

First Report of Microsporidian Xenomas from the Red Sea Bream *Pagrus major* (Temminck & Schlegel, 1843) (Sparidae) from the Red Sea off Jizan, KSA: A Light and Transmission Electron Microscopic Study

Kareem Morsy^{1,2*}, Saad Bin Dajem¹, Mohammed Al-Kahtani¹, Fahmy Elsaid^{1,4}, Ali Shati¹, Mashaal Bin-Meferij⁴ & Attalla El-Kott^{1,5}

¹Department of Biology, College of Science, King Khalid University, Abha, Saudi Arabia

²Department of Zoology, Faculty of Science, Cairo University, Cairo, Egypt

³Department of Zoology, Faculty of Science, Mansoura University, Mansoura, Egypt

⁴Department of Biology, Princess Nourah Bint Abdelrahman, Riyadh, Saudi Arabia

⁵Department of Zoology, Faculty of Science, Damanhour University, Damanhour, Egypt

Abstract: Parasites belonging to the phylum Microsporida comprise more than 1300 species, which are obligate parasites of marine fish. These parasites cause tissue atrophy to the infected fish. In the present study, five (16.66%) out of 30 examined specimens of *Pagrus major* (Sparidae) captured off Jizan, the Red Sea coast of Saudi Arabia, showed natural microsporidian infection. The parasite formed tumor masses or plasmodia in the muscle tissue and the intestinal epithelium of the peritoneum. The microsporidium was characterised by light as well as transmission electron microscopy (TEM). Light microscopy studies showed xenomas encircled by a fibrous layer enclosing the various developmental stages. Mature spores were 1.7 ± 0.6 ($1.5-2.7$) μm long and 1.5 ± 0.3 ($1.2-1.8$) μm wide, with an apparent posterior vacuole. Examination of ultrathin sections by TEM revealed the presence of uninucleated spores with anterior lamellar polaroplast and lateral 5–8 polar filament coils with electron dense centre. Generative cells were the early recognised stages underneath the surface coat, followed by uninucleated, binucleated, multinucleated meronts, sporoblasts and finally immature and mature spores. Xenomas as well as developmental stages of the parasite compared with microsporidian species recorded previously from different geographical areas, showed that the microsporidium belonged to *Pleistophora pagri* described from *Pagrus pagrus*.

Key words: Plasmodia, *Pleistophora pagri*, Ultrastructure, Marine fish, morphology.

Introduction

Microsporidia are single-cell pathogens related to the Fungi (VAVRA & LUKES 2013). More than 1300 species of over 187 genera belonging to the phylum Microsporidia were recorded as parasites of many organisms, from invertebrates to humans (AZEVEDO & MATOS 2002, 2003, BECKER & SPEARE 2004). Habitats and infected host species are the main categories for successful classification of microsporidia, often in association with molecular tools as com-

plementary (BELL et al. 2001, SMALL et al. 2014). Studying the type species and taxonomy of the recovered Microsporidia required a detailed ultrastructural description at the different stages of their plasmodia (NILSEN et al. 1998, BELL et al. 2001). Until now, about 156 species of Microsporidia infecting fishes have been recorded. Heavy infection with these parasites leads to severe muscle atrophy, body deformities and unpleasant taste for consumers

*Corresponding author: kareemsaid156@yahoo.com

and, finally, to economic loss, especially for commercial fishes (CASAL et al. 2008, JOH et al. 2007). Members of the genus *Pleistophora* Gurley, 1893 form xenomas in vertebrate animals, especially in bony fishes; they can be transmitted to healthy hosts by minute uninucleated spores released through plasmodial rupture (FIELDING et al. 2005).

The aim of the present study is to present ultrastructural characteristics of the microsporidium *Pleistophora pagri* isolated from muscle tissues as well as epithelium of different viscera of the red sea bream *Pagrus major* (Temminck & Schlegel, 1843) inhabiting off Jizan coasts of the Red Sea in Saudi Arabia.

Materials and Methods

A total of forty five red sea bream snappers *Pagrus major* (Sparidae) were collected from water locations along the Red Sea at Jizan coasts (16.8894° N, 42.5706° E) during the period January – April 2018. Fish were transported alive in ice boxes filled with the same water source to the laboratory of parasitology, where they were identified and further examined. After dissection, microsporidian infection was detected by the presence of xenomas (plasmodia) along the internal viscera and host muscles. Further elucidation of infection was carried out by spreading the contents of one plasmodium between glass slide and a cover slip. Fresh spores were photographed by Zeiss photomicroscope (135 Axiovert) equipped by a digital Canon camera. Morphometrics of spores were documented according to LOM & ARTHUR (1989) and expressed as mean \pm standard deviation with the range in parentheses. For TEM, the parasite plasmodia were fixed in 3% buffered glutaraldehyde, pH 7.4 for 3h and post-fixation in osmium tetroxide (2%) for 2h were employed. Samples were dehydrated in a graded ethanol series. Embedding was in Spurr's resin. One micrometer thick semi-thin sections cut on Leica ultracut (UCT) ultramicrotome and further stained in toluidine blue were examined. Uranyl acetate and lead citrate were used for staining ultrathin sections which were examined with Jeol 1220 transmission electron microscope. Infection parameters are as defined in BUSH et al. (1997).

