



Review

Pathogens of European Catfish *Silurus glanis* (L., 1758): A Review Within the One Health Approach

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Abstract

The European catfish (*Silurus glanis*, Linnaeus 1758), commonly known as the wels catfish, is one of the largest freshwater fish in Europe and an ecologically and economically important species in both natural ecosystems and aquaculture. Its broad native distribution, together with the rapid growth of farming practices, increases concerns about pathogen dissemination and their potential impact on biodiversity, animal health, and potential risks to human healthcare. This review is based on a structured literature search following PRISMA recommendations for narrative reviews and summarizes current knowledge on the main pathogen groups affecting *S. glanis*—viruses (ranaviruses, alloherpesviruses), bacteria (*Aeromonas* spp., *Edwardsiella* spp.), protozoan and metazoan parasites (*Ichthyophthirius multifiliis*, *Thaparocleidus* spp., *Eustrongylides* spp., *Contracaecum* larvae), and oomycetes (*Saprolegnia* spp., *Branchiomyces* spp.). Within the One Health approach, particular attention is given to zoonotic pathogens such as *Aeromonas* spp., *Edwardsiella tarda*, and helminths like *Eustrongylides* and *Contracaecum*, which may cause risks to human health through contaminated water or consumption of raw or undercooked fish. The review integrates findings from field surveys, regional case studies such as those from the Danube basin, and data from the authors' doctoral research. Because the wels catfish is increasingly cultivated and serves as an apex predator in natural habitats, its effective disease management is critical for both aquaculture and wild populations, and also for the food chains at all. Strengthened surveillance, health monitoring, and biosecurity measures are essential preventing the introduction and spread of pathogens into new hosts and habitats. Through the underlining of major catfish pathogen groups, this review highlights key challenges within the One Health approach and underscores the need for integrated health monitoring, biosecurity, and environmental management strategies.

Keywords: *Silurus glanis*; aquaculture; viral diseases; bacterial infections; fish parasites; oomycete infections; biodiversity; One Health



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1. Introduction

The European catfish (*Silurus glanis*, Linnaeus 1758) ranks among the largest freshwater species in Europe and plays a key role in both natural ecosystems and aquaculture [1–7]. In recent decades, farming of this species has grown steadily, supported by its rapid growth, broad environmental tolerance, and strong market demand [8]. Yet, with this expansion come serious challenges, particularly the threat of infectious diseases. Reports have documented outbreaks caused by all major groups of pathogens—viruses, bacteria, protozoan

and metazoan parasites, as well as oomycetes—with some cases resulting in heavy stock losses [9]. Historically, in traditional low-density polyculture ponds, wels catfish were even considered beneficial because they preyed on weak or diseased fish, thereby helping to reduce the spread of infections among carp [10]. Modern intensive farming systems, however, whether high-density ponds or recirculating aquaculture facilities, create conditions that favor rapid pathogen transmission, making viral and bacterial diseases especially problematic [9].

S. glanis hosts an exceptionally diverse community of parasites, a reflection of both its wide geographic distribution and predatory feeding habits. Surveys across native and introduced ranges have identified numerous taxa, including protozoans, monogeneans, digenean trematodes, cestodes, nematodes, acanthocephalans, crustaceans, and oomycetes [11]. This diversity is not only of academic interest but also of practical concern, since some pathogens directly impact catfish populations while others may carry broader ecological or even public health implications. Certain bacteria and parasites of catfish, for instance, are opportunistic or zoonotic and may infect other animals or humans through water contact or consumption of fish [12].

The One Health approach provides an essential interdisciplinary framework. One Health recognizes that the health of humans, animals, and ecosystems are connected, and the disease prevention and management require collaboration across veterinary, environmental, and public health sectors. International bodies such as the World Health Organization (WHO), Food and Agriculture Organization of the United Nations (FAO), World Organisation for Animal Health (OIE or WOAH), and United Nations Environment Programme (UNEP) promote the One Health framework through global strategies for zoonotic disease control, antimicrobial resistance mitigation, and ecosystem protection. At the regional level, the European Union (EU) has incorporated One Health principles into regulations governing aquaculture biosecurity, fish welfare, water quality, and trade. National legislations across Europe likewise require monitoring of aquatic animal diseases, responsible antimicrobial use, and environmental risk assessments—all integral components of the One Health concept.

For *S. glanis*, One Health is especially relevant because pathogen exchange can occur between wild fish, farmed stocks, invertebrate hosts, birds, and even humans in rare zoonotic cases. Managing the health of *S. glanis* is difficult because the species is exposed to a wide spectrum of pathogens. Its distribution—from natural rivers and lakes such as the Danube and its delta to artificial ponds and recirculating systems—creates multiple opportunities for infection. A thorough understanding of catfish pathogens is therefore critical for three main reasons: (1) Aquaculture—diseases can cause severe losses, and *S. glanis* often cohabits with other species, facilitating pathogen exchange. (2) Biodiversity—wild catfish may serve as reservoirs of infections that affect other fishes, while stocking or translocation can introduce novel pathogens into naïve ecosystems [13,14]. (3) Public health—although most catfish pathogens are not zoonotic, some bacteria and parasites may infect humans, especially through consumption of raw or undercooked fish, linking this issue to One Health concerns [15,16].

This review highlights the main pathogen groups reported in *S. glanis*: viruses, bacteria, oomycetes, and parasites (protozoans and helminths). For each, we outline representative species, associated diseases or pathological effects, diagnostic approaches, and implications for aquaculture and biodiversity. Emphasis is placed on recent research—more than 65% of the references are from the last five years (2021–2025)—and on findings from the species' native range, such as studies from the Danube River, to provide ecological and epidemiological context. To strengthen the systematic approach, a bibliometric analysis of the available literature is included, highlighting: (1) geographic origin of studies, (2) the

proportion conducted in natural vs. aquaculture settings, and (3) the number and diversity of pathogens documented in *S. glanis*. Furthermore, Materials and Methods and Discussion sections enhance scientific rigor and align the manuscript with recommended standards such as PRISMA guidelines [17] for structured reviews. Summary tables of key pathogens are included with a treatment options and corresponding references. By consolidating this information, we aim to support fish health specialists and researchers working in catfish culture and fisheries management, while identifying gaps that merit further study. Understanding the full spectrum of catfish pathogens is essential for effective disease management and for appreciating the role of *S. glanis* within aquatic health networks.

2. Materials and Methods

2.1. Literature Search Strategy

A structured literature search was performed following PRISMA recommendations for narrative reviews [17]. Databases consulted included Web of Science, Scopus, PubMed, and Google Scholar, and author’s PhD thesis. Searches covered publications from 1980 to 2025 and used combinations of keywords such as “*Silurus glanis*”, “European catfish”, “wels catfish”, “pathogens”, “viruses”, “bacteria”, “parasites”, “helminths”, “oomycetes”, “aquaculture diseases”, and “One Health”. Search terms, platforms, and timeframes are summarized in Tables 1–4. Titles and abstracts were screened independently by both authors using predefined inclusion and exclusion criteria. All retrieved records were combined and duplicates were removed. Full texts considered potentially relevant were subsequently reviewed in detail by the same two authors. Disagreements at any stage were resolved through discussion until full consensus was achieved. No automation tools were used during any part of the Literature Search Strategy. In accordance with the Journal’s Recommendations more than 50% of the cited references are within the last 5 years (2021–2025). Findings on key pathogen groups are synthesized in summary tables (Tables 5–9) that include affected tissues, clinical signs, zoonotic potential, treatment, and corresponding references.

