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Effects of anisakid nematodes *Anisakis simplex* (s.l.), *Pseudoterranova decipiens* (s.l.) and *Contracaecum osculatum* (s.l.) on fish and consumer health☆

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The anisakid nematodes *Anisakis simplex* (Rudolphi, 1809), *Pseudoterranova decipiens* (Krabbe, 1878) and *Contracaecum osculatum* (Rudolphi, 1802) occur as third-stage larvae in marine fish products and may infect consumers ingesting raw or under-cooked fish products. Clinical symptoms associated with the infection, termed anisakidosis, vary from irritation of the oesophagus and stomach, via nausea, vomiting and diarrhoea to severe epigastric and abdominal pain. Third-stage larvae of *A. simplex* are found in the body cavity, musculature and various organs, *P. decipiens* occur mainly in the fish musculature (fillet) and *C. osculatum* larvae reside predominantly in the liver, body cavity, mesenteries and pyloric caeca. Preventive measures, including mechanical removal of worms, heat treatment or freezing to kill worms, are needed in order to reduce the risk of human infections. The anisakid life cycle involves several hosts. *A. simplex* nematodes use cetaceans (whales) as final hosts whereas *P. decipiens* and *C. osculatum* have their adult stage in pinnipeds (seals). Eggs released by worms in these hosts pass with feces to seawater where free-living third-stage larvae hatch from the eggs. Various invertebrates – including euphausiids, copepods and amphipods – feed on these larvae, become infected and serve as intermediate hosts. A range of fish species may serve as transport hosts following ingestion of infected invertebrates and the final stage develops after two additional moults in the stomach of marine mammals which consumed infected fish. Control measures may be implemented to reduce infections of fish stocks and thereby risk of human infections.

Contents

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1. Introduction

Fishborne larval nematodes belonging to the family Anisakidae are widespread in fish populations worldwide (Nadler et al., 2005; Mattiucci and Nascetti, 2008; Karpiej et al., 2013). Representatives from the three anisakid genera *Anisakis, Pseudoterranova* and *Contracaecum* apply marine mammals as final host, invertebrates as intermediate host and fish as transport host (Køie and Fagerholm, 1995; Køie et al., 1995). The infective third stage larva in fish is able to infect consumers ingesting raw or undercooked fish products which may elicit severe clinical symptoms (Ishikura et al., 1993). The disease is termed anisakidosis referring to the causative agent belonging to the family anisakidae. In order to differentiate infections caused by worms from the different genera the term anisakiosis is used for infections by *Anisakis* spp. (Ishikura, 2003), the term pseudoterranovosis is applied for infections caused by *Pseudoterranova* spp. (Margolis, 1977) and finally infections due to *Contracaecum* spp. should accordingly be named contracaecosis. Each of the three genera is comprised of several sibling species (Mattiucci et al., 2014; Timi et al., 2014) but in this review the focus has been placed on *Anisakis simplex* (s.l.), *Pseudoterranova decipiens* (s.l.) and *Contracaecum osculatum* (s.l.).

2. Morphology and diagnosis

Macroscopic inspection of the anisakid larvae may reveal some minor differences between the parasites. *A. simplex* larvae are whitish to transparent (Fig. 1A), *P. decipiens* larvae are reddish to brownish (Fig. 1B) and *C. osculatum* larvae appear transparent via greyish to brownish (Fig. 1C). Specific diagnosis is not possible by macroscopic inspection or light microscopy but the three worm types can be differentiated by simple light microscopic examination of cephalic structures (booring tooth, excretory pore location), caudal end (tapering or rounded with mucron spine) and gastrointestinal elements (presence or not of intestinal caecum and ventricular appendix) (Fagerholm, 1982; Mattiucci et al., 1998; Quiazon et al., 2009). Specific diagnosis must rely on further molecular investigations. Allozyme markers are valuable for differentiating between *A. simplex, P. decipiens* and *C. osculatum* (Mattiucci et al., 1998; Mattiucci and Nascetti, 2007, 2008) but sequencing of genomic DNA encoding ribosomal RNA (18S, ITS1, 5.8S, ITS2, 28S) has proven useful for further species confirmation (Zhu et al., 2007; Jabbar et al., 2013; Mattiucci et al., 2014). The sequencing of the mitochondrial genome of *A. simplex* (Kim et al., 2006) allowed more specific diagnosis. The application of mitochondrial markers (cox1 and cox2) has shown to be valuable for further taxonomic resolution and differentiation of sibling species (Mohandas et al., 2014; Mattiucci et al., 2014; Liu et al., 2015).

3. Life cycle

A. simplex, with the vernacular name herring worm (or whale worm), obtain the adult stage in the stomach of cetaceans where copulation and oviposition take place (Fig. 2A). Eggs pass with feces to the marine environment where eggs hatch and

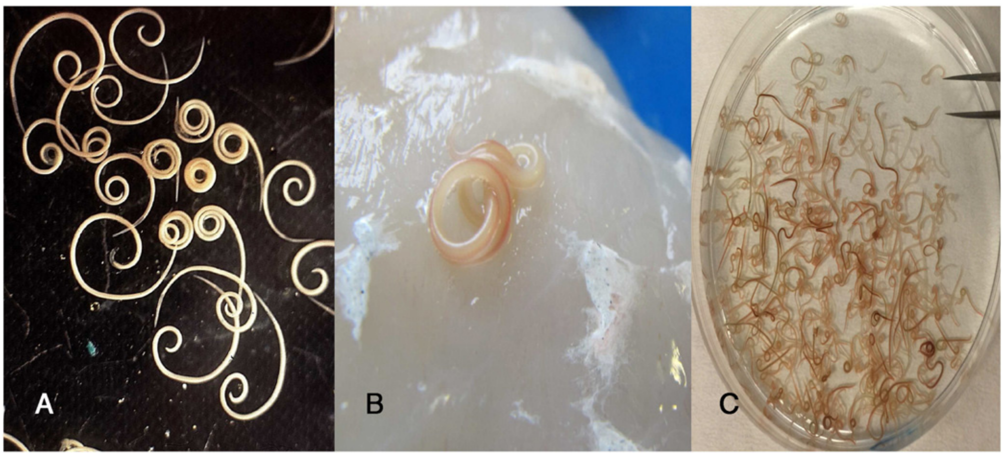

Fig. 1. A. *Anisakis simplex* (herring worm, whale worm) third stage larvae (30 mm total length) recovered from the body cavity of an Atlantic herring *Clupea harengus* body. B. *Pseudoterranova decipiens* (cod worm, seal worm) third stage larva (45 mm total length) escaping from encapsulation in fillet of Baltic cod (*Gadus morhua*). C. *Contracaecum osculatum* third stage larvae (20–30 mm total length) removed by artificial digestion from the liver of a Baltic cod (*Gadus morhua*).

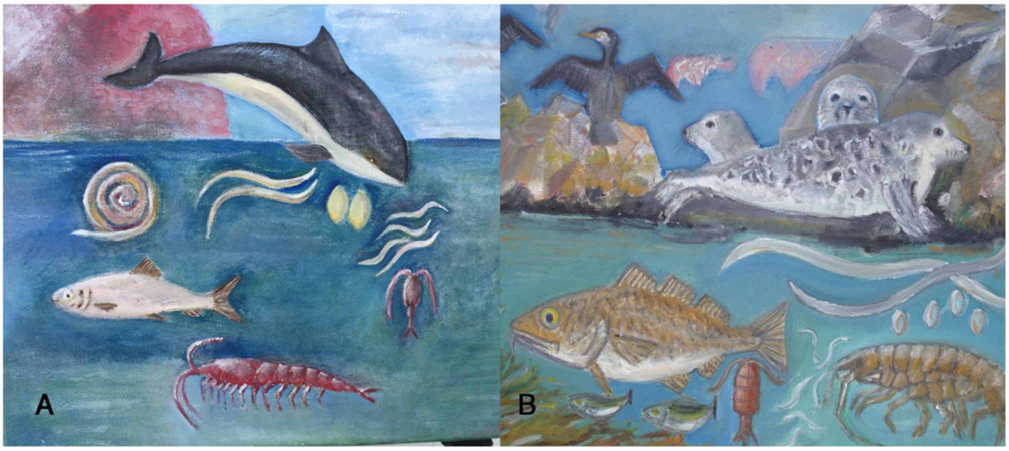

Fig. 2. A. Life cycle of *Anisakis simplex* with the adult worms in the stomach/intestine of cetaceans and release of worm eggs with host feces to the marine environment. Third stage larvae are released from eggs by hatching, and invertebrates, such as copepods and euphausiids, become infected by ingesting the larvae. Teleosts, acting as transport hosts following ingestion of infected invertebrate hosts, transmit larvae to cetaceans, in which the larvae develop to the adult stage. B. Life cycle of both *Pseudoterranova decipiens* and *Contracaecum osculatum*. The adult worm stage is found in the seal's stomach; released eggs are passed with feces into the marine environment. Third stage larvae hatch from eggs and are ingested by invertebrates (copepods, amphipods) which are subsequently ingested by teleosts to act as transport hosts. Adult stage develop in seals which ingest infected teleosts.

liberate the third stage larvae (Højgaard, 1998). A range of invertebrates may act as the first intermediate host but euphausiaceans and copepods are particularly important (Smith and Wootten, 1978; Køie et al., 1995). Fish achieve infection by ingesting infected invertebrates or smaller infected fish.