Results

Five out of 30 (16.66%) fish were naturally infected with tumor like masses up to 3 mm spread over the intestinal epithelial lining as well as muscle tissue (Fig. 1). The parasite was recorded with a high prevalence during winter. After rupture of xenomas, the liberated spores (Fig. 4) were morphologically examined by

light microscopy. They were ellipsoid with a posterior vacuole. No extruded polar filaments were observed. Spores measured 1.7 ± 0.6 (1.5–2.7) μm x 1.5 ± 0.3 (1.2–1.8) μm . Examination of semi-thin sections revealed the presence of fibrous layers covering plasmodia which were in close contact with the cytoplasm of the hypertrophied host cell (Figs. 3, 4). Several developmental stages were recorded in the ultrathin sections of the parasite xenomas examined by TEM. It was observed that plasmodia were covered by an amorphous coat (Fig. 6) surrounding developmental stages of the parasite. Spores were seen in aggregates called sporophorous vesicles and surrounded by a sporophorous wall (Fig. 7). The number of the spores within vesicles was not counted due to the sectioning plane during ultrathin sections preparation. The sporophorous wall surrounding vesicles constituted of two layers derived from cisternae of endoplasmic reticulum (ER) (Fig. 8). The generative cells (GC) were the first to be recognized within xenomas below the surface coat, while meronts were the following stages produced from the progressive development of GC. Plasmatomy of meronts yielded in sporonts which producing later sporoblasts. Sporoblasts (Figs. 9, 10) further matured (Figs. 11, 12) and finally transformed into immature spores (Figs. 13–15). Polar filament formation, exospore development as well as cell elongation were the trademarks of beginning the stage of sporogenesis. Mature spores were uninucleated, ellipsoid (Figs. 16–18) with an anterior central anchoring disc (Figs. 19–25). Polaroplast, the laminar membrane system, was composed of closely and loosely packed membrane regions (Figs. 18, 19). Exospore was the outer thick, electron dense layer covering mature spores while the endospore was the interior thin layer (Fig. 26). Polar filament of each spore formed 5–9 coils encircled around posterior vacuole (Figs. 16–20, 27). Each coil was divided into three distinct areas in cross section: outer dense, electron transparent layer, and the amorphous inner layer (Fig. 27). Based on the light and ultrastructure features as well as the infection site the detected in *Pagrus major* microsporidium was determined as *Pleistophora pagri*.

Taxonomic summary

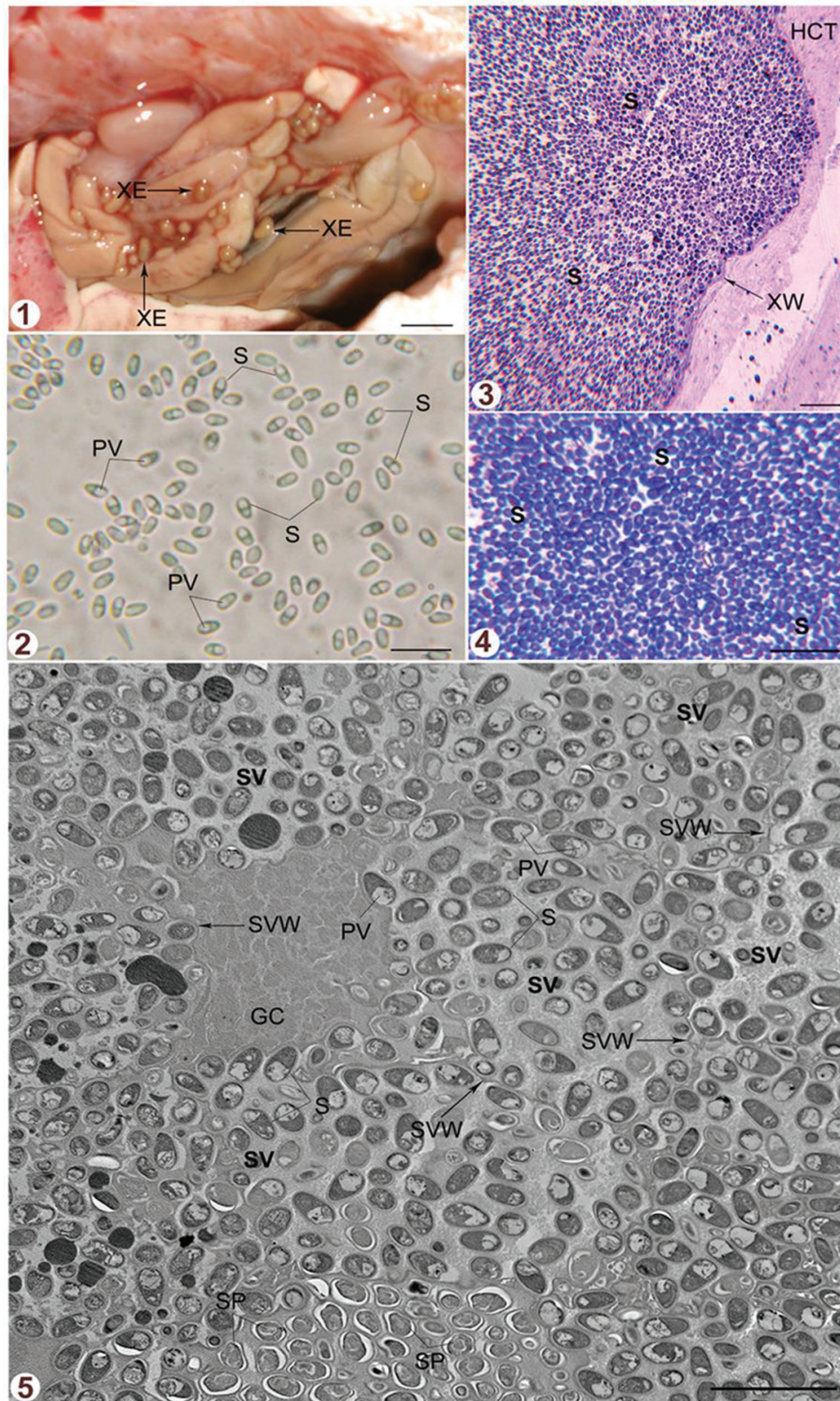
Parasite: *Pleistophora pagri* Morsy et al., 2012 (Microsporidia, Pleistophoridae).