Table 1. Keywords combinations and total number of results in Web of Science.

Keywords	All Articles	Article	Review Article	Date of the Last Search
<i>Silurus glanis</i> , pathogens	15	14	1	9 September 2025
European catfish, pathogens	59	51	8	9 September 2025
wels catfish, pathogens	2	2	2	9 September 2025
<i>Silurus glanis</i> , viruses	31	3	28	9 September 2025
European catfish, viruses	59	51	8	9 September 2025
wels catfish, viruses	3	3	0	9 September 2025
<i>Silurus glanis</i> , bacteria	11	10	1	9 September 2025
European catfish, bacteria	47	42	5	9 September 2025
wels catfish, bacteria	2	2	0	9 September 2025
<i>Silurus glanis</i> , parasites	30	28	2	9 September 2025
European catfish, parasites	94	87	7	9 September 2025
wels catfish, parasites	7	7	0	9 September 2025
<i>Silurus glanis</i> , helminths	5	5	0	9 September 2025
European catfish, helminths	7	7	0	9 September 2025
wels catfish, helminths	2	2	0	9 September 2025

Table 1. Cont.

Keywords	All Articles	Article	Review Article	Date of the Last Search
<i>Silurus glanis</i> , oomycetes	1	1	0	9 September 2025
European catfish, oomycetes	1	1	0	9 September 2025
wels catfish, oomycetes	0	0	0	9 September 2025
<i>Silurus glanis</i> , aquaculture diseases	19	15	4	9 September 2025
European catfish, aquaculture diseases	79	66	13	9 September 2025
wels catfish, aquaculture diseases	2	2	0	9 September 2025
<i>Silurus glanis</i> , One Health	15	15	0	9 September 2025
European catfish, One Health	36	32	4	9 September 2025
wels catfish, One Health	2	2	0	9 September 2025

Table 2. Keywords combinations and total number of results in Scopus.

Keywords	All Articles	Article	Review Article	Date of the Last Search
<i>Silurus glanis</i> , pathogens	15	12	3	10 September 2025
European catfish, pathogens	22	14	8	10 September 2025
wels catfish, pathogens	1	1	0	10 September 2025
<i>Silurus glanis</i> , viruses	31	26	5	10 September 2025
European catfish, viruses	41	33	8	10 September 2025
wels catfish, viruses	6	6	0	10 September 2025
<i>Silurus glanis</i> , bacteria	13	13	0	10 September 2025
European catfish, bacteria	21	18	3	10 September 2025
wels catfish, bacteria	4	4	0	10 September 2025
<i>Silurus glanis</i> , parasites	34	33	1	10 September 2025
European catfish, parasites	33	32	1	10 September 2025
wels catfish, parasites	9	9	0	10 September 2025
<i>Silurus glanis</i> , helminths	7	7	0	10 September 2025
European catfish, helminths	2	2	0	10 September 2025
wels catfish, helminths	2	2	0	10 September 2025
<i>Silurus glanis</i> , oomycetes	2	1	1	10 September 2025
European catfish, oomycetes	1	1	0	10 September 2025
wels catfish, oomycetes	0	0	0	10 September 2025
<i>Silurus glanis</i> , aquaculture diseases	14	12	2	10 September 2025
European catfish, aquaculture diseases	26	17	9	10 September 2025
wels catfish, aquaculture diseases	2	2	0	10 September 2025
<i>Silurus glanis</i> , One Health	8	7	1	10 September 2025
European catfish, One Health	15	12	3	10 September 2025
wels catfish, One Health	1	1	0	10 September 2025

Table 3. Keywords combinations and total number of results in PubMed.

Keywords	All Articles	Article	Review Article	Date of the Last Search
<i>Silurus glanis</i> , pathogens	11	10	1	11 September 2025
European catfish, pathogens	18	14	4	11 September 2025
wels catfish, pathogens	2	2	0	11 September 2025
<i>Silurus glanis</i> , viruses	14	13	1	11 September 2025
European catfish, viruses	26	24	2	11 September 2025
wels catfish, viruses	3	3	0	11 September 2025
<i>Silurus glanis</i> , bacteria	22	21	1	11 September 2025
European catfish, bacteria	34	32	2	11 September 2025
wels catfish, bacteria	6	6	0	11 September 2025
<i>Silurus glanis</i> , parasites	26	25	1	11 September 2025
European catfish, parasites	26	23	3	11 September 2025
wels catfish, parasites	8	8	0	11 September 2025
<i>Silurus glanis</i> , helminths	17	17	0	11 September 2025
European catfish, helminths	13	12	1	11 September 2025
wels catfish, helminths	4	4	0	11 September 2025
<i>Silurus glanis</i> , oomycetes	1	1	0	11 September 2025
European catfish, oomycetes	2	2	0	11 September 2025
wels catfish, oomycetes	0	0	0	11 September 2025
<i>Silurus glanis</i> , aquaculture diseases	5	4	1	11 September 2025
European catfish, aquaculture diseases	13	11	2	11 September 2025
wels catfish, aquaculture diseases	0	0	0	11 September 2025
<i>Silurus glanis</i> , One Health	5	5	0	11 September 2025
European catfish, One Health	9	8	1	11 September 2025
wels catfish, One Health	2	2	0	11 September 2025

Table 4. Keywords combinations and total number of results in Google Scholar.

Keywords	All Articles	Article	Review Article	Date of the Last Search
<i>Silurus glanis</i> , pathogens	1770	1583	187	12 September 2025
European catfish, pathogens	1070	938	132	12 September 2025
wels catfish, pathogens	325	277	48	12 September 2025
<i>Silurus glanis</i> , viruses	894	795	99	12 September 2025
European catfish, viruses	622	544	78	12 September 2025
wels catfish, viruses	151	129	22	12 September 2025
<i>Silurus glanis</i> , bacteria	2390	2193	197	12 September 2025
European catfish, bacteria	1310	1186	124	12 September 2025
wels catfish, bacteria	416	362	54	12 September 2025
<i>Silurus glanis</i> , parasites	2160	1983	177	12 September 2025
European catfish, parasites	917	823	94	12 September 2025

Table 4. Cont.

Keywords	All Articles	Article	Review Article	Date of the Last Search
wels catfish, parasites	406	361	45	12 September 2025
<i>Silurus glanis</i> , helminths	406	376	30	12 September 2025
European catfish, helminths	122	111	11	12 September 2025
wels catfish, helminths	54	49	5	12 September 2025
<i>Silurus glanis</i> , oomycetes	62	55	7	12 September 2025
European catfish, oomycetes	49	41	8	12 September 2025
wels catfish, oomycetes	11	8	3	12 September 2025
<i>Silurus glanis</i> , aquaculture diseases	14	14	0	12 September 2025
European catfish, aquaculture diseases	9	9	0	12 September 2025
wels catfish, aquaculture diseases	2	2	0	12 September 2025
<i>Silurus glanis</i> , One Health	234	223	11	12 September 2025
European catfish, One Health	108	98	10	12 September 2025
wels catfish, One Health	22	18	4	12 September 2025

2.2. Inclusion and Exclusion Criteria

Included sources met at least one of the following criteria: (1) Reported pathogens detected in *S. glanis*. (2) Described clinical signs, pathology, or epidemiology in wild or farmed catfish. (3) Examined One Health-related aspects such as zoonotic potential, environmental drivers, or biosecurity. (4) Provided molecular characterisation, distribution data, or experimental infection results. (5) Papers were published in peer-reviewed scientific journals. (6) Papers were published within the last five years (2021–2025).

