbothriosis in rainbow trout (*Oncorhynchus mykiss*) musculature. *Scientific and Technical Review of the Office International des Epizooties*. 30, pp. 931-937.
- Torres, P, Quintanilla, JC, Rozas, M, Miranda, P, Ibarra, R, San Martín, MF et al. (2010). Endohelminth parasites from salmonids in intensive culture from southern Chile. *J Parasitol*. 96, pp. 669-670.

- Torres, P, Ruiz, E, Rebolledo, C, Mira, A, Cubillos, V, Navarrete, N et al. (1990). Parasitismo en peces y comunidades humanas ribereñas de los lagos Huillinco y Natri (Isla Grande de Chiloé), Chile. *Bol Chil Parasitol.* 45, pp. 47-55.
- Torres, P, Torres, J, Garrido, O and Thibaut, J (1989). Investigaciones sobre Pseudophyllidea (Carus, 1813) en el sur de Chile. X Observaciones experimentales sobre la coexistencia de plerocercoides de *Dipyllobothrium latum* (L.) y *Diphyllbothrium dendriticum* (Nitzsch) en salmónidos de la cuenca del. *Arch Med Vet.* 21, pp. 51-57.
- Torres, P, Villalobos, L and Woelfl, S (2007). Experimental infection of copepods from four lakes in southern Chile with *Diphyllbothrium latum* (Linnaeus, 1758) coracidia. *Comp Parasitol.* 74, pp. 167-170.
- Torres, P, Villalobos, L, Woelfl, S and Puga, S (2004). Identification of the copepod intermediate host of the introduced broad fish tapeworm, *Diphyllbothrium latum*, in southern Chile. *The Journal of Parasitology.* 90, pp. 1190-1193.
- Torres, P, Franjola, R, Pérez, J, Auad, S, Uherek, F, Miranda, JC et al. (1989). Epidemiología de la difilobotriasis en la cuenca del río Valdivia, Chile. *Revista de Saúde Pública.* 23, pp. 45-57. doi: 10.1590/S0034-89101989000100007.
- Torres, P, Leyán, V and Puga, S (2012). Prevalence, intensity, and abundance of infection and pathogenesis caused by *Diphyllbothriosis* in vulnerable, native fish and introduced trout in Lake Panguipulli, Chile. *J Wildlife Dis.* 48, pp. 937-950. doi: 10.7589/2011-08-235.
- Tsuboi, T, Torii, M and Hirai, K (1993). Light and scanning electron microscopy of *Diphyllbothrium pacificum* expelled from a man. *Japanese Journal of Parasitology.* 42, pp. 422-428.
- Tung, C-C, Lin, J-W and Chou, F-F (2005). Sparganosis in male breast. *Journal of the Formosan Medical Association = Taiwan yi zhi.* 104, pp. 127-8.
- Unger, P and Palm, HW (2016). Parasitisation of sea trout (*Salmo trutta trutta* L.) from the spawning ground and German coastal waters off Mecklenburg-Western Pomerania, Baltic Sea. *Parasitology Research.* 115, pp. 165-174. doi: 10.1007/s00436-015-4732-4.
- Venturini, L (1989). El ciclo evolutivo experimental de *Diphyllbothrium erinaceieuropei* en *Paracyclops fimbriatus*, larvas de *Bufo arenarum* y caninos. *Rev Inst Med Trop Sao Paulo.* 31, pp. 308-312.
- Vik, R (1957). Studies on the helminth fauna of Norway I. Taxonomy and ecology of *Diphyllbothrium latum* (L.). *Nytt Magasin for Zoology.* 5, pp. 25-93.
- Vuylsteke, P, Bertrand, C, Verhoef, GEG and Vandenberghe, P (2004). Case of megaloblastic anemia caused by intestinal taeniasis. *Annals of hematology.* 83, pp. 487-488. doi: 10.1007/s00277-003-0839-2.
- Waeschenbach, A, Brabec, J, Scholz, T, Littlewood, D and Kuchta, R (2017). The catholic taste of broad tapeworms-multiple routes to human infection. *Int J Parasitol.* 47, pp. 831-843. doi: 10.1016/j.ijpara.2017.06.004. Epub 2017 Aug 3. PubMed.
- Wardle, R and McLeod, J (1968). *The zoology of tapeworms.* New York pp. 780.
- Weinbach, E and Garbus, J (1969). Mechanism of action of reagents that uncouple oxidative phosphorylation. *Nature.* 221, pp. 1016-1018.
- Wicht, B, Marval, F, Gottstein, B and Peduzzi, R (2008). Imported diphyllbothriasis in Switzerland: molecular evidence of *Diphyllbothrium dendriticum* (Nitsch, 1824). *Parasitol Res.* 102, pp. 201-204. doi: 10.1007/s00436-007-0745-y.
- Wicht, B, Scholz, T, Peduzzi, R and Kuchta, R (2008). First record of human infection with the tapeworm *Diphyllbothrium nihonkaiense* in North America. *American Journal of Tropical Medicine and Hygiene.* 78, pp. 235-238.
- Wicht, B., Gustinelli, A., Fioravanti, M.L, Invernizzi, S. and Peduzzi, R. (2009). Prevalence of the broad tapeworm *Diphyllbothrium latum* in perch (*Perca fluviatilis*) and analysis of abiotic factors influencing its occurrence in Lake Lario (Como, Italy). *Bulletin of the European Association of Fish Pathologists.* 29, pp. 58-65.
- Wicht, B, de Marval, F and Peduzzi, R (2007). *Diphyllbothrium nihonkaiense* (Yamane et al., 1986) in Switzerland: First