P. decipiens, also termed codworm (or seal worm), apply seals as final host (Jensen and Idås, 1992; Aspholm et al., 1995; Ólafsdóttir and Hauksson, 1997, 1998) and a range of invertebrates, including crustaceans, serve as transport hosts for these worm larvae (Køie et al., 1995; McClelland, 2002) (Fig. 2B). Fish obtain infection during feeding on these invertebrates. Larger fish may also gain infection through feeding on infected smaller fish.

C. osculatum has been given the vernacular name "liver worm" based on a rather prevalent occurrence in livers of Baltic cod (Mehrdana et al., 2014) but it may be found in other organs as well. The species uses seals as the final host (Fig. 2B) (Valtonen et al., 1988; Skrzypczak et al., 2014; Lunneryd et al., 2015), invertebrates as intermediate host (Køie and Fagerholm, 1995) and several fish species as transport hosts (Fagerholm, 1982; Perdiguero-Alonso et al., 2008; Haarder et al., 2014). Field observations from the Baltic sea have indicated that copepods act as first intermediate hosts of *C. osculatum*, sprat feeding on copepods act as transport host and cod, eating quantities of sprat, may accumulate significant parasite burdens (Zuo et al., 2016).

For all three anisakid species the life cycle includes two final moults in the stomach of the mammalian host following their ingestion of infected fish or invertebrates carrying third stage larvae.

4. Anisakids and fish health

The anisakid third stage larvae are not strictly host specific and a wide range of marine teleost species may be found infected in most waters ranging from the Atlantic via the Mediterranean and the Pacific to the Antarctic area (Adroher et al., 1996; Mattiucci et al., 1998; McClelland and Martell, 2001). Third stage larvae of anisakid worms are mainly ingested by fish when they predate on the crustacean intermediate host or teleostean transport hosts. In the fish stomach larvae are activated and penetrate the stomach wall in order to seek residence in the peritoneal cavity, musculature or organs such as liver (Levsen and Lunestad, 2010; Mehrdana et al., 2014). Severe inflammatory reactions with tissue deformation, including marked cellular infiltration in the stomach wall and mucosa, of Atlantic cod has been associated with numerous penetrating *A. simplex* larvae (Levsen and Berland, 2012) and the term "stomach crater syndrome" was applied for this pathological reaction. Atlantic salmon and sea trout returning to rivers in Scotland, England and Wales were found infected with a high number of *A. simplex* larvae causing a "red vent syndrome", characterized by haemorrhages and inflammation around the vent (Beck et al., 2008; Noguera et al., 2009). This species clearly provokes an inflammatory reaction in salmonids (Haarder et al., 2013) which first is seen as attraction of inflammatory cells and partly encapsulation of the worm (Fig. 3A). Likewise, *P. decipiens* become encapsulated by host cells in cod muscle tissue (Fig. 3B) and *C. osculatum* in cod's livers (Fig. 3C and 3D) and it is believed that the mere presence of larvae in the tissues affect the normal function of these organs. *A. simplex* infections may be associated with the loss of condition of fish hosts, but in cases where the larvae are sequestered outside essential organs the effect may be less harmful. *A. simplex* liver infections in North Atlantic cod can be severe (Fig. 4A) but the effect on liver condition of the host may not be as devastating as expected from the worm load. Thus, a large part of the parasite's infrapopulation is located in an encapsulated state on the surface of the organs which may be observed when worms are removed by peeling off the surface layer of cod's livers. Anisakid nematode larvae such as *P. decipiens* and *C. osculatum* may affect the physiological state, health and survival of the host. Codworm infection of the fish muscle reduces swimming performance in smelt and eel which can lead to increased mortality in the wild

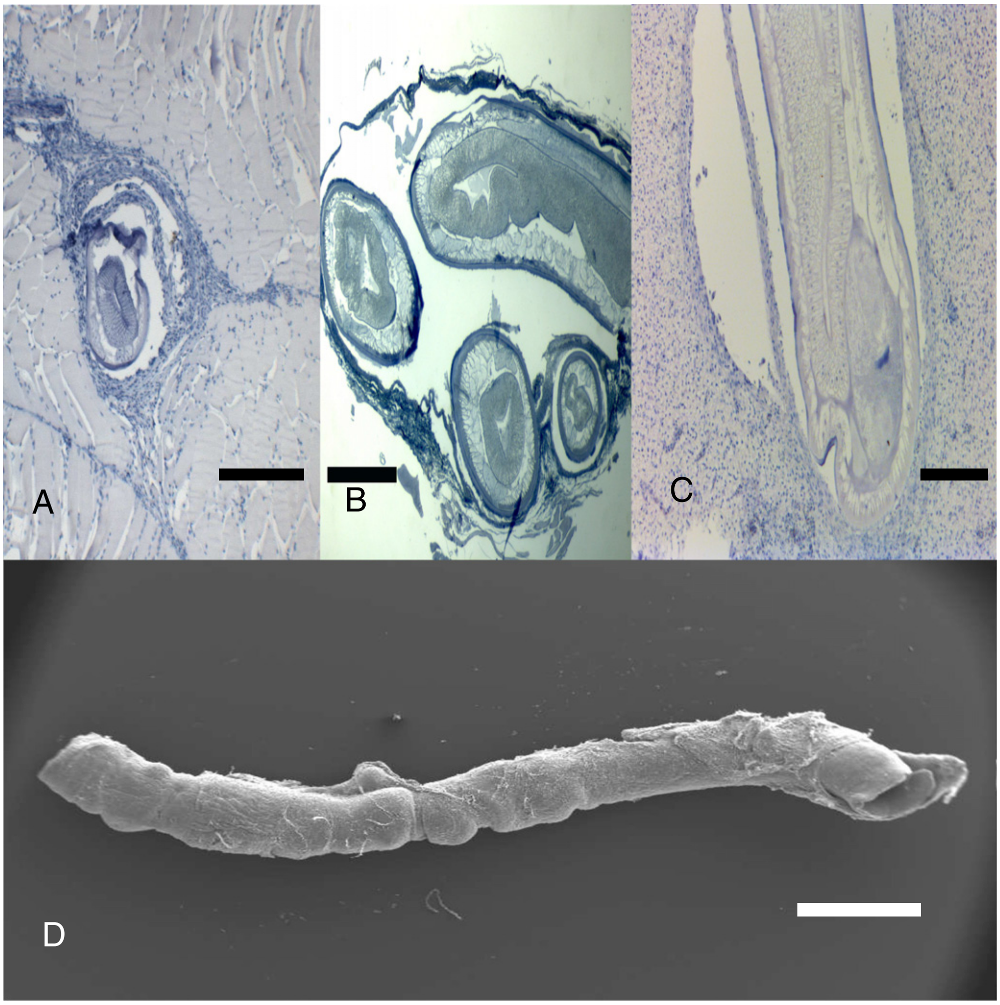

Fig. 3. A. Histological section of third stage larva of *Anisakis simplex* in infected liver tissue of Atlantic salmon, *Salmo salar*. A tissue reaction encapsulates the worm (scale bar 0.5 mm). B. Histological section of third stage larva of *Pseudoterranova decipiens* in the musculature of Baltic cod, *Gadus morhua*. Host cells encapsulate the larva (scale bar 1 mm). C. Histological section of third stage larva of *Contracaecum osculatum* invading the liver of a Baltic cod, *Gadus morhua*, with marked encapsulation by host cells (scale bar 1 mm). Scanning electron microscopy of a tube shaped encapsulated material (host cells from the liver of a Baltic cod) after removal of *Contracaecum osculatum* third stage larva (scale bar 2 mm).