Locality: off Jizan coasts (16.8894° N, 42.5706° E), Red Sea.

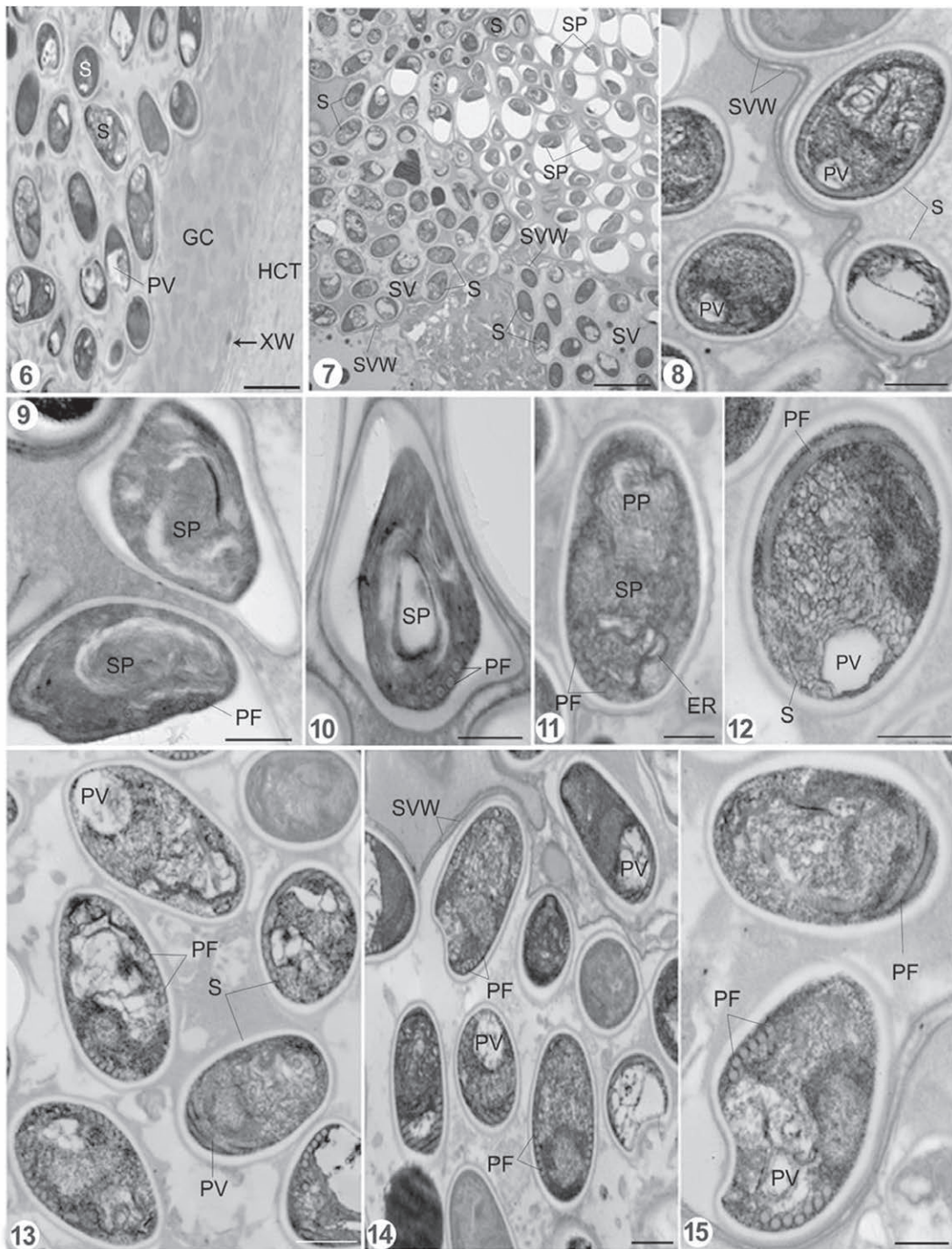
Host: Red Sea Bream *Pagrus major* (family Sparidae).

Site of infection: Tumor like masses embedded in host muscles and in epithelia of visceral organs.