molecular evidence and case reports. *Parasitology International*. 56, pp. 195–199. doi: 10.1016/j.parint.2007.02.002.

Wilhelm, O (1958). Pseudophyllidea (Diphyllbothrium y Diplogonoporus) en Chile. *Bol Soc Biol Concepción Chile*. 33, pp. 125–130.

Wiwanitkit, V (2005). A review of human sparganosis in Thailand. *Int J Infect Dis*. 9, pp. 312–316.

Wootten, R and Smith, JW (1979). The occurrence of plerocercoids of *Diphyllbothrium* spp. in wild and cultured salmonids from The Loch Awe area. *Scottish Fisheries Research Report*. 13, pp. 1–8.

Wright, M and Curtis, M (2000). Temperature effects on embryonic development and the life cycle of *Diphyllbothrium dendriticum*. *Int J Parasitol*. 30, pp. 849–852. doi: 10.1016/S0020-7519(00)00061-8.

Yamane, Y (1968). On the fine structure of *Diphyllbothrium erinacei* with special reference to the tegument. *Yonago Acta Med*. 12, pp. 169–181.

Yamane, Y, Bylund, G, Abe, K, Osaki, Y and Okamoto, T (1989). Scanning electron microscopic study of four *Diphyllbothrium* species. *Parasitology Research*. 75, pp. 238–244.

Yamane, Y, Shiwaku, K, Abe, K, Osaki, Y and Okamoto, T (1989). The taxonomic differences of embrionic hooks in *Diphyllbothrium nihonkaiense*, *D. latum* and *D. dendriticum*. *Parasitology Research*. 75, pp. 549–553.

Yamane, Y, Kamo, H, Bylund, G and Wikgren, BJungar P (1986). *Diphyllbothrium nihonkaiense* sp. nov. (Cestoda: diphyllbothriidae) - revised identification of japanese broad tapeworm. *Shimane J Med Sci*. 10, pp. 28–48.

Yamasaki, H, Ohmae, H and Kuramochi, T (2012). Complete mitochondrial genomes of *Diplogonoporus balaenopterae* and *Diplogonoporus grandis* (Cestoda: Diphyllbothriidae) and clarification of their taxonomic relationships. *Parasitology International*. 61, pp. 260–266. doi: 10.1016/j.parint.2011.10.007.

Yamasaki, H, Kumazawa, H, Sekikawa, Y, Oda, R, Hongo, I, Tsuchida, T et al. (2016). First confirmed human case of *Diphyllbothrium stemmacephalum* infection and molecular verification of the synonymy of *Diphyllbothrium yonagoense* with *D. stemmacephalum* (Cestoda: Diphyllbothriidae). *Parasitology international*. 65, pp. 412–421. doi: 10.1016/j.parint.2016.06.003.