Excluded criteria were: Reason (1) Studies lacking confirmed identification of pathogens. Reason (2) Reports unrelated to fish health or aquaculture. Reason (3) Literature without accessible methodological details. Reason (4) Papers were published in non-peer-reviewed journals. Reason (5) Papers were published before the last five years (2021–2025).

2.3. Compliance with the PRISMA Guidelines

The review process complies fully with the PRISMA guidelines [17]. A completed PRISMA 2020 flow diagram is provided in the manuscript (Figure 1), illustrating all stages of record identification, screening, eligibility assessment, and inclusion. Because the figure reports the full numerical details, these numbers are not repeated here in the text. This review was not registered in a public repository.

2.4. Bibliometric Summary

Analysis of the literature indicates that approximately 60% of the studies originate from Europe, predominantly Central and Eastern regions—Hungary, Czech Republic, Bulgaria, Germany, etc. Around 25% come from Western Asia—Turkey and Iran, and the remaining 15% come from other regions including China. Roughly 55% of the studies were conducted in wild habitats and 45% in aquaculture ponds. Across all sources, more than 90 pathogens of different taxonomic groups have been recorded in *S. glanis*, highlighting a rich and complex disease landscape but some of the pathogens were mentioned only once as an incident finding or a susceptible species.

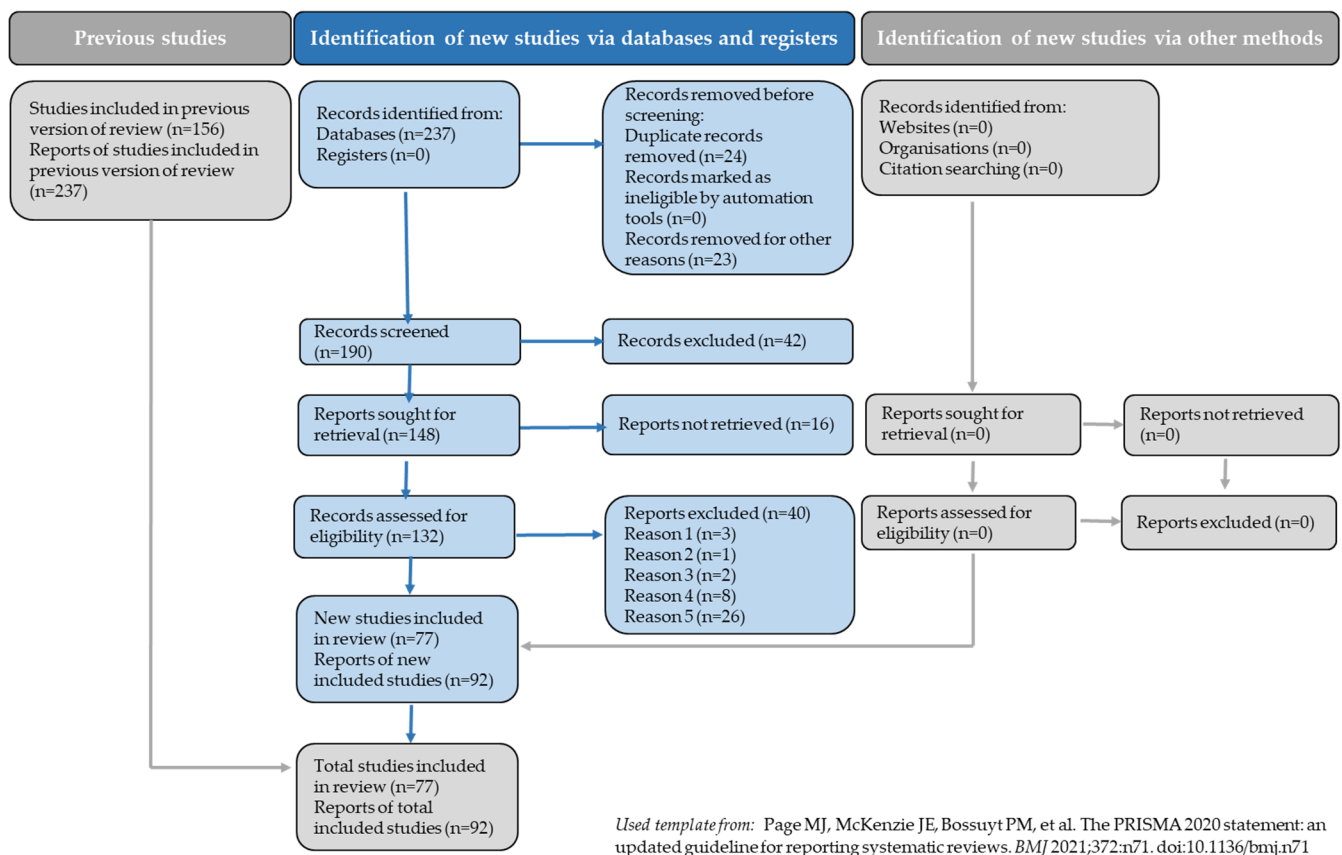


Figure 1. PRISMA flow diagram (used template from [17]).

3. Results

3.1. Viral Pathogens

Reports of viral infections in *S. glanis* are relatively scarce, yet several are known to be highly virulent. An earlier review [18] observed that only a few finfish viruses significantly affect Siluriformes, notably a herpesvirus and two iridoviruses associated with disease in catfish. More recent studies confirm that knowledge of *S. glanis* virology is still limited. The main documented infections involve alloherpesviruses (family *Alloherpesviridae*) and ranaviruses (family *Iridoviridae*), while newer reports also describe papillomaviruses, circoviruses, and rhabdoviruses in this host [19–23].

3.1.1. Alloherpesviruses

Herpesviruses were among the earliest viruses identified in *S. glanis*. In the 1980s, a herpesvirus infection causing epidermal hyperplasia—commonly known as “fish pox,” with papilloma-like skin lesions—was documented in Hungary [24]. This condition, often referred to as sheatfish herpesvirus disease, typically produces proliferative lesions on the skin and fins. Although generally benign, such lesions can predispose fish to secondary infections. The suspected causative agent, likely an alloherpesvirus, could not be cultured at the time and remained poorly described. Decades later, molecular studies revealed a novel alloherpesvirus in farmed *S. glanis* displaying widespread carp pox-like skin lesions [22]. Partial sequencing confirmed it as a distinct member of the family *Alloherpesviridae* [20]. Phylogenetic analysis indicates that while it is related to other fish herpesviruses, it represents a unique lineage in catfish. This emerging virus, proposed as “*Silurid herpesvirus*,” highlights that *S. glanis* can harbor its own herpesvirus clade. Clinical cases in adults appear limited to benign skin tumors, but its effects on juvenile survival and latency within populations remain unresolved [25,26]. Ongoing monitoring is important,

since new variants could evolve or spread, potentially reducing existing disease resistance in cultured stocks.