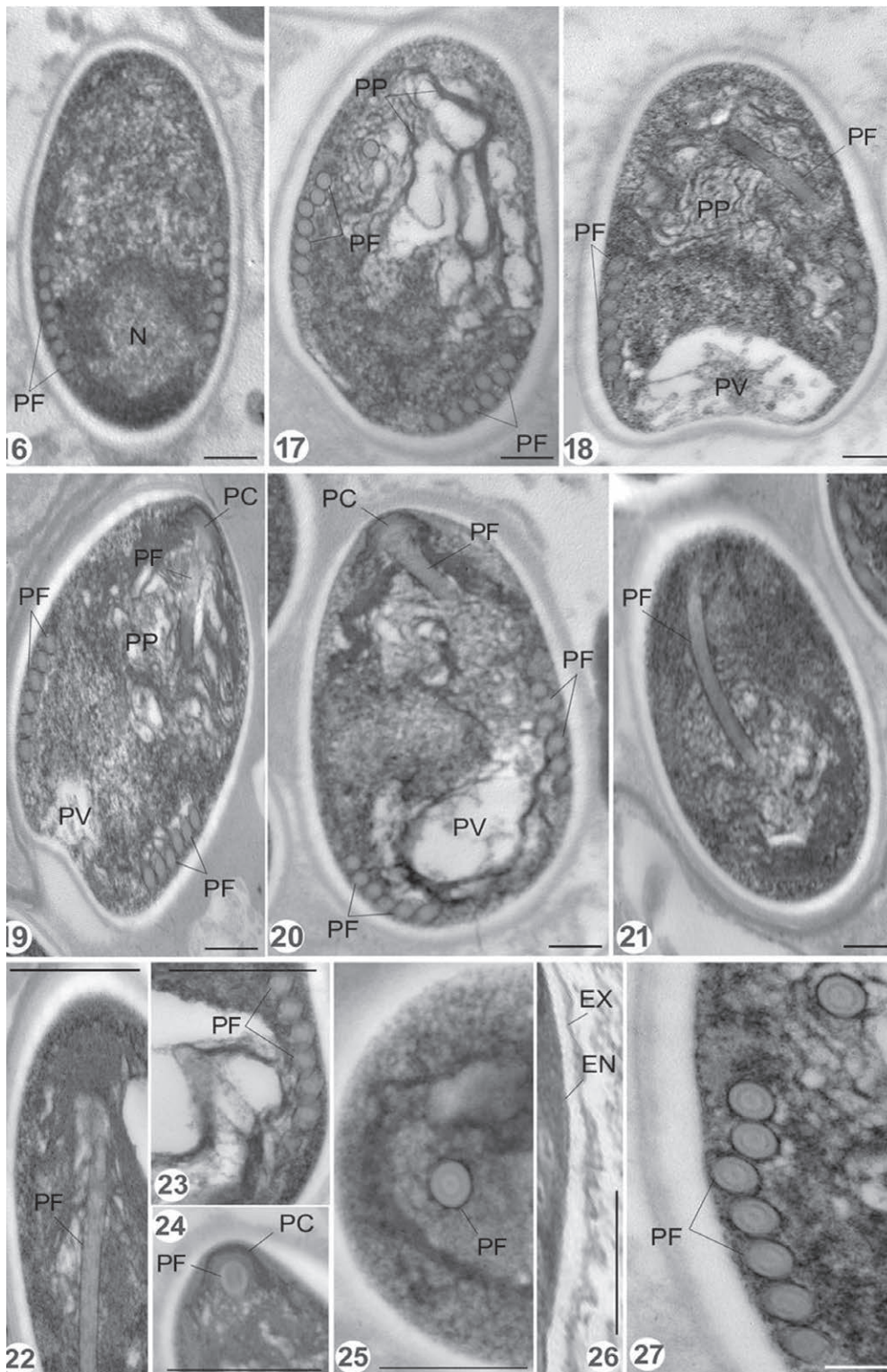
Prevalence: Five out of 30 (16.66%).



Figs 1–5. 1. *Pagrus major* infected with the microsporidian parasite *Pleistophora pagri*. The infection appears in the form of brownish xenomas (XE) embedded in the viscera of peritoneum. Scale bar 1 cm. 2–7. Photomicrographs: 2. Unfixed, free fresh spores (S) released after rupture of cysts. A posterior vacuole (PV) is observed away from the spore apex. Scale bar 10 μ m. 3, 4. Semi-thin sections through part of a xenoma stained with toluidine blue. Note that xenoma bordered by a thin wall (XW) of parasite origin and covered by a layer of connective tissue of host origin (HCT). Scale bar 20 μ m. 5. TEM micrograph showing top view of a xenoma with different developmental stages of the parasite arranged within sporophorous vesicle (SV) limited by sporophorous vesicle wall (SVW). The stages include generative cells (GC), sporoblasts (SP), immature and mature spores (S). Scale bar 10 μ m.