(Sprengel and Lüchtenberg, 1991; Rohlwing et al., 1998). Excretions from *P. decipiens* contain several pentanols and pentanons and it has been suggested that these compounds act as local anaesthetics in the cod muscle during worm penetration (Ackman and Gjelstad, 1975) and effects on muscle contractility may therefore be expected. Reduced swimming abilities of a fish host will ease predation by marine mammals, including seals, whereby this pathogenicity factor will optimize the life cycle of anisakids. Decreased body mass indices were recorded in Antarctic ice-fishes carrying high burdens of *C. osculatum* larvae which may indicate a parasite induced host effect (Santoro et al., 2013). In recent years Danish and Polish investigators (Buchmann and Kania, 2012; Mehrdana et al., 2014; Horbowy et al., 2016) have documented a marked increase of *P. decipiens* and *C. osculatum* infections of Baltic cod when compared to studies in the 1970s and 1980s (Möller, 1975; Thulin et al., 1989; Myjak et al., 1994) when seal abundance was low. The cod living in the Southern Baltic area may be infected by up to several hundred third stage larvae of *C. osculatum* (Haarder et al., 2014; Mehrdana et al., 2014; Nadolna and Podolska, 2014) which may challenge integrity and function of this key organ and affect growth and the nutritional state (Mehrdana et al., 2014; Zuo et al., 2016). Although the tissue disturbance by *C. osculatum* larvae is significant, even in a relatively well nourished cod liver (Fig. 4B), the impact is more prominent when liver size has decreased (Fig. 4C). Such a negative association was already claimed by Petrushevski and Shulman (1955) studying *C. osculatum* infection of Baltic cod in the 1940s and 1950s. A negative association between high parasite loads and the fish population size was noted by Eero et al., 2015 demonstrating reduced abundance of larger cod, concomitant with increasing worm occurrence, despite successful recruitment of young cod. This notion was further substantiated by Zuo et al. (2016) who showed that young Baltic cod with a body length below 30 cm were largely uninfected whereas cod larger than 30 cm harbored severe worm burdens. It is nevertheless difficult from field data to separate the potential

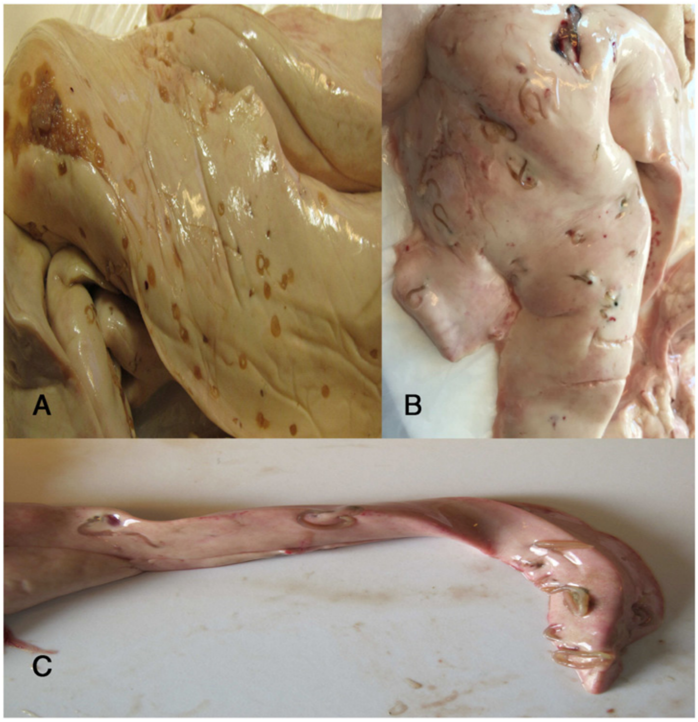

Fig. 4. A. North Atlantic cod (*Gadus morhua*) liver heavily infected with encapsulated third stage larvae of *Anisakis simplex* on the organ surface. B. Relatively well nourished liver of Baltic cod infected with third stage larvae of *Contracaecum osculatum* which are encapsulated inside the liver tissue. Only parts of the worms are visible on the surface. C. Small nutritionally deprived liver from a Baltic cod heavily infected with encapsulated third stage larvae of *Contracaecum osculatum*.

parasite-related effects on condition and liver indices from more manageable impacts such as feed availability (Buchmann and Børresen, 1988). Therefore controlled laboratory trials must be performed in order to confirm this phenomenon.

5. Anisakids and fish product quality

Third stage larvae of the three anisakid species affect the fish product quality differently. Larvae of *A. simplex* reach 10–30 mm in total length, a width less than 1 mm, are whitish to transparent and are not easily detected by the naked eye when they reside deeply embedded in fish fillets. When occurring in high numbers in the body cavity or on the liver surface of cod it can be detected but many of the worms can be removed before industrial processing. This can be achieved simply by peeling off the liver capsule with associated worm larvae which significantly reduce the parasite burden. Larvae in the fillets need further efforts for removal and it should be noted that allergens from *A. simplex* still may be present in the products after removal or killing of intact worms (Audicana et al., 1997). *P. decipiens* larvae are longer and may reach a total length of 4–5 cm and a width of 1–2 mm. It is often reddish or brownish in color, and in thin fish fillets worms can be detected by the naked eye although some worms can be over-looked in thicker fillets. The industry should apply candling and mechanical trimming of fish products in order to remove worms from the fish fillet which may otherwise be discarded by consumers (McClelland, 2002; Levsen et al., 2005). Cod activates a strong cellular host reaction when *P. decipiens* larvae penetrate the musculature and the reaction immobilizes the larva in an encapsulated stage which protects the live worm but also adds to the disturbance of the fillet structure. *C. osculatum* third stage larva is semitransparent to greyish with a length of 2–3 cm and a width of approximately 1 mm or less. In Baltic cod livers only a part of the worm is visible as the main part of the parasite is embedded in the tissue under the liver surface. Cellular host reactions encapsulate the larva in a thick layer of host cells leaving the worm in an inactive stage which may last for at least three years. When occurring in high numbers in relatively small livers the organ is left useless for the industry. Surface peeling of the liver is not an option as the *C. osculatum* larvae do not attach to the liver epithelium but mechanical, chemical and enzymatic treatment of the entire liver may be a possible worm extraction method.

6. Zoonotic potential

Species within the three genera are also known to cause anisakidosis in humans upon accidental ingestion of live third stage larvae in unprocessed sea food. The main part of recorded anisakidosis cases are caused by *A. simplex* but also *P. decipiens* and *C. osculatum* are being recognized as responsible for severe gastrointestinal infections of man (Table 1). A. *simplex* is responsible for the majority of anisakiosis cases (Ishikura et al., 1993) but *A. pegreffi* has also shown its human-pathogenic potential

Table 1
Geographic distribution and genus of the causative pathogen in anisakidosis cases.

Location	Genus of worm causing anisakidosis			References
	Anisakis	*Pseudoterranova*	*Contracaecum*	
Australia	−	−	+	Shamsi and Butcher, 2011
Canada	+	−	−	Couture et al., 2003
Chile	−	+	−	Mercado et al., 2001; Torres et al., 2007
Croatia	+	−	−	Mladineo et al., 2014
Denmark	+	−	−	Andreassen and Jörring, 1970
France	+	−	−	Bourree et al., 1995
Germany	+	−	+	Schaum and Müller, 1967; Möller and Schröder, 1987
Holland	+	−	−	Van Thiel et al., 1960
Iceland	−	+	−	Skirnisson, 2006
Italy	+	−	−	Fumarola et al., 2009
Japan	+	+	+	Kagei and Isogaki, 1992; Nagasawa, 2012
Korea	+	+	−	Yu et al., 2001; Choi et al., 2009; Na et al., 2013
Norway	+	−	−	Lin et al., 2014
Spain	+	−	−	Repiso et al., 2003; Puente et al., 2008;
South Africa	+	−	−	Nieuwenhuizen et al., 2006
Taiwan	+	−	−	Li et al., 2015
USA	+	+	−	Pinkus et al., 1975; Amin et al., 2000