3.1.2. Iridoviruses

Systemic infections caused by iridoviruses represent a serious threat to both catfish hatcheries and wild populations. *S. glanis* is susceptible to certain ranaviruses (*Iridoviridae*) that induce acute hemorrhagic disease. The first reported outbreak occurred in Germany in 1988, when fry suffered mortality rates approaching 100% [27]. The isolated virus, later termed European sheatfish virus (ESV), proved highly virulent, killing fry within days of exposure through waterborne transmission [28]. Affected fingerlings displayed lethargy, loss of equilibrium, petechial skin hemorrhages, exophthalmia, and severe necrosis of internal organs [29]. Histopathological studies showed that ESV targets endothelial and hematopoietic tissues, leading to edema and hemorrhagic shock [28,30]. Older fish may survive infection but act as carriers, particularly when stressed or co-infected with bacteria [31].

A related ranavirus, European catfish virus (ECV), was detected in wild populations across Europe in the 1990s, with initial cases in France and Italy causing mass mortality in lakes where invasive black bullhead (*Ameiurus melas*) were abundant [32–34]. Although ESV and ECV were initially considered the same, genetic studies later distinguished them: differences in neurofilament protein genes and host range confirmed that they are separate strains [35]. Both belong to the amphibian-like ranavirus clade and are closely related to epizootic hematopoietic necrosis virus (EHNV) from Australia [36]. Full genome sequencing of ESV revealed approximately 136 putative genes and about 88% nucleotide identity to EHNV [36], while ECV is presumed to have a similarly large double-stranded DNA genome. These ranaviruses are now recognized as significant emerging pathogens of European catfish, spreading rapidly in warm-water conditions and causing high juvenile mortality, with the potential to infect other species such as bullheads [9]. Alarming, ECV was even identified in ornamental *Pangasius* shipments imported to South Korea [37]. Given their impact, strict biosecurity and the development of vaccines are strongly recommended to control ranavirus outbreaks in cultured and wild catfish.

3.1.3. Papillomaviruses

In addition to the high-mortality agents such as ranaviruses and herpesviruses, European catfish also harbor other viruses of interest. A recently described example is a papillomavirus named *Silurus glanis* papillomavirus 1 (SgPV1) [22]. This small circular DNA virus (~5.6 kb) was detected in skin lesions of farmed catfish that closely resembled those caused by herpesvirus infections [22,24]. Genomic analysis showed that SgPV1 is distinct from known mammalian papillomaviruses and has been proposed as the founding member of a new genus (*Nunpapillomavirus*) within the family Papillomaviridae [22,38]. Infected fish typically develop multiple benign papilloma-like growths on the skin. Although these tumors are non-lethal, they may interfere with respiration or osmoregulation and reduce marketability due to their appearance. The discovery of SgPV1 expands the known host range of papillomaviruses and demonstrates that *S. glanis* can be co-infected with multiple tumorigenic viruses (e.g., herpes- and papillomaviruses) [39]. Co-infections have already been documented, though the precise role of each virus in lesion formation remains uncertain.

3.1.4. Circoviruses

Circoviruses represent another emerging group of fish pathogens. A circovirus later named *Silurus glanis* circovirus (CfCV) was first reported in adult European catfish from Lake Balaton, Hungary, around 2011 [40]. Its complete genome (~1.97 kb) was sequenced

and found to encode the characteristic replicase (Rep) and capsid (Cap) proteins of circoviruses [40–42]. Detection coincided with unexplained summer mortalities in brood-fish, although a direct causal role was not firmly established [40]. Current evidence suggests that circoviruses in fish tend to cause subclinical, persistent infections, with clinical signs appearing mainly under stress or in the presence of coinfections [40]. In *S. glanis*, CfCV has not yet been linked to specific pathology, but its occurrence highlights the hidden diversity of the catfish virome and the possibility that apparently healthy populations may still harbor latent viral agents.

3.1.5. Rhabdoviruses

Rhabdoviruses have also been sporadically reported in European catfish. Some studies describe rhabdovirus-like particles resembling vesiculoviruses or novirhabdoviruses in diseased *S. glanis* showing hemorrhagic symptoms [42–44]. However, no catfish-specific rhabdovirus has yet been isolated or fully characterized. It is possible that catfish act as incidental hosts for certain aquatic rhabdoviruses, such as spring viremia of carp virus or pike fry rhabdovirus, particularly in polyculture systems. Further investigation is needed to clarify their role and potential impact on *S. glanis*.

Overall, beyond ranaviruses and alloherpesviruses, knowledge of viruses in *S. glanis* remains limited. The evidence so far indicates that other viruses—such as papillomaviruses, circoviruses, and rhabdoviruses—usually cause subacute or chronic effects rather than sudden, large-scale mortality. Nevertheless, their presence contributes to the overall pathogen burden and may interact with environmental stressors or other infections to compromise catfish health. Continuous surveillance is therefore important to track these agents and to understand their potential role in shaping disease dynamics in both farmed and wild populations. A summary of the key viral pathogens identified in *S. glanis* is presented in Table 5.

Table 5. Key viral pathogens of *S. glanis*.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
Silurid herpesvirus 2	Double-stranded DNA virus (<i>Herpesviridae</i> -like)	Skin, gills, fins	Grey-white patches, papilloma-like skin lesions, epidermal changes, and gill pathology	No	No specific antiviral; supportive care; reduction in stress	[20,24]
European catfish virus (ECV)	Double-stranded DNA virus (Ranavirus)	Kidney, spleen, liver, hematopoietic tissue, endothelium	High fry mortality, anorexia, apathy, ataxia, abnormal swimming, skin petechiae, swollen pale liver and spleen, edema, hemorrhagic diathesis, circulatory failure	No	Biosecurity, water disinfection, temperature management	[21,32]
European sheatfish virus (ESV)	Double-stranded DNA virus (Ranavirus)	Kidney, spleen, liver, hematopoietic tissue, endothelium	High fry mortality, anorexia, apathy, ataxia, abnormal swimming, skin petechiae, swollen pale liver and spleen, edema, hemorrhagic diathesis, circulatory failure	No	Biosecurity, sanitation, limiting fish movements	[27,28]
<i>Silurus glanis</i> papillomavirus 1 (SgPV1)	Double-stranded DNA virus (Papillomavirus)	Skin, epidermis	Papilloma-like epidermal hyperplasia, skin tumors	No	No specific treatment; improved hygiene	[22]
<i>Silurus glanis</i> circovirus (CfCV)	Single-stranded DNA virus (Circovirus)	Liver, spleen, kidney, gills, gonads	Mortality during spawning season, dermal lesions, vascular dilations in skin, gastrointestinal tract inflammation, and nuclear fragmentation in hematopoietic cells	No	No targeted therapy; avoid stress	[40]
Spring viremia of <i>Rhabdovirus carpio</i> -like	Single-stranded RNA virus (Rhabdovirus)	Blood, kidney, liver, spleen	Hemorrhages on skin and organs, anemia, and mortality	No	Water quality management; biosecurity	[43]

3.2. Bacterial Pathogens

Bacterial infections in European catfish most often emerge under conditions of intensive rearing or environmental stress and are typically expressed as ulcerative or septicemic syndromes. Many of the bacteria that affect *S. glanis* are opportunistic species commonly found in freshwater habitats, but the species is also vulnerable to several well-known fish pathogens. Unlike the North American channel catfish (*Ictalurus punctatus*), which suffers from the host-specific *Edwardsiella ictaluri*, the European catfish has no endemic bacterium of comparable impact [45]. Instead, outbreaks in *S. glanis* usually involve generalist pathogens that exploit suboptimal rearing conditions.