Figs 6–15. TEM micrographs. **6.** Periphery of a xenoma showing the surrounding fibrillar cyst wall (XW) covered by a layer of connective tissue of host origin (HCT), generative cells (GC) are the early recognizable stages below the cyst wall, immature and mature spores (S) appeared as going to the cyst interior. A posterior vacuole (PV) is observed situated away from the spore apex. Scale bar 2 μ m. **7.** Part of a xenoma showing the different developmental stages: sporoblasts (SP) and mature spores (S); they are mostly included within special structures known as sporophorous vesicle (SV) limited by sporophorous vesicle wall (SVW). Scale bar 2 μ m. **8.** The surrounding double layered sporophorous wall (SVW) surrounding spores. Scale bar 500 nm. **9, 10.** Early stages of sporoblasts (SP) and their developing polar filaments (PF). Scale bar 500 nm. **11, 12.** Advanced stages of sporoblasts with developed polar filaments (PF), polaroplasts (PP) and cell organelles such as endoplasmic reticulum (ER). The posterior vacuole (PV) appeared in these stages. Scale bar 500 nm. **13–15.** Immature spores surrounded by incompletely formed membranes. Extrusion apparatus still missing at this stage, only the polar filament coils (PF) were observed. Scale bar 500 nm.



Figs 16–27. TEM micrographs. **16–18.** Nearly mature spores. They are uninucleated (N). Posterior vacuoles (PV) are absent in the early stages and appear in the progressive stages of spore maturation. Completely formed filament coils, polar tube as well as polaroplast membranes are observed. Scale bar 500 nm. **19, 20.** Mature spores with completely formed spore apex with the extrusion apparatus composed of the anchoring disk or polar cap (PC), which is formed of a cap-like structure that plugged the anterior portion of the spore. Polar filament coils (PF) and polaroplast (PP) are seen. Scale bar 500 nm. **21–25.** Cross sections of mature spores showing the polar filament (PF). **26.** Membranes covering spore in the form of an outer thick exospore (EX) and an inner thin endospore (EN). Scale bar 500 nm. **27** Details of the filament coils (PF) arranged in one row at each side of the posterior vacuole; each coil consists of three layers: an outer, inner dense layers and a middle lighter one. Scale bar 100 μ m.

Table 1. Comparison between *Pleistophora pagri* as described in the present study and some xenoma-forming microsporidia.

Species	Host	Infection site	Spore			References
			Length	Width	Form	
<i>Pleistophora typicalis</i>	<i>Myoxocephalus scorpioides</i>	Striated muscle	4.4–7.5	2.3–3.0	Microspores uninucleated Macrospores binucleated	GURLEY (1893)
<i>Pleistophora aegyptiaca</i>	<i>Saurida tumbil</i>	Peritoneal cavity	1.7±0.6 (1.5–2.7)	1.5±0.3 (1.2–1.8)	Elongated, ovoid or ellipsoidal	ABDEL-GHAFFAR et al. (2012)
<i>Glugea stephani</i>	<i>Pleuronectes americanus</i>	Liver tissue	3.9	1.7	Oblong–ovate, slightly attenuated at the anterior end	WEIDNER (1976)
<i>Glugea anomala</i>	<i>Gasterosteus aculeatus</i>	Spleen, kidney, peritoneal cavity, subcutaneously	4.9 (4.5–5.5)	2.6 (2.4–2.8)	Elongated, ovoid	MONIEZ (1887)
<i>Loma psittaca</i>	<i>Colomesus psittacus</i>	Adherent to the intestinal mucosa	4.2 ± 0.4	2.8 ± 0.4	disporoblastic ovoid spores	CASAL et al. (2009)
<i>Tetramicra brevifilum</i>	<i>Scophthalmus maximus</i>	The conjunctiva layers of muscle surface	3.7±0.03	2.7±0.03	Ovoid, wider at the posterior end	MATTHEWS & MATTHEWS (1980)
<i>Spraguea lophii</i>	<i>Lophius piscatorius</i>	Spinal column close to kidney	—	—	Cylindrical spores are binucleated, whereas oval spores are uninucleated	MANSOUR et al. (2013)
<i>Heterosporis anguillarum</i>	<i>Anguilla japonica</i>	Skeletal muscle	1.5–2	—	Elongated with a large posterior vacuole filled with dense floccular material	HOSHINA (1951)
<i>Microgenma caulleryi</i>	<i>Hyperoplus lanceolatus</i>	Liver tissue	2.6 (2.3–2.9)	1.2 (1.1–1.4)	Ovoid and uninucleated	LEIRO et al. (1999)
<i>Microgenma tincae</i>	<i>Symphodus tinca</i>	Liver tissue	—	—	Ovocylindrical	MANSOUR et al. (2005)
<i>Microsporidium prosopium</i>	<i>Prosopium williamsoni</i>	Skeletal musculature	5.6 (5–7)	3.2 (3–4)	Uninucleate, ovoid to pyriform	KENT et al. (1999)
<i>Microsporidium seriolaie</i>	<i>Seriola quinqueradiata</i>	Muscle fibres	—	—	—	EGUSA (1982)
<i>Pleistophora pagri</i>	<i>Pagrus pagrus</i>	Epithelial cells of gut and viscera in the peritoneal cavity	1.7±0.6 (1.5–2.7)	1.5±0.3 (1.2–1.8)	Ellipsoid in the peritoneal cavity	MORSY et al. (2012)
<i>Pleistophora pagri</i>	<i>Pagrus major</i>	Muscles, connective tissue and intestinal epithelium	1.7±0.6 (1.5–2.7)	1.5±0.3 (1.2–1.8)	Ellipsoid	Present study