(Fumarola et al., 2009; Nascetti, 2011). Numerous investigations of clinical cases have documented the pathogenicity of the anisakid worm larvae in man which has called for further investigations using experimental animals in order to further characterize pathogenesis. Rats, rabbits, dogs and pigs may be applied for experimental infections and studies have shown that *A. simplex* (Desowitz, 1986), *P. decipiens* and *C. osculatum* (Strøm et al., 2015) penetrate the mucosa of the gastro-intestinal tract. This can elicit eosinophilic granulomatous formation corresponding to reactions reported to be associated with human infections. *A. simplex* larvae penetrating the stomach wall, the intestine or performing extra-intestinal migrations in humans (Testini et al., 2003), may elicit severe clinical symptoms including epigastric pain, vomiting, diarrhoea and nausea (Kagei and Isogaki, 1992; Ishikura et al., 1993; Caramello et al., 2003). Systemic reactions include eosinophilia and IgE titer increase (Perteguer et al., 2000; Valiñas et al., 2001) and the IgE titer measurements can be used for screening of infection rates in populations (Lin et al., 2014; Mladineo et al., 2014). *P. decipiens* larvae will only in rare cases perform severe extra-intestinal migrations in the human accidental host (Little and MacPhail, 1972; Amin et al., 2000) but the problems associated with ingestion of raw infected fish meat must be framed because symptoms associated with even light infections of non-penetrating worms include diarrhoea, gastric pain, tickling feelings, vomiting and coughing from the gastro-intestinal system (Little and Most, 1973; Pinel et al., 1996; Skirnisson, 2006). Relatively few human cases of *C. osculatum* larvae causing anisakidosis have been reported but it cannot be excluded that it is under-reported due to misidentification of the causative agent. Symptoms reported include gastro-intestinal pain, vomiting, diarrhoea and nausea. In one Australian case the worm was expelled spontaneously with bowel motion after 5–6 weeks disease history whereafter symptoms resided (Shamsi and Butcher, 2011). Two cases have been reported from Japan (Nagasawa, 2012) and one from the Baltic area in Europe (Schaum and Müller, 1967). The latter case involved both surgery and subsequent anthelmintic treatment in order to cure the patient.

7. Allergy induced by anisakid worms

The symptoms, clinical manifestations and immunological reactions in anisakidosis patients suggest that allergic reactions at least partly are involved in the disease development (Daschner and Pascual, 2005; Daschner et al., 2005; Choi et al., 2009). The mechanical impact of penetrating larvae on inner organs may cause some symptoms but increase of serum IgE titers, eosinophilia (systemic and local), skin symptoms, urticaria, airway obstruction and even anaphylactic reactions call for additional explanations (Caballero and Moneo, 2002; Nieuwenhuizen et al., 2006). A range of allergens have been isolated from *A. simplex* and these can cause disease in sensitized patients even without exposure to the live worm (Caballero et al., 2011; Arcos et al., 2014; Fæste et al., 2014). Thus, ingestion of processed fish products with heat and frost stable worm allergens have been reported to elicit allergic reactions in consumers (Audicana et al., 1997) but it is generally believed that an active infection with a live penetrating *Anisakis* larva is needed to sensitize and establish the allergic condition in patients (Alonso-Gómez et al., 2004). Subsequent exposure to *Anisakis* allergens (with or without live or dead worms) may then be sufficient to elicit allergic reactions. It is worth noting that several of the antigens are stable towards freezing, heating, pepsin exposure and autoclaving (Kobayashi et al., 2007; Caballero et al., 2008; Carballeda-Sangiao et al., 2014). Allergens released from *P. decipiens* and *C. osculatum* have not been described in detail but due to biological similarities to *A. simplex* it cannot be ruled out that the two species release compounds with a allergenic potential.

8. Geographic distribution of anisakidosis

Anisakidosis cases have primarily been recorded in countries where consumption of undercooked fish is common. Anisakiosis cases are commonly reported in Japan (Oshima, 1987; Suzuki et al., 2010) but infections are also known from Korea, Taiwan, Europe (Denmark, Norway, Germany, Holland, France, Italy, Spain, Croatia), North America (USA and Canada) and South Africa (Table 1). Most pseudoterranovosis cases have been reported from Japan (Sawada et al., 1983; Ishikura et al., 1993), Korea (Koh et al., 1999; Yu et al., 2001; Na et al., 2013), Iceland (Skirnisson, 2006), North America (Hitchcock, 1950; Chitwood, 1975; Kliks, 1983), South America (Mercado et al., 2001; Torres et al., 2007) but changing eating habits in most countries (involving dishes such as ceviche, sushi, sashimi and corresponding preparations) may expose consumers globally to infection. Human infections due to *C. osculatum* were reported from Germany (Schaum and Müller, 1967), Australia (Shamsi and Butcher, 2011) and Japan (Nagasawa, 2012) but due to the widespread and global occurrence of the species in a range of teleost species attention should be given to new cases of contracaecosis.

9. Treatment

Infections with anisakid third stage larvae may in some cases progress without symptoms and patients may be cured spontaneously. In other cases severe symptoms due to larval penetration of host tissues are evident and patients have been hospitalized and subjected to medical examination. By use of ultrasonography (Ido et al., 1998), fiber-gastroscopic and endoscopic equipment (Matsumoto et al., 1992) some worms may be localized in situ whereby immediate removal of the pathogen can be conducted. In other cases surgery involving resection of the affected organ area has shown necessary for diagnosis and treatment (Bourree et al., 1995; Pampiglione et al., 2002; Repiso et al., 2003). Treatment with anthelmintics may be a way for elimination of the causative agents. Thus, application of albendazole has been advocated for *Anisakis* infections in humans (Moore et al., 2002; Pacios et al., 2005) and thiabendazole was used successfully against a human *C. osculatum* infection (Schaum and Müller, 1967) when symptoms re-occurred after surgical removal of some worm larvae. A series of screenings have suggested that also herbal drugs may affect survival of anisakids. Thus, monoterpenes (Hierro et al., 2004, 2006), geraniol, citronellal (Barros et al., 2009) and *Matricaria chamomilla* oils (Romero et al., 2012) have all shown in vitro effects but require clinical trials for confirmation.

10. Prevention by food processing

The risk of contracting anisakidosis following ingestion of raw or semi-raw seafood products (Gardiner, 1990; Couture et al., 2003; Puente et al., 2008) have been known since the 1950s when the first case of anisakiosis was detected in the Netherlands (Van Thiel et al., 1960). Later this zoonotic problem was recognized in other countries including Japan where eating habits including raw fish dishes increased risk of infection (Oshima, 1987; Ishikura et al., 1993). Research focused subsequently on how worms could be rendered inactive and non-infective. Heating to 60 °C for minutes and freezing to −20 °C for 24 h were methods which were shown to kill worm larvae and recommendations for pre-treatment of fish products were published (Wharton and Aalders, 2002). Freezing regulations were then implemented in Holland and since then in the EU (EFSA, 2010). It is also known that prolonged salting at high NaCl concentrations will kill anisakid larvae but a series of investigations have demonstrated that vinegar and salt used in marinating procedures do not readily inactivate worm larvae as this process may take weeks (Karl et al., 1994). Therefore the recommendations for marine food products as stated above should be followed. Quality assurance and worm content status of fish products should be performed regularly by several methods. Test of product samples can be conducted by full artificial digestion in a pepsin and hydrochloric acid solution at 37 °C or by mechanical compression of fish products in order to record the worm content status. Candling on light tables may be used by not all anisakid larvae may be recovered by this method (Levsen et al., 2005) although the methodology has been improved (Yang et al., 2013). *A. simplex* and *P. decipiens* can be detected due to fluorescence if the fish fillet is compressed, frozen and subsequently illuminated by UV-light (Karl and Leinemann, 1993) but this does not apply for *C. osculatum* in cod liver tissue (Zuo et al., 2016). Several immune-chemical and molecular techniques have been developed for detection of anisakids in fish products. Thus, enzyme linked immunosorbent analysis (ELISA) (Xu et al., 2010; Werner et al., 2011), restriction fragment length polymorphism following PCR (RFLP-PCR) (Espiñeira et al., 2010), realtime PCR (Lopez and Pardo, 2010; Herrero et al., 2011) may be used by the industry to secure the status of their products.