3.2.1. *Aeromonas* Genus

Among bacterial threats to *S. glanis*, motile *Aeromonas* species are considered the most common. *Aeromonas hydrophila* and related taxa, such as *A. veronii*, are responsible for motile *Aeromonas* septicemia (MAS), which is characterized by hemorrhagic skin lesions, abdominal swelling, and septicemia. A severe outbreak of MAS was reported in 2016 at a catfish farm in Tianjin, China, where *A. veronii* biovar *sobria* was identified as the causative agent [46]. Diseased fish showed petechial hemorrhages, ascites, and inflamed vent regions, and experimental infections reproduced high mortality with an LD50 of $\sim 1.6 \times 10^6$ CFU/mL in zebrafish [46]. Although *Aeromonas* spp. are opportunistic and tend to proliferate under poor water quality or high organic loads, virulent strains can cause acute epidemics in catfish, especially at higher temperatures. These bacteria also pose a zoonotic risk, as *Aeromonas* can infect humans, causing gastroenteritis, wound infections, or septicemia [16,47]. Furthermore, the emergence of antibiotic-resistant strains in aquaculture highlights the need for careful antimicrobial use and the exploration of alternatives such as vaccines or probiotics [48].

3.2.2. *Edwardsiella* Genus

Species of the genus *Edwardsiella* have occasionally been isolated from *S. glanis*. *Edwardsiella tarda*, known to cause emphysematous putrefactive disease in fish, has been recovered from European catfish showing septicemia with hemorrhagic signs [45]. Although infections by *E. tarda* in *S. glanis* are relatively uncommon, the bacterium is widely distributed and infects many freshwater fish species [49,50]. By contrast, *Edwardsiella ictaluri*, the causative agent of enteric septicemia of catfish (ESC) in North American ictalurids, has not yet emerged as a significant pathogen in European catfish aquaculture [51,52]. This absence may reflect both geographic and phylogenetic differences. Nonetheless, the possibility of ESC introduction into *S. glanis* populations through trade or co-culture cannot be excluded. Given the devastating impact of *E. ictaluri* on channel catfish farming, continuous monitoring remains essential.

3.2.3. *Flavobacterium* Genus

Opportunistic bacteria of the genus *Flavobacterium* are also frequently associated with disease in European catfish. *Flavobacterium columnare*, the causative agent of columnaris disease, produces characteristic yellowish-brown lesions on the skin and gills [53]. In *S. glanis*, *F. columnaris* outbreaks are most common under warm and crowded conditions, where gill tissues often show necrotic patches and a typical “saddleback” lesion may appear along the dorsum [54].

Many bacterial infections in catfish are linked to stress factors such as handling, crowding, or temperature fluctuations, which compromise immune defenses and trigger disease [55]. Mixed bacterial infections are not unusual, making it difficult to identify the primary pathogen. Standard preventive measures—good husbandry practices, adequate

nutrition, and minimizing stress—are essential to reduce the risk. Vaccination against bacteria such as *Aeromonas* or *Flavobacterium* is under investigation, but no commercial vaccines are yet widely used in *S. glanis* aquaculture. Table 6 summarizes key bacterial pathogens of *S. glanis*.

Table 6. Key bacterial pathogens of *S. glanis*.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Aeromonas hydrophila</i> (Chester, 1901) Stanier, 1943 (motile <i>Aeromonas</i> septicemia, MAS)	Gram-negative bacteria	Skin, gills, kidney, liver, spleen, blood	Hemorrhages on skin and fins, ulcers, exophthalmia, ascites, septicemia, and high mortality	Opportunistic	Oxytetracycline; florfenicol; improved water quality	[47,55]
<i>Aeromonas veronii</i> Hickman-Brenner et al., 1988	Gram-negative bacteria	Skin, intestine, blood	Septicemia, hemorrhages, internal organ necrosis	Opportunistic	Antibiotic therapy (sensitivity-guided), probiotics	[47,48]
<i>Aeromonas veronii</i> biovar sobria Hickman-Brenner et al., 1988	Gram-negative bacteria	Skin, intestine, blood	Septicemia, hemorrhages, internal organ necrosis	Opportunistic	Antimicrobials; reduction in stressors	[46]
<i>Edwardsiella ictaluri</i> Hawke et al., 1981 (enteric septicemia of catfish, ESC)	Gram-negative bacteria	Intestine, liver, spleen, and kidney	Ulcerations, granulomas, septicemia, abnormal swimming, anorexia	Opportunistic	Vaccination (channel catfish models), antimicrobials	[51,52]
<i>Edwardsiella tarda</i> Ewing and McWhorter, 1965	Gram-negative bacteria	Intestine, liver, spleen, kidney, muscle	Abscesses in internal organs, hemorrhages, and chronic systemic infection	Yes	Antibiotics (based on antibiogram)	[49,50]
<i>Flavobacterium columnare</i> (Bernardet and Grimont, 1989) Bernardet et al., 1996 (Columnaris disease)	Gram-negative bacteria	Skin, gills, fins	Gill necrosis, skin erosion, fin rot, and high mortality in warm water	No	Hydrogen peroxide; copper; enhanced aeration	[53,55]

3.3. Parasitic Pathogens

Parasites constitute a significant component of the disease burden in European cat-fish. Owing to its predatory habits and benthic lifestyle, *S. glanis* can acquire a wide array of parasites through the food chain and environment. Parasite infestations in catfish are particularly relevant in extensive or natural settings, but they can also pose problems in aquaculture (especially for juvenile fish).

3.3.1. Protozoan Parasites

Ichthyophthirius Genus

Ichthyophthirius multifiliis, the causative agent of white spot disease or ichthyophthiriasis, is arguably the most problematic protozoan parasite for *S. glanis*. *Ichthyophthirius* is an obligate ectoparasitic ciliate that infects skin and gills of freshwater fish [56]. European catfish are highly susceptible to *Ichthyophthirius*, particularly in fingerling stages. The parasite forms characteristic white cysts (0.5–1 mm) embedded in the epithelium. The intense irritation caused by *Ichthyophthirius* often prompts fish to flash or scrape against surfaces. If one observes such behavior in catfish along with white spots on the skin, an ichthyophthiriasis outbreak is likely underway and requires prompt intervention to save the stock. Heavy infestations lead to irritation, excess mucus production, breathing difficulty, and often death if untreated. Historically, outbreaks caused significant fingerling mortalities in wels catfish pond culture [57]. Treatments with novel compounds could be effective, but the ban of some chemicals in aquaculture fish food has made disease control more challenging. Today, prevention via quarantine and environmental management (e.g., avoiding sudden temperature drops that favor parasite reproduction) is key, and some newer therapeutics (like formalin, hydrogen peroxide, or herbal extracts, etc.) are used with varying success [10,58–60].

Trichodina Genus

Another common protozoan parasite is *Trichodina* spp., a genus of disk-shaped peritrichous ciliates. *Trichodina* are found on the skin and gills of catfish as ectocommensals or mild parasites [61]. In low numbers, it usually does not cause clinical disease, but heavy infestations can irritate the gills, causing respiratory stress and poor growth. Trichodinosis often indicates deteriorating water quality or high organic load, as these ciliates proliferate in such conditions [62]. Under the microscope, *Trichodina* can be identified by its distinctive circular denticulate ring. In *S. glanis* hatcheries, routine checks for *Trichodina* on gill/skin scrapes are recommended, and if they are present, a corrective action, such as improving water quality or applying a mild disinfectant, can prevent more serious issues.