Discussion

Totally, 156 microsporidian species have been recorded infecting fish; they are classified in 14 genera (AZEVEDO & MATOS 2002, LOM & NILSEN 2003). Infections caused by microsporidian parasites are the main cause for muscle destruction and tissue atrophy (LOM & DYKOVA 2005, STENTIFORD et al. 2013). The microsporidium recorded in the present study was detected as tumor masses embedded in muscle tissues as well as in organ epithelia; this observation is in agreement with CASAL et al. (2008) and LOVY et al. (2007). The infected fish with these parasites possesses opaque, unflashy muscles, which is unpleasant for consumers (SUTHERLAND et al. 2000, 2004). After infection, the parasite attacks host cells where they developed and proliferate (DYKOVÁ & LOM 1978). According to VAVRA & LUKES (2013), members of the Microsporidia were differentiated by the shape of spores and their polar filament coils number. The TEM investigations in the present study revealed that different developmental stages observed within the plasmodia were in accordance with those recorded previously from different microsporidian xenomas (STEPHENS 2009, ABDEL-GHAFFAR et al. 2011). The prevalence of infection for the parasites recorded was high during winter, in agreement with FIELDING et al. (2005), who reported that the infection with *P. mulleri* isolated from *Gammarus duebenicelticus* during winter was 44.9% versus 8.5 % during summer. BECKER & SPEARE (2004) reported that the plasmodia number in the infected fish decreased by decreasing water temperature.

Xenomas recovered in the present study were compared with the most similar xenoma-inducing Microsporidia. *Glugea* Thélohan, 1891 differed from the parasite recorded in this study by the presence of a surface plasmalemma with tubular extensions, numerous folds and pinocytotic vesicles. *Ichthyosporidium* Caullery & Mesnil, 1905 is distinguished by different arrangement and granular content of the developmental stages within xenomas (LOM 2002). *Microgemma* Ralphs & Matthews, 1986 possesses xenomas with microvilli-bearing coats, which are not recorded herein. *Amazonspora* Azevedo & Matos, 2003 were characterised by the presence of collagen fibres arranged as 22 crossed layers. *Nosemoides* Vinckier, 1975 possesses binucleated spores. The microsporidium spores detected in the present study are uninucleated. *Tetramicra* Matthews & Matthews, 1980 is distinguished by its entire labellum, which is the characteristic feature discriminating this genus. Sporonts represent one of the main developmental stages observed during the

TEM examination of the present ultrathin sections, while members of *Spraguea* Vávra and Sprague, 1976 lack these stages and the schizont stages develop directly into sporoblasts (MANSOUR et al. 2013). Xenomas of the present microsporidium were recorded as tumor like masses containing its developmental stages (generative cells, meronts, sporoblasts and spores); these observations coincide with those on *P. pagri* by MORSY et al. (2012), who have isolated and described previously this species from *Pagrus pagrus* in Egypt. Also, the presence of 10–15 spores encircled within sporophorous vesicles is in accordance with the description of CANNING & NICHOLAS (1980) for *P. typicalis*. The number of coils within each spore was 5–8, with electron dense centres and a polaroplast (lamellar membrane system) as the first apparent vacuole, similar to the records of LOM & CORLISS (1997) for *P. hypheobryconis* and MORSY et al. (2012) for *P. pagri*. Since the infection site and the type of developmental stages within plasmodia correspond to those recorded for *Pleistophora pagri* from *Pagrus pagrus*, we determined the microsporidium recovered from *P. major* as *P. pagri* with new host and locality records. Comparison between spore measurements for the present parasite and those recorded for different microsporidians is shown in Table 1.

Acknowledgement: The authors extend their appreciation to the Deanship of Scientific Research at King Khalid University for funding this work through Research group Project under grant number (R.G.P.1–56–39) and to the Deanship of Scientific Research at Princess Nourah Bint Abdelrahman University through the Fast-Track Research Funding Program.

References

- ABDEL-GHAFFAR F., BASHTAR A.R., MEHLHORN H., AL-RASHEID A., AL-OLAYAN E., KOURA E. & MORSY K. 2009. Ultrastructure, development, and host–parasite relationship of a new species of the genus *Pleistophora* – a microsporidian parasite of the marine fish *Epinephelus chlorostigma*. *Parasitology Research* 106: 39–46.
- ABDEL-GHAFFAR F., BASHTAR A.R., MEHLHORN H., AL-RASHEID K. & MORSY K. 2011. Microsporidian parasites: a danger facing marine fishes of the Red Sea. *Parasitology Research* 108(1): 219–225.
- ABDEL-GHAFFAR F., BASHTAR A.R., MORSY K., MEHLHORN H., AL QURAIISHY S., AL-RASHEID K. & ABDEL-GABER R. 2012. Morphological and molecular biological characterization of *Pleistophora aegyptiaca* sp. nov. infecting the Red Sea fish *Saurida tumbil*. *Parasitology Research* 110(2): 741–752.
- AWAKURA T. 1974. Studies on the microsporidian infection in salmonid fishes. *Scientific Reports of the Hokkaido Salmon Hatchery* 29: 1–93.
- AZEVEDO C. & MATOS E. 2002. Fine structure of a new spe-