11. Prevention by management

If a parasite-free status of certain sea-food types can be documented the risk of anisakidosis should be significantly reduced. This may allow for the lifting of freezing regulations which are currently needed to inactivate anisakids. Aquacultured fish such as marine-cultured rainbow trout, Atlantic salmon, sea bass, sea bream and turbot, which have been raised in isolation from infected prey organisms and have been fed exclusively heat treated and parasite-free feed pellets, are eligible through adequate documentation for parasite-free status (EFSA, 2010). However, one of the reasons that the problem with worm infected fish products is increasing in certain geographic areas is associated with the increasing populations of marine mammals. Seal population sizes in Iceland were previously shown to be correlated with *P. decipiens* infections (Hauksson, 2002, 2011). Similar associations were shown in Norway (Jensen and Idås, 1992) and recently grey seal population in the eastern Baltic sea has expanded significantly to around 50,000 individuals since the year 2000 leading to increased infections of *P. decipiens* and *C. osculatum* (Buchmann and Kania, 2012; Mehrdana et al., 2014; Horbowy et al., 2016). It is a local and relatively stationary seal population living together with a local and stationary cod population. Thus, the local cod population has experienced a marked increase of *P. decipiens* and *C. osculatum* infection levels.

It may be debated whether regulation of marine mammal populations in specified areas should be implemented in order to reduce the effect on fish stock size and fish product quality. Such management efforts may conflict with the conservation of the final host populations which often have protected status. The effect of hunting (culling) efforts and contraceptive measures should be evaluated in this regard. The use of anthelmintics in mass treatment of final hosts, such as seals, in order to reduce the general infection pressure, may be viewed as a theoretical option, but this solution raises environmental concerns and may be difficult to perform on a large scale.

Conflict of interests

The authors declare that they have no conflict of interests.

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References

Ackman, R.G., Gjelstad, R.T., 1975. Gas chromatographic resolution of isomeric pentanols and pentanones in the identification of volatile alcohols and ketones in the codworm *Terranova decipiens*. Anal. Biochem. 67, 684–687.

Adroher, F.J., Valero, A., Ruiz-Valero, J., Iglesias, L., 1996. Larval anisakids (Nematoda:Ascaridoidea) in horse mackerel (*Trachurus trachurus*) from the fish market in Granada, Spain. Parasitol. Res. 82 (4), 319–322.

Alonso-Gómez, A., Moreno-Ancillo, A., López-Serrano, M.C., Suarez-de-Parga, J.M., Daschner, A., Caballero, M.T., Barranco, P., Cabañas, R., 2004. *Anisakis simplex* only provokes allergic symptoms when the worm parasitises the gastrointestinal tract. Parasitol. Res. 93 (5), 378–384.

Amin, O.M., Eidelman, W.S., Domke, W., Bailey, J., Pfeifer, G., 2000. An unusual case of anisakiasis in California, USA. Comp. Parasitol. 67 (1), 71–75.

Andreassen, J., Jörring, K., 1970. Anisakiasis in Denmark. Infection with nematode larvae from marine fish. Nord. Med. 84, 1492–1495 in Danish.

Aspholm, P.E., Ugland, K.I., Jodestol, K.A., Berland, B., 1995. Seal worm (*Pseudoterranova decipiens*) infection in common seals (*Phoca vitulina*) and potential intermediate fish hosts from the outer Oslo Fjord. Int. J. Parasitol. 25, 367–373.

Arcos, S.C., Ciordia, S., Roberston, L., Zapico, I., Jiménez-Ruiz, Y., Gonzalez-Muñoz, M., Moneo, I., Carballeda-Sangiao, N., Rodriguez-Mahillo, A., Albar, J.P., Navas, A., 2014. Proteomic profiling and characterization of differential allergens in the nematodes *Anisakis simplex* sensu stricto and *A. pegreffi*. Proteomics 14 (12), 1547–1568.

Audicana, L., Audicana, M.T., de Corres, L.F., Kennedy, M.W., 1997. Cooking and freezing may not protect against allergic reactions to ingested *Anisakis simplex* antigens in humans. Vet. Rec. 140, 235.

Barros, L.A., Yamanaka, A.R., Silva, L.E., Vanzeler, M.L.A., Braum, D.T., Bonaldo, J., 2009. *In vitro* larvicidal activity of geraniol and citronellal against *Contracaecum* sp (Nematoda: Anisakidae). Braz. J. Med. Biol. Res. 42, 918–920.

Beck, M., Evans, R., Feist, S.W., Stebbing, P., Longshaw, M., Harris, E., 2008. *Anisakis simplex sensu lato* associated with red vent syndrome in wild Atlantic salmon *Salmo salar* in England and Wales. Dis. Aquat. Org. 82, 61–65.

Bourree, P., Paugam, A., Petithory, J.C., 1995. Anisakidosis: Report of 25 cases and review of the literature. Comp. Immunol. Microbiol. Infect. Dis. 18, 75–84.

Buchmann, K., Kania, P., 2012. Emerging *Pseudoterranova decipiens* (Krabbe, 1878) problems in Baltic cod, *Gadus morhua* L., associated with grey seal colonization of spawning grounds. J. Fish Dis. 35, 861–866.

Buchmann, K., Børresen, T., 1988. The effect of different food types and rations on the liver and muscle of cod (*Gadus morhua* L.). Acta Vet. Scand. 29, 57–59.

Caballero, M.L., Moneo, I., 2002. Specific IgE determination to Ani s 1, a major allergen from *Anisakis simplex*, is a useful tool for diagnosis. Ann. Allergy Asthma Immunol. 89 (1), 74–77.

Caballero, M.L., Moneo, I., Gómez-Aguado, F., Corcuera, M.T., Casado, I., Rodríguez-Pérez, R., 2008. Isolation of Ani s 5, an excretory–secretory and highly heat-resistant allergen useful for the diagnosis of Anisakis larvae sensitization. Parasitol. Res. 103 (5), 1231–1233.

Caballero, M.L., Umpierrez, A., Moneo, I., Rodriguez-Perez, R., 2011. Ani s 10, a new *Anisakis simplex* allergen: cloning and heterologous expression. Parasitol. Int. 60 (2), 209–212.

Caramello, P., Vitali, A., Canta, F., Caldana, A., Santi, F., Caputo, A., Lipani, F., Balbiano, R., 2003. Intestinal localization of anisakiasis manifested as acute abdomen. Clin. Microbiol. Infect. 9, 734–737.

Carballeda-Sangiao, N., Olivares, F., Rodriguez-Mahillo, A.I., Careche, M., Tejada, M., Moneo, I., González-Muñoz, M., 2014. Identification of autoclave resistant *Anisakis simplex* allergens. J. Food Prot. 4, 605–609.

Chitwood, M., 1975. *Phocanema*-type larval nematode coughed up by a boy in California. Am.J.Trop. Med. Hyg. 24 (4), 710–711.

Choi, S.J., Lee, J.C., Kim, M.J., Hur, G.Y., Shin, S.Y., Park, H.S., 2009. The clinical characteristics of *Anisakis* allergy in Korea. Kor. J. Intern. Med. 24 (2), 160–163.

Couture, C., Measures, L., Gagnon, J., Desbiens, C., 2003. Human intestinal anisakiosis due to consumption of raw salmon. Am. J. Surg. Pathol. 27, 1167–1172.

Daschner, A., Pascual, C.Y., 2005. *Anisakis simplex*: sensitization and clinical allergy. Curr. Opin. Allergy Clin. Immunol. 5, 281–285.

Daschner, A., Vega dela Osada, F., Pascual, C.Y., 2005. Allergy and parasites reevaluated: wide-scale induction of chronic urticaria by the ubiquitous fish-nematode *Anisakis simplex* in an endemic region. Allergol Immunopathol (Madr) 33 (1), 31–37.

Desowitz, R.S., 1986. Human and experimental anisakiasis in the United States. Hokkaido Igaky Zasshi 61, 358–371.

Eero, M., Hjelm, J., Behrens, J., Buchmann, K., Cardinale, M., Casini, M., Gasyukov, P., 2015. Eastern Baltic cod in distress: Biological changes and challenges for stock assessment. ICES J. Mar. Sci. 72 (8), 2180–2186.

EFSA, 2010. EFSA panel on biological hazards (BIOHAZ); scientific opinion on risk assessment of parasites, in fishery products. EFSA J. 8 (4), 1543 (2010, 91 pp.).

Espiñeira, M., Herrero, B., Vieites, J.M., Santaclara, F.J., 2010. Detection and identification of anisakids in seafood by fragment length polymorphism analysis and PCR–RFLP of ITS-1 region. Food Control 21, 1051–1060.