Flagellated Protozoa

Various flagellated protozoa may inhabit the gastrointestinal tract of catfish, though reports are less common. Genera such as *Hexamita* and *Spironucleus* (collectively hexamitids) are known to cause intestinal parasitosis in other fishes (e.g., cichlids) and have occasionally been noted in catfish intestines [63]. *Ichthyobodo necator* (syn. *Costia necatrix*) is observed on the epithelium of a catfish gill [64]. Clinical hexamitosis in *S. glanis* would manifest as emaciation and malabsorption, but detailed studies in this host are lacking.

Blood Parasites

Blood parasites have also been documented in *S. glanis*. Trypanosomes (genus *Trypanosoma*) are flagellate protozoa transmitted by leeches that infect the blood of many freshwater fish, including *S. glanis* [65]. These blood parasites rarely cause overt disease in healthy adult catfish, but in young or stressed fish, they might contribute to anemia or lethargy. Similarly, *S. glanis* may host *Trypanoplasma* (also known as *Cryptobia*), another group of blood flagellates [66]. The ecological relevance of these protozoans is notable: they illustrate the catfish's role as a reservoir for parasites that involve intermediate hosts (leeches) and potentially other vertebrates.

Myxobolus Genus

Another protozoan gill parasite found in *S. glanis* is the spore-forming *Myxobolus pfeifferi* [61]. It parasitizes a wide range of fish hosts, causing them the disease known as myxoboliosis. Fish have difficulty in swimming and a slow growth rate. *Myxobolus* passes through two stages of development in its biological cycle: vegetative and spore. The spore stage represents both a form of resistance and reproduction [67].

S. glanis does not have many host-specific protozoan pathogens, but it could be used as a reservoir host that transmits parasites, which can cause serious losses if not properly managed. Table 7 summarizes key protozoan parasites of *S. glanis*.

Table 7. Key protozoan parasites of *S. glanis*.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Ichthyophthirius multifiliis</i> Fouquet, 1876 (white spot disease)	Ciliate protozoan	Skin, gills, fins	White cyst-like spots on skin and gills, respiratory distress, anorexia, and high mortality in fry	No	Formalin; hydrogen peroxide; increased temperature (controlled)	[10,56]
<i>Trichodina</i> spp. Ehrenbeg, 1830	Ciliate protozoan	Skin, gills	Excess mucus, frayed fins, skin irritation, reduced growth, respiratory problems	No	Salt baths; improved water quality	[57,62]
<i>Hexamita</i> spp. Dujardin 1838	Flagellated protozoan	Intestine	Intestinal catarrh, malabsorption, weight loss, anorexia; in severe cases, systemic infection	Possible	Metronidazole; sanitation	[63]

Table 7. Cont.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Ichthyobodo necator</i> (Henneguy, 1883) Pinto, 1928	Flagellated protozoan	Skin, gills	Skin opacity, frayed fins, respiratory distress, high fry mortality	No	Formalin; improved aeration	[64]
<i>Trypanosoma</i> spp. Gruby, 1843	Flagellated protozoan	Blood	Weakness, anemia, reduced growth; often subclinical	No	Typically none; vector control (leeches)	[65]
<i>Myxobolus</i> spp. Bütschli, 1882	Spore-forming	Skin, gills, muscle, internal organs	Cysts/tumors in tissues, growth retardation, deformities	No	No direct treatment; pond management	[66,67]

3.3.2. Helminths and Other Metazoan Parasites

European catfish harbor a rich assemblage of helminth parasites—worms that infect various organs [68]. Many of them have complex life cycles involving intermediate hosts, reflecting *S. glanis*' position as an apex predator that accumulates parasites from the food chain, and some can infect humans via raw or undercooked fish.

Monogenean Parasites

Monogenean parasites are among the most important ectoparasites of the European catfish, colonizing the gills, skin, and fins and often causing significant pathological changes. Two species of particular importance are *Thaparocleidus siluri* and *Thaparocleidus vistulensis* (formerly genus *Dactylogyrus* or *Ancylodiscoides*). These monogeneans are flatworms with a direct life cycle (no intermediate hosts) and attach to gill tissue via hooks, feeding on mucus and blood. Together, these monogeneans represent a significant health concern for European catfish [69]. *Thaparocleidus siluri* is a gill monogenean specific to European catfish. Surveys of aquaculture systems report it frequently co-occurring with other monogeneans, where it contributes to hyperplasia, mucus hypersecretion, and respiratory impairment, particularly in warm-water or intensive culture environments [70]. *Thaparocleidus vistulensis* is another major gill parasite of *S. glanis*. Light infestations may go unnoticed, but the heavy loads cause gill monogenosis, leading to hyperplasia of gill epithelium, anemia, and respiratory distress. Infected gills show pale patches and increased mucus, and fish may exhibit rapid gill movements or gasp at the surface due to hypoxia. Histopathological studies have documented deep anchor penetration into lamellae, causing fusion, clubbing, and intralamellar hemorrhages [57,71]. Experimental work has shown its high fecundity and resilience under varying temperature and light conditions, supporting its capacity to thrive in aquaculture. Monogeneans proliferate in varying light–dark conditions and water at 10–30 °C, and the parasite's fecundity is optimal at 15 °C, eggs hatched fastest at 30 °C, whereas no hatching occurred at 5 °C and 35 °C [72]. Standardized cohabitation-based sampling methods have been developed to study their life cycle and spatial distribution on gills, typically preferring mid-distal sectors [71]. An outbreak of *T. vistulensis* in a catfish farm can stunt growth and predispose fish to bacterial gill infections. Control is usually achieved by periodic antiparasitic treatments (e.g., formalin, praziquantel, etc.) and by avoiding the introduction of wild fish that carry these worms. Recent studies have examined the pathology of *Thaparocleidus* on catfish gills and noted that even moderate infections can provoke inflammation and secondary bacterial invasion [73,74]. Thus, managing monogeneans is an important aspect of catfish health. The *Thaparocleidus* genus represents a challenge for the aquaculture of European catfish. Although species are not zoonotic, their impact on fish welfare, growth, and susceptibility to other pathogens makes them economically significant. Ongoing studies suggest that routine monitoring, molecular identification, and improved management practices are essential for controlling monogenean infections in catfish farms [73].

Digenean Trematode Parasites

Digenean trematode parasites of European catfish include *Diplostomum* and *Orientocreadium* species, which affect different organs and impact fish health [68]. *Diplostomum spathaceum* is a trematode widely distributed in freshwater ecosystems. Its metacercariae migrate to the eye lens, causing “eye fluke” disease or diplostomosis, causing cataracts, impaired vision, and behavioral changes. *S. glanis* can accumulate *Diplostomum* cysts in its lens or vitreous humor if it inhabits waters with the snail and bird hosts that complete the fluke’s life cycle [75]. While vision is not the primary hunting sense for catfish (which rely more on smell and mechanosensation), severe eye damage can still stress the fish and make them more prone to predation or reduced foraging efficiency. *Orientocreadium siluri* is another specific species found primarily in the gastrointestinal tract, especially in the mid- and foregut of European catfish in Asia and Eastern Europe [11,68,76].