Yamasaki, H and Kuramochi, T (2009). A case of *Diphyllbothrium nihonkaiense* infection possibly linked to salmon consumption in New Zealand. *Parasitol Res*. 105, pp. 583–586. doi: 10.1007/s00436-009-1468-z.

Yanagida, T, Matsuoka, H, Kanai, T, Nakao, M and Ito, A (2010). Anomalous segmentation of *Diphyllbothrium nihonkaiense*. *Parasitol Int*. 59, pp. 268–270. doi: 10.1016/j.parint.2009.12.006.

Yazaki, S, Kamo, H, Kawasaki, H and Yamane, Y (1984). Experimental Infection to Japanese men with *Diphyllbothrium latum* (Linné, 1758) from Finland. *Yonago Acta Medica*. 27, pp. 19–30.

Yera, H, Nicolaud, J and Dupouy-Camet, J (Submitted). Use of nuclear and mitochondrial DNA PCR sequencing for molecular identification of *Diphyllbothrium* isolates potentially infective for humans. *Parasite*. 15, pp. 402–407. doi: 10.3947/ic2013.45.1.32. Epub2013 Mar 29. Review. PMID: 24265948..

Yera, H, Estran, C, Delaunay, P, Martine, G-T, Dupouy-Camet, J and Marty, P (2006). Putative *Diphyllbothrium nihonkaiense* acquired from a Pacific salmon (*Oncorhynchus keta*) eaten in France; genomic identification and case report. *Parasitol Int*. 55, pp. 45–49. doi: 10.1016/j.parint.2005.09.004.

Yera, H, Kuchta, R, Brabec, J, Peyron, F and Dupouy-Camet, J (2013). First identification of eggs of the Asian fish tapeworm *Bothriocephalus acheilognathi* (Cestoda: Bothriocephalidae) in human stool. *Parasitology international*. 62, pp. 268–271. doi: 10.1016/j.parint.2013.02.001.

Yildirimhan, H, Bursey, C and Goldberg, S (2007). Helminth parasites of the grass snake, *Natrix natrix*, and the dice snake, *Natrix tessellata* (Serpentea: colubrida) from Turkey. *Comp Parasitol*. 74, pp. 343–354.

- Zamudio-Perez, E, Rojas-Valencia, N, Chairez, I and Torres, LG (2013). Coliforms and helminths eggs removals by coagulation-flocculation treatment based on natural polymers. *Journal of Water Resource and Protection*. 5, pp. 1027-1036.
- Zhang, M, Cui, FY, Liu, DM, He, WJ and Han, HD (2009). Removing efficiency study on Cyclops cooperating with water treatment process by alternative oxidants. *Huan Jing Ke Xue*. 30, pp. 3568-3572.
- Zhang, W, Che, F, Tian, S, Shu, J and Zhang, X (2015). Molecular Identification of *Diphyllbothrium nihonkaiense* from 3 Human Cases in Heilongjiang Province with a Brief Literature Review in China.. *The Korean journal of parasitology*. 53, pp. 683-688. doi: 10.3347/kjp.2015.53.6.683.
- Zhao, ZW, Cui, FY, Lin, T and Liu, GP (2007). Inactivation and removal of chlorine dioxide on cyclops of zooplankton. *Huan Jing Ke Xue*. 28, pp. 1759-1762.
- Zhou, BJ, Wang, WL and Lei, L (2005). Prevalence of *Sparganosis mansoni* in Yunnan province, China. *J Trop Med*. 5, pp. 207-208.
- Zhu, X, Beveridge, I, Berger, L, Barton, D and Gasser, R (2002). Single-strand conformation polymorphism-based analysis reveals genetic variation within *Spirometra erinacei* (Cestoda: Pseudophyllidea) from Australia. *Mol Cell Probes*. 16, pp. 159-165. doi: 10.1006/mcpr.2002.0406.
- von Bonsdorff, B. (1977). *Diphyllbothriasis in man*. New York pp. 189.
- von Bonsdorff, B and Bylund, G (1982). The ecology of *Diphyllbothrium latum*. *Ecology of Diseases*. 1, pp. 26-26.