Fagerholm, H.P., 1982. Parasites of fish in Finland. VI nematodes. Acta Acad. Aboensis Ser. B 40, 5–128.

Fumarola, L., Monno, R., Ierardi, E., Rizzo, G., Giannelli, G., Lalle, M., Pozio, E., 2009. *Anisakis pegreffi* etiological agent of gastric infections in two Italian women. Foodborne Pathog. Dis. 6 (9), 1157–1159.

Fæste, C.K., Jonscher, K.R., Dooper, M.M.W.B., Egge-Jacobsen, W., Moen, A., Daschner, A., Egaas, E., Christians, U., 2014. Characterisation of potential novel allergens in the fish parasite *Anisakis simplex*. EuPA Open Proteomics 4, 140–155.

Gardiner, M.A., 1990. Survival of *Anisakis* in cold smoked Salmon. Can. Inst. Food Sci. Technol. J. 23 (2/3), 143–144.

Haarder, S., Kania, P.W., Bahlool, Q.M., Buchmann, K., 2013. Expression of immune relevant genes in rainbow trout following exposure to live *Anisakis simplex* larvae. Exp. Parasitol. 135, 564–569.

Haarder, S., Kania, P.W., Galatius, A., Buchmann, K., 2014. Increased *Contracaecum osculatum* infection in Baltic cod (*Gadus morhua*) livers (1982–2012) associated with increasing grey seal (*Halichoerus gryphus*) populations. J. Wildl. Dis. 50 (3), 537–543.
Hauksson, E., 2002. Decreases in sealworm (*Pseudoterranova* sp.) abundance in short-spined sea scorpion (*Myoxocephalus scorpius*) following declines in numbers of seals at Hvalseyjar, western Iceland. Polar Biol. 25, 531–537.
Hauksson, E., 2011. The prevalence, abundance, and density of *Pseudoterranova* sp. (p) larvae in the flesh of cod (*Gadus morhua*) relative to proximity of grey seal (*Halichoerus grypus*) colonies on the coast off Drangar, Northwest Iceland. J. Mar. Biotechnol. 235832.
Herrero, B., Vieites, J.M., Espiñeira, M., 2011. Detection of anisakids in fish and seafood products by real-time PCR. Food Control 22, 933–939.
Hierro, I., Valero, A., Pérez, P., González, P., Cabo, M.M., Montilla, M.P., Navarro, M.C., 2004. Action of different monoterpenic compounds against *Anisakis simplex* s.l. L3 larvae. Phytomedicine 11 (1), 77–82.
Hierro, I., Valero, A., Navarro, M.C., 2006. *In vivo* larvicidal activity of monoterpenic derivatives from aromatic plants against L3 larvae of *Anisakis simplex* s. L. Phytomedicine 13, 527–531.
Hitchcock, D.J., 1950. Parasitological study on the Eskimos in the Bethel area of Alaska. J. Parasitol. 36 (3), 232–234.
Horbowy, J., Podolska, M., Nadolna-Altyn, K., 2016. Increasing occurrence of anisakid nematodes in the liver cod (*Gadus morhua*) from the Baltic Sea: Does infection affect the condition and mortality of fish? Fish. Res. 179, 98–103.
Højgaard, D.P., 1998. Impact of temperature, salinity and light on hatching of eggs of *Anisakis simplex* (Nematoda, Anisakidae), isolated by a new method, and some remarks on survival of larvae. Sarsia 83 (1), 21–28.
Ido, K., Yuasa, H., Ide, M., Kimura, K., Toshimitsu, K., Suzuki, T., 1998. Sonographic diagnosis of small intestinal anisakiasis. J. Clin. Ultrasound 26 (3), 125–130.
Ishikura, H., Kikuchi, K., Nagasawa, K., Ooiwa, T., Takamiya, H., Sato, N., Sugane, K., 1993. Anisakidae and anisakidosis. In: Sun, T. (Ed.), Progress in Clinical Parasitology 8. Springer Verlag, N. Y., pp. 43–102.
Ishikura, H., 2003. Anisakiasis (2) clinical pathology and epidemiology. In: Otsuru, M., Kamegai, S., Hayashi, S. (Eds.), Progress of Medical Parasitology in Japan. 8, pp. 451–473.
Jabbar, A., Fong, R.W., Kok, K.X., Lopata, A.L., Gasser, R.B., Beveridge, I., 2013. Molecular characterization of anisakid nematode larvae from 13 species of fish from Western Australia. Int. J. Food Microbiol. 161, 247–253.
Jensen, T., Idås, K., 1992. Infection with *Pseudoterranova decipiens* (Krabbe, 1878) larvae in cod (*Gadus morhua*) relative to proximity of seal colonies. Sarsia 76, 227–230.
Kagei, N., Isogaki, H., 1992. A case of abdominal syndrome caused by the presence of a large number of *Anisakis* larvae. Int. J. Parasitol. 22 (2), 251–253.
Karl, H., Leinemann, M., 1993. A fast and quantitative detection method for nematodes in fish fillets and fishery products. Arch. Leb. 44, 105–128.
Karl, H., Roepstorff, A., Huss, H.H., Bloemsma, B., 1994. Survival of *Anisakis* larvae in marinated herring fillets. Int. J. Food Sci. Technol. 29, 661–670.
Karpiej, K., Dzido, J., Rokicki, J., Kijewska, A., 2013. Anisakid nematodes of Greenland halibut *Reinhardtius hippoglossoides* from the Barents Sea. J. Parasitol. 99 (4), 650–654.
Kim, K.H., Eom, K.S., Park, J.K., 2006. The complete mitochondrial genome of *Anisakis simplex* (Ascaridida: Nematoda) and phylogenetic implications. Int. J. Parasitol. 36, 319–328.
Kliks, M.M., 1983. Anisakiasis in the western United States: four new case reports from California. Am.J.Trop. Med. Hyg. 32 (3), 526–532.
Koh, M.S., Huh, S., Sohn, W.M., 1999. A case of gastric pseudoterranoviasis in a 43-year-old man in Korea. Korean J. Parasitol. 37 (1), 47–49.
Kobayashi, Y., Shimakura, K., Ishizaki, S., Nagashima, Y., Shiomi, K., 2007. Purification and cDNA cloning of a new heat stable allergen from *Anisakis simplex*. Mol. Biochem. Parasitol. 155, 138–145.