Cestodean Parasites

Cestodes (tapeworms) are frequently recorded in European catfish. Catfish are definitive hosts for certain tapeworms. One example is *Silurotaenia siluri*, a specific tapeworm of wels catfish [77,78]. Adult *S. siluri* live in the catfish intestine, sometimes reaching considerable lengths [68]. They absorb nutrients through their tegument, potentially causing nutritional robbing and mechanical irritation. *Proteocephalus osculatus* is another cestode often found in *S. glanis* intestines [78]. *Triaenophorus meridionalis* and *Caryophyllaeus laticeps* are other cestodes found in *S. glanis* intestine [68]. Heavy tapeworm burdens can lead to emaciation or intestinal blockage in severe cases, although mild infections may be asymptomatic. These tapeworms have complex life cycles involving intermediate hosts like copepods (first host) and fish (second host). Thus, their presence in catfish indicates a long ecological chain and a healthy population of intermediate hosts in the ecosystem. Control in aquaculture is rarely needed unless catfish are fed raw fish containing larval cestodes; in such cases, freezing the feed fish can break the cycle.

Nematodean Parasites

Nematodes (roundworms) form a large portion of the catfish parasite fauna. A comprehensive review discusses 21 species of nematodes infecting *S. glanis* [79]. Common examples include *Raphidascaris acus* (a gastrointestinal nematode) and larvae of *Eustrongylides* spp. (a tissue nematode). *R. acus* adults reside in the catfish intestine, where they attach to the mucosa. They can cause inflammation, granulomas, and occasional perforation of the gut wall when present in high numbers. This nematode is acquired when catfish eat infected intermediate hosts (small fish carrying larvae), and it has been reported widely in Europe and Asia in wels catfish [11,57,79,80]. Perhaps more dramatic are the larvae of *Eustrongylides* species, specifically *Eustrongylides excisus*, a large red nematode with a life cycle involving aquatic birds. Catfish can serve as paratenic or second intermediate hosts for their larvae, which typically embed in the body cavity or musculature. These larvae can grow several centimeters long and cause granulomatous inflammation in the tissues where they encyst. In infected *S. glanis*, one might notice coiled red worms in the visceral cavity during necropsy [11,68,79,81]. While often not causing acute death, *E. excisus* can weaken the fish and render it unmarketable. Importantly, *E. excisus* is a zoonotic parasite—humans who consume raw or undercooked fish containing these larvae can develop a serious illness (gastrointestinal or tissue-invasive eustrongylidosis). Although human cases are rare, the presence of *E. excisus* in food fish like catfish necessitates proper evisceration and cooking. Catfish farmers mitigate this risk by avoiding bird predation in ponds (since fish-eating birds are the definitive hosts shedding *E. excisus* eggs). Other nematodes recorded in *S. glanis* include *Camallanus lacustris* (a common intestinal worm

with copepod intermediate host) and *Contracaecum* larvae (often encysted in viscera, from pelican or cormorant lifecycles). Each of these reflects the complex food web connections of catfish [79,82].

Acanthocephalan Parasites

Acanthocephalan parasites of European catfish include *Acanthocephalus lucii* and *Pomphorhynchus laevis*. *A. lucii* is a widely distributed acanthocephalan species infecting various freshwater fish across Europe, including the wels catfish [83]. Adult worms attach to the fish intestine using a hooked proboscis, embedding into the mucosa and absorbing nutrients through their tegument—often causing mechanical damage, hemorrhages, and reduced growth in heavy infestations [84]. Recent studies highlight the ecological significance of *A. lucii* as biosentinels for heavy metal pollution. These worms bioaccumulate trace metals (e.g., Cd, Pb, Tl) at concentrations many-fold higher than those in host tissues, making them useful indicators of contamination and possibly even mitigating metal toxicity to their fish hosts by sequestering metals in the intestinal lumen [85–87]. *P. laevis* is another acanthocephalan species infecting various freshwater fish, including the wels catfish [68]. This parasite attaches to the intestinal wall of its host using a retractable proboscis armed with hooks, causing mechanical damage and potentially leading to inflammation and reduced growth in the host [88]. The presence of *P. laevis* in freshwater fish populations can have ecological implications, particularly in terms of host health and the dynamics of parasite communities [89]. Understanding the distribution and impact of these parasites is crucial for managing fish health and biodiversity in freshwater ecosystems.

Crustacean Parasites

Apart from worms, *S. glanis* can suffer from crustacean parasites. Two noteworthy examples are the anchor worm *Lernaea cyprinacea* and the fish louse *Argulus foliaceus*. *Lernaea* is a copepod that burrows into the skin, typically on the fins or caudal area, with an erupting worm-like trunk and anchoring hooks. In catfish, heavy *Lernaea* infestations cause focal ulcers and blood loss. *Argulus* are disk-like branchiuran crustaceans that clamp onto the skin or gills and feed on blood. They can cause irritation, anemia, and transmit other pathogens. Both *Lernaea* and *Argulus* thrive in warm stagnant waters; they have been reported on wild and pond-held *S. glanis* [11,61]. Treatment involves manual removal or chemical bath treatments (e.g., organophosphates), and prevention is aided by keeping ponds free of invasive small fish, which can carry juvenile stages of these parasites.

The helminth and metazoan parasites of *S. glanis* are diverse. Table 8 summarizes the key species. From an ecological standpoint, the presence of these parasites indicates *S. glanis*' integration into aquatic food nets (serving as definitive or intermediate host). Some parasites (like *Diplostomum* or *Eustrongylides*) highlight connections between catfish and avian predators, while others (like *Camallanus* or *Lernaea*) tie catfish to invertebrate communities. In aquaculture, parasite management focuses on breaking life cycles (e.g., lining ponds to prevent snail or copepod populations, or fallowing to disrupt parasite transmission). With the trend toward recirculating systems, many of these parasites become less common, but any facility drawing surface water or stocking wild-caught juveniles must remain vigilant for parasite introduction.

Table 8. Key helminths and other metazoan parasites of *S. glanis*.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Thaparocleidus siluri</i> Zandt, 1924	Monogenean (gill fluke, family Dactylogyridae)	Gills	Epithelial hyperplasia, respiratory distress, and reduced growth	No	Formalin baths; praziquantel	[69,70]