- cies, *Loma myrophis* (Phylum Microsporidia), parasite of the Amazonian fish *Myrophis platyrhynchus* (Teleostei, Ophichthidae). *European Journal of Protistology* 37: 445–452.
- AZEVEDO C. & MATOS E. 2003. *Amazonspora hassar* sp. n. (phylum Microsporidia: fam. Glugeidae), a parasite of the Amazonian teleost *Hassar orestis* (Fam. Doradidae). *Journal of Parasitology* 89: 336–341.
- BECKER J.A. & SPEARE D.J. 2004. Impacts of water temperature shift on xenoma clearance and recovery time during a *Loma salmonae* (Microsporidia) infection in rainbow trout *Oncorhynchus mykiss*. *Diseases of Aquatic Organisms* 58: 185–191.
- BELL A.S., AOKI T. & YOKOYAMA H. 2001. Phylogenetic relationships among microsporidia based on rDNA sequence data, with particular reference to fish-infecting *Microsporidium* Balbiani, 1884 species. *Journal of Eukaryotic Microbiology* 48(3): 258–265.
- BUSH A.O., LAFFERTY K.D., LOTZ J.M. & SHOSTAK A.W. 1997. Parasitology meets ecology on its own terms: Margolis et al. revisited. *Journal of Parasitology* 83: 575–583.
- CANNING E.U. & NICHOLAS J.P. 1980. Genus *Pleistophora* (Phylum Microspora): re-description of the type species, *Pleistophora typicalis* Gurley, 1893 and ultrastructural characterization of the genus. *Journal of Fish Diseases* 3: 317–328.
- CASAL G., MATOS E., TELES-GRILLO M. L. & AZEVEDO C. 2008. A new microsporidian parasite, *Potaspormorhaphis* n. gen., n. sp. (Microsporidia) infecting the teleostean fish, *Potamorhaphis guianensis* from the River Amazon. *Morphological, ultrastructural and molecular characterization*. *Parasitology* 135(9): 1053–1064.
- CASAL G., MATOS E., TELES-GRILLO M.L. & AZEVEDO C. 2009. Morphological and genetical description of *Loma psittaca* sp. n. isolated from the Amazonian fish species *Colomesus psittacus*. *Parasitology Research* 105(5): 1261–71.
- CAULLERY M. & MESNIL F. 1905. Sur des haplosporidies parasites de poissons marins. *C R Social Biology* 58: 640–643.
- DYKOVÁ I. & LOM J. 1978. Tissue reaction to *Glugea plecoglossi* infection by its natural host, *Plecoglossus altivelis*. *Folia Parasitologica (Praha)* 27: 213–216.
- EGUSA S. 1982. A microsporidian species from yellowtail juveniles, *Seriola quinqueradiata*, with “Beko” disease. *Fish Pathology* 16: 187–192.
- FIELDING N. J., MACNEIL C., ROBINSON N., ELWOOD D. J. T., RW T. R. S., RUIZ Z. & DUNN A. M. 2005. Ecological impacts of the microsporidian parasite *Pleistophora mulleri* on its freshwater amphipod host *Gammarus dubeniceliticus*. *Parasitology* 131: 331–336
- GURLEY R. R. 1893. On the classification of Myxosporidia, a group of protozoan parasites infesting fishes. *Bulletin of the United States Fish Commission* 11: 407–420
- HOSHINA T. 1951. On a new microsporidian, *Pleistophora anguillarum* n. sp., from the muscle of the eel, *Anguilla japonica*. *Journal of the Tokyo University of Fisheries* 38: 35–49
- JOH S. J., KWON Y. K., KIM M. C., KIM M. J., KWON H. M. & PARK J. W. 2007. *Heterosporis anguillarum* infections in farm cultured eels (*Anguilla japonica*) in Korea. *Journal of Veterinary Science* 8(2): 147–149
- KENT M. L., DOCKER J., KHATTRA J., VOSSBRINCK C. R., SPEARE D. J. & DEVLIN R. H. 1999. A new *Microsporidium* sp. (Microsporidia) from the musculature of the mountain whitefish *Prosopium williamsoni* from British Columbia: morphology and phylogeny. *Journal of Parasitology* 85: 1114–1119
- LEIRO J., PARAMA A.Â., ORTEGA M., SANTAMARINA M. T. & SANMARTÔÂN M. L. 1999. Redescription of *Glugea caulleryi*, a microsporidian parasite of the greater sand-eel, *Hyperoplus lanceolatus* (Le Sauvage), (Teleostei: Ammodytidae), as *Microgemma caulleryi* comb. nov. *Parasitology* 22: 101–110
- LOM J. 2002. A catalogue of described genera and species of microsporidians parasitic in fish. *Systematic Parasitology* 53: 81–99.
- LOM J. & ARTHUR J. R. 1989. A guideline for the preparation of species descriptions in Myxosporia. *Parasitology* 12: 151–156.
- LOM J. & CORLISS J. O. 1997. Ultrastructural observations on the development of the microsporidian protozoon *Pleistophora hypheosobryconis* Schaperclaus. *Journal of Protozoology* 14: 141–152.
- LOM J., DYKOVA I. 2005. Microsporidian xenomas in fish seen in wider perspective. *Folia Parasitologica* 52: 69–81.
- LOM J. & NILSEN F. 2003. Fish microsporidia: fine structural diversity and phylogeny. *International Journal of Parasitology* 33: 107–127.
- LOVY J., WRIGHT G. M. & SPEARE D. J. 2007. Ultrastructural examination of the host inflammatory response within gills of netpen reared chinook salmon (*Oncorhynchus tshawytscha*) with microsporidial gill Disease. *Fish and Shellfish Immunology* 22(1–2): 131–149.
- MANSOUR L., BEN HASSINE O. K., VIVARES C. P. & CORNILLOT E. 2013. *Spraguea lophii* (Microsporidia) parasite of the teleost fish, *Lophius piscatorius* from Tunisian coasts: Evidence for an extensive chromosome length polymorphism. *Parasitology International* 62: 66–74
- MANSOUR L., PRENSIER G., JEMAA S. B., HASSINE O. K., MÉTÉNIER G., VIVARÈS C. P. & CORNILLOT E. 2005. Description of a xenoma-inducing microsporidian, *Microgemma tincae* n. sp., parasite of the teleost fish *Symphodus tinca* from Tunisian coasts. *Diseases of Aquatic Organisms* 65(3): 217–226
- MATTHEWS R. A. & MATTHEWS B. F. 1980. Cell and tissue reactions of turbot *Scophthalmus maximus* (L.) to *Tetramicra brevifilum* gen. n. sp. n. (Microspora). *Journal of Fish Diseases* 3: 495–515
- MORSY K., ABDEL-GHAFFAR F., MEHLHORN H., BASHTAR A. R. & ABDEL-GABER R. 2012. Ultrastructure and molecular phylogenetics of a new isolate of *Pleistophora pagri* sp. nov. (Microsporidia, Pleistophoridae) from *Pagrus pagrus* in Egypt. *Parasitology Research* 111:1587–1597
- MORSY K., BASHTAR A. R., ABDEL-GHAFFAR F. & AL-QURAIISHY S. 2013. Morphological and phylogenetic description of a new xenoma-inducing microsporidian, *Microsporidium aurata* nov. sp., parasite of the gilthead seabream *Sparus aurata* from the Red Sea. *Parasitology Research* 112(11): 3905–3915
- NILSEN F., ENDRESEN C. & HORDVIK I. 1998. Molecular phylogeny of microsporidians with particular reference to muscle infecting species of fishes. *Journal of Eukaryotic Microbiology* 45: 535–543
- RALPHS J. R. & MATTHEWS R. A. 1986. Hepatic microsporidiosis due to *Microgemma hepaticus* n. gen., n. sp. in juvenile grey mullet *Chelon labrosus*. *Journal of Fish Diseases* 9: 225–242