Køie, M., Fagerholm, H.P., 1995. The life cycle of *Contracaecum osculatum* (Rudolphi, 1802) sensu stricto (Nematoda, Ascaridoidea, Anisakidae) in view of experimental infections. Parasitol. Res. 81, 481–489.
Køie, M., Berland, B., Burt, M.D.B., 1995. Development to third stage larvae occurs in the eggs of *Anisakis simplex* and *Pseudoterranova decipiens* (Nematoda, Ascaridoidea, Anisakidae). Can. J. Fish. Aquat. Sci. 52, 134–139.
Levsen, A., Lunestad, B.T., Berland, B., 2005. Low detection efficiency of candling as a commonly recommended inspection method for nematode larvae in the flesh of pelagic fish. J. Food Prot. 68, 828–832.
Levsen, A., Lunestad, B.T., 2010. *Anisakis simplex* third stage larvae in Norwegian spring spawning herring (*Clupea harengus* L.), with emphasis on larval distribution in the flesh. Vet. Parasitol. 4 (3–4), 247–253.
Levsen, A., Berland, B., 2012. Anisakis species. In: Woo, P.T.K., Buchmann, K. (Eds.), Fish Parasites, Pathobiology and Protection 18. CAB International, pp. 298–309.
Li, S.W., Shiao, S.H., Weng, S.C., Liu, T.H., Su, K.E., Chen, C.C., 2015. A case of human infection with *Anisakis simplex* in Taiwan. Gastrointest. Endosc. 82 (4), 757–758.
Lin, A.H., Nepstad, I., Florvaag, E., Egaas, E., Van Do, T., 2014. An extended study of seroprevalence of anti-*Anisakis simplex* IgE antibodies in Norwegian blood donors. Scand. J. Immunol. 79 (1), 61–67.
Little, M.D., MacPhail, J.C., 1972. Large nematode larva from the abdominal cavity of a man in Massachusetts. Am. J. Trop. Med. Hyg. 21 (6), 948–950.
Little, M.D., Most, H.A.R.R.Y., 1973. Anisakid larva from the throat of a woman in New York. Am.J.Trop. Med. Hyg. 22 (5), 609.
Liu, S.S., Liu, G.H., Zhu, X.Q., Weng, Y.B., 2015. The complete mitochondrial genome of *Pseudoterranova azarasi* and comparative analysis with other anisakid nematodes. Infect. Genet. Evol. 33, 293–298.
Lopez, I., Pardo, M.A., 2010. Evaluation of a real-time polymerase chain reaction (PCR) assay for detection of *Anisakis simplex* parasite as a food-borne allergen source in seafood products. J. Agric. Food Chem. 58 (3), 1469–1477.
Lunneryd, S.G., Boström, M.K., Aspholm, P.E., 2015. Sealworm (*Pseudoterranova decipiens*) infection in grey seals (*Halichoerus grypus*), cod (*Gadus morhua*) and shorthorn sculpin (*Myoxocephalus scorpius*) in the Baltic Sea. Parasitol. Res. 114 (1), 257–264.
Margolis, L., 1977. Public health aspects of "codworm" infection: a review. J. Fish. Res. Board Can. 34, 887–898.
Matsumoto, T., Iida, M., Kimura, Y., Tanaka, K., Kitada, T., Fujishima, M., 1992. Anisakiasis of the colon: radiologic and endoscopic features in six patients. Radiology 183 (1), 97–99.
Mattiucci, S., Nascetti, G., 2007. Genetic diversity and infection levels of anisakid nematodes parasitic in fish and marine mammals from Boreal and Austral hemispheres. Vet. Parasitol. 148, 43–57.
Mattiucci, S., Nascetti, G., 2008. Advances and trends in the molecular systematics of anisakid nematodes, with implications for their evolutionary ecology and ost-parasite co-evolutionary process. Adv. Parasitol. 66, 47–148.
Mattiucci, S., Paggi, L., Nascetti, G., Ishikura, H., Kikuchi, K., Sato, N., Cianchi, R., Bullini, L., 1998. Allozyme and morphological identification of *Anisakis*, *Contracaecum* and *Pseudoterranova* from Japanese waters (Nematoda, Ascaridoidea). Syst. Parasitol. 40, 81–92.
Mattiucci, S., Cipriani, P., Webb, S.C., Paoletti, M., Marcer, F., Bellisario, B., Gibson, D.I., Nascetti, G., 2014. Genetic and morphological approaches distinguish the three sibling species of the *Anisakis simplex* species complex, with a species designation as *Anisakis berlandi* n. sp. for *A. simplex* sp. C (Nematoda: Anisakidae). J. Parasitol. 100 (2), 199–214.
McClelland, G., Martell, D.J., 2001. Surveys of Larval Sealworm (Pseudoterranova Decipiens) Infection in Various Fish Species Sampled from Nova Scotian Waters between 1988 and 1996, with an Assessment of Examination Procedures. 3. NAMMCO Scientific Publications, pp. 57–76.
McClelland, G., 2002. The trouble with sealworms (*Pseudoterranova decipiens* species complex, Nematoda): a review. Parasitology 124, 183–203.
Mehrdana, F., Bahlool, Q.Z., Skov, J., Marana, M.H., Sindberg, D., Mundeling, M., Overgaard, B.C., Korbut, R., Strøm, S.B., Kania, P.W., Buchmann, K., 2014. Occurrence of zoonotic nematodes *Pseudoterranova decipiens*, *Contracaecum osculatum* and *Anisakis simplex* in cod (*Gadus morhua*) from the Baltic Sea. Vet. Parasitol. 205 (3–4), 581–587.
Mercado, R., Torres, P., Muñoz, V., Apt, W., 2001. Human infection by *Pseudoterranova decipiens* (Nematoda, Anisakidae) in Chile: report of seven cases. Mem. Inst. Oswaldo Cruz 96 (5), 653–655.
Mladineo, I., Poljak, V., Martínez-Sernández, V., Ubeira, F.M., 2014. Anti-*Anisakis* IgE seroprevalence in the healthy Croatian coastal population and associated risk factors. PLoS Negl. Trop. Dis. 8 (2), 2673.
Mohandas, N., Jabbar, A., Podolska, M., Zhu, X.Q., Littlewood, D.T., Jex, A.R., Gasser, R.B., 2014. Mitochondrial genomes of *Anisakis simplex* and *Contracaecum osculatum* (sensu stricto)-comparisons with selected nematodes. Infect. Genet. Evol. 21, 452–462.

Moore, D.A.J., Girdwood, R.W.A., Chiodini, P.L., 2002. Treatment of anisakiasis with albendazole. Lancet 360, 54.
Möller, H., Schröder, S., 1987. Neue Aspekte der Anisakiasis in Deutschland (New aspects of anisakidosis in Germany). Arch. Leb. 38, 123–128 in German.
Möller, H., 1975. Die Parasiten des Dorsches (*Gadus morhua* L.) in der Kieler Förde. Ber. Dt. Wiss. Kommn. Meeresforsch. 24, 71–78.
Myjak, P., Szostakowska, B., Wojciechowski, J., Pietkiewicz, H., Rokicki, J., 1994. Anisakid larvae in cod from the southern Baltic Sea. Arch. Fish. Mar. Res. 42, 149–161.
Na, H.K., Seo, M., Chai, J.Y., Lee, E.K., Jeon, S.M., 2013. A case of anisakidosis caused by *Pseudoterranova decipiens* larva. Korean J. Parasitol. 51 (1), 115–117.
Nadler, S.A., D'Amelio, S., Dailey, M.D., Paggi, L., Siu, S., Sakanari, J.A., 2005. Molecular phylogenetics and diagnosis of *Anisakis, Pseudoterranova,* and *Contracaecum* from northern Pacific marine mammals. J. Parasitol. 91, 1413–1429.
Nadolna, K., Podolska, M., 2014. Anisakid larvae in the liver of cod (*Gadus morhua*) L. from the southern Baltic Sea. J. Helminthol. 88, 237–246.
Nagasawa, K., 2012. The biology of *Contracaecum osculatum sensu lato* and *C. osculatum* A (Nematoda: Anisakidae) in Japanese waters: a review. Biosphere Sci. 51, 61–69.
Nascetti, G., 2011. First molecular identification of the zoonotic parasite *Anisakis pegreffii* (Nematoda: Anisakidae) in a paraffin-embedded granuloma taken from a case of human intestinal anisakiasis in Italy. BMC Infect. Dis. 11, 82.
Nieuwenhuizen, N., Lopata, A.L., Jeebhay, M.F., Herbert, D.R., Robins, T.G., Brombacher, F., 2006. Exposure to the fish parasite *Anisakis* causes allergic airway hyperreactivity and dermatitis. J. Allergy Clin. Immunol. 117, 1098–1105.
Noguera, P., Collins, C., Bruno, D., Pert, C., Turnbull, A., McIntosh, A., Lester, K., Bricknell, I., Wallace, S., Cook, P., 2009. Red vent syndrome in wild Atlantic salmon *Salmo salar* in Scotland is associated with *Anisakis simplex sensu stricto* (Nematoda: Anisakidae). Dis. Aquat. Org. 87, 199–215.
Ólafsdóttir, D., Hauksson, E., 1997. Anisakid (Nematoda) infestations in Icelandic grey seals (*Halichoerus grypus* Fabr.). J. Northwest Atl. Fish. Sci. 22, 259–269.
Ólafsdóttir, D., Hauksson, E., 1998. Anisakid nematodes in the common seal *Phoca vitulina* L. in Icelandic waters. Sarsia 83, 309–316.
Oshima, T., 1987. Anisakiasis- is the sushi bar guilty? Parasitol. Today 3 (2), 44–48.
Pacios, E., Arias-Diaz, J., Zuloaga, J., Gonzalez-Armengol, J., Villarroel, P., Balibrea, J.L., 2005. Albendazole for the treatment of anisakiasis ileus. Clin. Infect. Dis. 41 (12), 1825–1826.
Pampiglione, S., Rivasi, F., Criscuolo, M., De Benedittis, A., Gentile, A., Russo, S., Testini, M., Villan, M., 2002. Human anisakiasis in Italy: a report of eleven new cases. Pathol. Res. Pract. 198 (6), 429–434.
Perdiguero-Alonso, D., Montero, F., Raga, J.A., Kostadinova, A., 2008. Composition and structure of the parasite faunas of cod, *Gadus morhua* L. (Teleostei: Gadidae) in the North East Atlantic. Parasites Vectors 1, 1–18.
Perteguer, M.J., Chivato, T., Montoro, A., Cuellar, C., Mateos, J.M., Laguna, R., 2000. Specific and total IgE in patients with recurrent, acute urticarial caused by *Anisakis simplex*. Ann. Trop. Med. Parasitol. 94 (3), 259–268.
Petrushevski, G.K., Shulman, G.G., 1955. Infection of Baltic cod liver with roundworms. Tr. Akad. Nauk Litovskoj SSR Ser. B. 2, 119–125 (In Russian).
Pinel, C., Beaudevin, M., Chermette, R., Grillot, R., Ambroise-Thomas, P., 1996. Gastric anisakidosis due to *Pseudoterranova decipiens* larva. Lancet 347 (9018), 1829.
Pinkus, G.S., Coolidge, C., Little, M.D., 1975. Intestinal anisakiasis. First case report from North America. A. J. Med. 59, 114–120.
Puente, P., Anadón, A., Rodero, M., Romarís, F., Ubeira, F., Cuéllar, C., 2008. *Anisakis simplex*: the high prevalence in Madrid (Spain) and its relation with fish consumption. Exp. Parasitol. 118, 271–274.
Quiazon, K.M., Yoshinaga, T., Santos, M.D., Ogawa, K., 2009. Identification of larval *Anisakis* spp. (Nematoda: Anisakidae) in Alaska pollock (*Theragra chalcogramma*) in northern Japan using morphological and molecular markers. J. Parasitol. 95 (5), 1227–1232.
Repiso, O.A., Alcántara, T.M., González, F.C., de Artaza, V.T., Rodríguez, M.R., Valle Muñoz, J., Martínez Potenciano, J.L., 2003. Gastrointestinal anisakiasis. Study of a series of 25 patients. Gastroenterol. Hepatol. 26 (6), 341–346 (In Spanish).
Rohlwing, T.M., Palm, H.W., Rosenthal, H., 1998. Parasitisation with *Pseudoterranova decipiens* (Nematoda) influences the survival rate of the European smelt *Osmerus eperlanus* retained by a screen wall of a nuclear power plant. Dis. Aquat. Org. 32, 233–236.
Romero, C., Valero, A., Martín-Sánchez, J., Navarro-Moll, M.C., 2012. Activity of *Matricaria chamomilla* essential oil against anisakiasis. Phytomedicine 19 (6), 520–523.
Sawada, Y., Moriyama, Y., Ebina, T., Sasaki, H., Yoshida, Y., Tanabe, K., Chiba, R., 1983. Gastric terranovasis: report of 14 cases. Gastroenterol. Endosc. 25, 713–717.
Santoro, M., Mattiucci, S., Work, T., Cimmaruta, R., Nardi, V., Cipriani, P., Bellisario, B., Nascetti, G., 2013. Parasitic infection by larval helminths in Antarctic fishes: pathological changes and impact on the host body condition index. Dis. Aquat. Org. 105 (2), 139–148.
Schaum, E., Müller, W., 1967. Heterocheilidiasis (case report). Dtsch. Med. Wochenschr. 92, 2230–2233.
Shamsi, S., Butcher, A.R., 2011. First report of human anisakidosis in Australia. Med. J. Aust. 194, 199–200.
Skirnisson, K., 2006. *Pseudoterranova decipiens* (Nematoda, Anisakidaeae) larvae reported from humans in Iceland after consumption of insufficiently cooked fish. Laeknabladid J. 92, 21–25 (in Icelandic).
Skrzypczak, M., Rokicki, J., Pawliczka, I., Najda, K., Dzido, J., 2014. Anisakids of seals found on the southern coast of Baltic Sea. Acta Parasitol. 59, 165–172.
Smith, J.W., Wootten, R., 1978. *Anisakis* and anisakiasis. Adv. Parasitol. 16, 93–163.
Sprengel, G., Lüchtenberg, H., 1991. Infection by endoparasites reduces swimming speed of European smelt *Osmerus eperlanus* and European eel *Anguilla anguilla*. Dis. Aquat. Org. 11, 31–35.
Strøm, S.B., Haarder, S., Korbut, R., Mejer, H., Thamsborg, S.M., Kania, P.W., Buchmann, K., 2015. Third-stage nematode larvae of *Contracaecum osculatum* from Baltic cod (*Gadus morhua*) elicit eosinophilic granulomatous reactions when penetrating the stomach mucosa of pigs. Parasitol. Res. 114, 1217–1220.
Suzuki, J., Murata, R., Hosaka, M., Araki, J., 2010. Risk factors for human *Anisakis* infection and association between the geographic origins of *Scomber japonicus* and anisakid nematodes. Int. J. Food Microbiol. 137, 88–93.
Testini, M., Gentile, A., Lissidini, G., Di Venere, B., Pampiglione, S., 2003. Splenic anisakiasis resulting from a gastric perforation: an unusual occurrence. Int. Surg. 88, 126–128.
Thulin, J., Höglund, J., Lindesjöö, E., 1989. Fish Diseases in Coastal Waters (Fisksjukdomar I Kustvatten). Naturvårdsverket (Natural Agency), Almänna Förlaget, Solna (126 pp.).
Timi, J.T., Paoletti, M., Cimmaruta, R., Lanfranchi, A.L., Alarcos, A.J., Garbin, L., George-Nascimento, M., Rodríguez, D.H., Giardino, G.V., Mattiucci, S., 2014. Molecular Identification, Morphological Characterization and New Insights into the Ecology of Larval *Pseudoterranova cattani* in Fishes from the Argentine Coast with its Differentiation from the Antarctic Species, *P. decipiens* Sp. E (Nematoda: Anisakidae). Vet. Parasitol. 199 (1–2), 59–72.
Torres, P., Jercic, M.I., Weitz, J.C., Dobrew, E.K., Mercado, R.A., 2007. Human pseudoterranovosis, an emerging infection in Chile. J. Parasitol. 93 (2), 440–443.
Valiñas, B., Lorenzo, S., Eiras, A., Figueiras, A., Sanmartín, M.L., Ubeira, F.M., 2001. Prevalence of and risk factors for IgE sensitization to *Anisakis simplex* in a Spanish population. Allergy 56 (7), 667–671.
Valtonen, E.T., Fagerholm, H.P., Helle, E., 1988. *Contracaecum osculatum* (Nematoda: Anisakidae) in fish and seals in Bothnian Bay (norteastern Baltic Sea). Int. J. Parasitol. 18, 365–370.
Van Thiel, P.H., Kuipers, F.C., Roskam, T.H., 1960. A nematode parasitic to herring, causing acute abdominal syndromes in man. Trop. Geogr. Med. 12, 97–113.
Werner, M.T., Fæste, C.K., Levsen, A., Egaas, E., 2011. A quantitative sandwich ELISA for the detection of *Anisakis simplex* in seafood. Eur. Food Res. Technol. 232, 157–166.
Wharton, D.A., Aalders, O., 2002. The response of *Anisakis* larvae to freezing. J. Helminthol. 76, 363–368.
Xu, X., Sui, J., Cao, L., Lin, H., 2010. Direct competitive enzyme-linked immunosorbent assay (ELISA) for rapid screening of anisakid larvae in seafood. J. Sci. Food Agric. 90, 877–881.
Yang, X., Nian, R., Lin, H., Duan, C., Sui, J., Cao, L., 2013. Detection of anisakid larvae in cod fillets by UV fluorescent imaging based on principal component analysis and gray value analysis. J. Food Prot. 76 (7), 1288–1292.
Yu, J.R., Seo, M., Kim, Y.W., Oh, M.H., Sohn, W.M., 2001. A human case of gastric infection by *Pseudoterranova decipiens* larva. Korean J. Parasitol. 39 (2), 193–196.
Zhu, X.Q., Podolska, M., Liu, J.S., Yu, H.Q., Chen, H.H., Z., L., Luo, C., Song, H.Q., Lin, R.Q., 2007. Identification of anisakid nematodes with zoonotic potential from Europe and China by single-strand conformation polymorphism analysis of nuclear ribosomal DNA. Parasitol. Res. 101, 1703–1707.
Zuo, S., Al-Jubury, A., Korbut, R., Christensen, N.H., Kania, P.W., Buchmann, K., 2016. Host size dependent infection dynamics *of Contracaecum osculatum* (Nematoda, anisakidae) in Baltic cod (*Gadus morhua*) associated with differential food preferences. Dis. Aquat. Org. 120, 69–75.