- SANO M., SATO J. & YOKOYAMA H. 1998. Occurrence of Beko disease caused by *Microsporidium seriolae* (Microspora) in hatchery-reared juvenile yellowtail. *Fish Pathology* 33: 11–16
- SMALL H. J., MEYER G. R., STENTIFORD G. D., DUNHAM J. S., BATEMAN K. & SHIELDS J. D. 2014. *Ameson metacarcini* sp. nov. (Microsporidia) infecting the muscles of Dungeness crabs *Metacarcinus magister* from British Columbia, Canada. *Diseases of Aquatic Organisms* 110(3): 213–225
- STENTIFORD G. D., FEIST S. W., STONE D. M., BATEMAN K. S. & DUNN A. M. 2013. Microsporidia: diverse, dynamic, and emergent pathogens in aquatic systems. *Trends in Parasitology* 29(11): 567–578
- STEPHENS F. J. 2009. Unidentified microsporidian in western pygmy perch *Edelia vittata* (Nannopercidae) from Australia. *Australian Veterinary Journal* 87(1): 61–62
- SUTHERLAND D., COOPER S., STELZIG P., MARCQUENSKI S., MARCINO J., LOM J., DYKOVA I., NILSEN F., HSU H.M., JAHNS W., HOYLE J. & PENNEY R. 2004. *Heterosporis* sp. (Microspora): a new parasite from yellow perch (*Perca flavescens*) and walleye (*Stizostedion vitreum*) in Minnesota, Wisconsin and Lake Ontario, North America. 13th International Conference on Aquatic Invasive Species, Ennis, County Clare, Ireland.
- SUTHERLAND D., MARCQUENSKI S., MARCINO J., LOM J., DYKOVA I., HSU H.M., JAHNS W. & NILSEN F. 2000. *Heterosporis* sp. (Microspora: Glugeidae): a new parasite from *Perca flavescens* in Wisconsin and Minnesota. Proceedings of the 62nd Midwest Fish and Wildlife Conference, Minneapolis, Minnesota.
- VAVRA J. & LUKES J. 2013. Microsporidia and ‘The Art of Living Together’ In: Rollinson D, editor. *Advances in Parasitology* 82: 253–319.
- VÁVRÁ J. & SPRAGUE V. 1976. Biology of the microsporidia. In: Bulla, L. A., Jr. & Cheng, T. C. (ed.), *Comparative Pathobiology*, Plenum Press, New York 1: 1–369.
- VINCKIER D. 1975. *Nosemoides* gen. n., *N. vivieri* (Vinckier, Devauchelle and Prensier, 1970) comb. Nov. (Microsporidie): étude de la différenciation sporoblastique et génèse des différentes structures de la spore. *Journal of Protozoology* 22: 170–184.
- WEIDNER E. 1976. Ultrastructure of the peripheral zone of the *Glugea* induced xenoma. *Journal of Protozoology* 23: 234–239.

Received: 15.07.2018

Accepted: 21.09.2018