Table 8. Cont.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Thaparocleidus vistulensis</i> Sivak, 1932	Monogenean (gill fluke, family Dactylogyridae)	Gills, skin	Hyperplasia, mucus hypersecretion, impaired respiration, secondary infections	No	Praziquantel; temperature management	[71,72]
<i>Diplostomum spathaceum</i> (Rudolphi, 1819) Olsson, 1876	Trematode (eye fluke, family Diplostomidae)	Eye lens (metacercariae)	Cataracts, impaired vision, and behavioral changes	No	Snail control; pond drying	[76]
<i>Orientocreadium siluri</i> Bychowsky and Dubinina, 1954	Trematode (intestinal fluke, family Orientocreadiidae)	Gastrointestinal tract (mid- and foregut)	Reduced growth, impaired digestion, and potential gut inflammation	No	Anthelmintics (praziquantel)	[68]
<i>Silurotaenia siluri</i> (Batsch, 1786) Nybelin, 1941	Cestode (tapeworm, family Triaenophoridae)	Intestine (midgut and hindgut)	intestinal obstruction (if heavily infested), poor growth, emaciation, reduced vitality	No	Niclosamide; praziquantel	[77]
<i>Proteocephalus osculatus</i> (Goeze, 1782) La Rue, 1911	Cestode (tapeworm, family Proteocephalidae)	Intestine (midgut and hind-gut)	Reduced growth, emaciation, and intestinal damage may predispose fish to secondary infections	No	Praziquantel	[75]
<i>Triaenophorus meridionalis</i> Kuperman, 1968	Cestode (tapeworm, family Triaenophoridae)	Intestine of predatory freshwater fish (definitive hosts), larvae in the liver and other organs of intermediate fish hosts	In intermediate hosts: liver damage, necrosis, reduced growth, increased mortality; in definitive hosts: intestinal obstruction if infestation is heavy	No	Avoid raw feed fish; freezing intermediates	[68]
<i>Caryophyllaeus laticeps</i> (Pallas, 1781) Lühe, 1910	Cestode (unsegmented tapeworm, family Caryophyllaeidae)	Intestine of predatory freshwater fish	Intestinal inflammation, obstruction in heavy infestations, emaciation, and reduced growth	No	Praziquantel	[76]
<i>Raphidascaris acus</i> (Bloch, 1779) Railliet and Henry, 1915	Nematode (roundworm, family Anisakidae)	Intestine, sometimes the stomach of predatory freshwater fish	Intestinal damage, inflammation, reduced growth, and emaciation in heavy infections	No	Anthelmintics; avoid infected prey fish	[11]
<i>Eustrongylides excisus</i> Jägerskiöld, 1909	Nematode (large red roundworm, family Dioctophymatidae)	Body cavity, stomach wall, liver, and muscles of fish	Visible red worms, severe tissue damage, hemorrhages, impaired condition, increased mortality	Yes	Removal of bird predators; proper evisceration	[66]
<i>Camallanus lacustris</i> (Zoega in Müller, 1776) Railliet and Henry, 1915	Nematode (intestinal roundworm, family Camallanidae)	Intestine (midgut and hindgut) of freshwater fish	Prolapse of reddish worms from the anus, intestinal inflammation, emaciation, and reduced growth	No	Levamisole; sanitation	[68]
<i>Contracaecum</i> larvae Railliet and Henry, 1912	Nematode (larval anisakid, family Anisakidae)	Body cavity, liver, mesenteries of fish	Usually asymptomatic in fish, but heavy infections may cause organ damage, reduced condition	Yes	Freezing fish prey; remove piscivorous birds	[11]
<i>Acanthocephalus lucii</i> (Müller, 1776) Lühe, 1911	Acanthocephalan (thorny-headed worm, family Echinorhynchidae)	Intestine of freshwater fish	Intestinal wall damage from proboscis, hemorrhages, inflammation, reduced growth, and possible perforations in heavy infections	No	Anthelmintics; reduce intermediate hosts	[83]
<i>Pomphorhynchus laevis</i> (Zoega in Müller, 1776) Porta, 1908	Acanthocephalan (thorny-headed worm, family Pomphorhynchidae)	Intestine (penetrates the intestinal wall with proboscis)	Intestinal damage, hemorrhages, perforations, weakened growth, increased susceptibility to secondary infections	No	Anthelmintics	[76]
<i>Lernaea cyprinacea</i> Linnaeus, 1758	Crustacean parasite ("anchor worm", Copepoda: Lernaeidae)	Skin, muscles, and sometimes gills of freshwater fish	Visible thread-like worms penetrating tissue, hemorrhages, ulcers, secondary bacterial/fungal infections, reduced growth	No	Organophosphate baths; manual removal	[57]
<i>Argulus foliaceus</i> Linnaeus, 1758	Crustacean parasite ("fish louse", Branchiura: Argulidae)	Skin, fins, gills	Visible flattened parasites on the body surface, irritation, scratching, anemia, ulceration, and predisposition to secondary infections	No	Salt baths; organophosphates	[11]

3.3.3. Oomycetes Infections

Oomycete diseases in European catfish are generally opportunistic, arising secondary to other stressors or lesions. Oomycetes can infect catfish, especially when water quality deteriorates or fish are immunocompromised.

Saprolegnia Genus

Saprolegniosis is caused by water molds of the genus *Saprolegnia* (oomycetes) and typically manifests as whitish, cotton-like patches on the skin or gills of fish. *S. parasitica*, *S. ferax*, and *S. australis* are reported in European catfish [90]. In *S. glanis*, *Saprolegnia* spp. often colonizes areas of prior damage—for instance, a net handling injury or a parasite lesion, and can expand into larger patches that compromise the epidermis. While *Saprolegnia* primarily consumes dead tissue, its growth on live fish can lead to osmotic stress, secondary bacterial invasion, and potentially mortality if extensive. Catfish eggs are also very susceptible to saprolegniasis, which can wipe out incubating eggs in the absence of antifungal treatments. The key to controlling saprolegniosis is prevention: maintaining good water quality and minimizing skin injuries. Once lesions appear, topical treatments (e.g., potassium permanganate or hydrogen peroxide baths) are used. Overall, *Saprolegnia* is a ubiquitous water mold, and in catfish, its presence is an indicator of underlying stress or trauma that should be addressed.

Branchiomyces Genus

Branchiomycosis, also known as gill rot, is a more severe and specific condition caused by *Branchiomyces* spp., which are filamentous oomycetes that invade gill blood vessels. *B. sanguinis* and *B. demigrans* are responsible for outbreaks of gill rot in warm-water fish [11]. The European catfish is among the species susceptible to branchiomycosis, particularly in eutrophic ponds during the summer [91]. In *S. glanis*, branchiomycosis can cause sudden high mortality [92]. The oomycete infects gill tissues, leading to thrombosis of branchial blood vessels and necrosis of gill filaments. Grossly, gills develop irregular patchy discoloration, with maroon to white necrotic areas interspersed with healthy tissue, giving a marbled appearance. Affected catfish may show signs of respiratory distress (gasping, reduced activity), and death can occur quickly due to loss of gill function. Outbreaks are often precipitated by high water temperatures (over 25 °C), organic-rich water, and crowding, which favor the proliferation of *Branchiomyces* spores in the environment [18]. Because branchiomycosis is acute and difficult to treat (few fungicides can be used in fish food at needed concentrations), emphasis is on prevention: avoiding excessive organic loading, reducing crowding in summer, and removing dead fish promptly (to reduce spore reservoirs). If an outbreak occurs, improving water flow and lowering temperature (if possible) can mitigate losses, and sometimes copper sulfate treatments are applied as a control measure with variable success. *Branchiomyces* outbreaks in catfish ponds have been recorded in Central and Eastern Europe, occasionally causing severe epizootics with 50–70% mortality in stocked fish (based on field reports) [92].

Oomycetes tend to be secondary invaders in *S. glanis*; all infections occur after some primary insult (such as parasitic or mechanical skin damage) [18]. Therefore, maintaining overall fish health and promptly treating primary diseases goes a long way in preventing oomycete issues. It is also important to note that some fungal-like infections of catfish might be underdiagnosed; for example, *Ichthyophonus* (a mesomycetozoea) could potentially infect catfish that consume infected prey, although it is more commonly reported in other predatory fish [93]. No significant *Ichthyophonus* outbreaks have been reported in *S. glanis* to date [92]. Lastly, while not a pathogen of catfish per se, molds can produce toxins in feed (mycotoxins) that indirectly affect catfish health; careful feed storage is thus part of

good health management. Table 9 summarizes the knowledge about saprolegniosis and branchiomycosis.

Table 9. Key oomycete pathogens of *S. glanis*.

Pathogen	Type	Affected Tissues	Clinical Signs	Zoonotic Potential	Treatment	References
<i>Saprolegnia</i> spp. Nees, 1823	Water molds (Oomycetes)	Skin, fins, gills, eggs, and sometimes musculature	Cotton-like patches on the skin or gills of fish, skin lesions, gill damage, fin erosion, high egg and fry mortality	No	Potassium permanganate baths; hydrogen peroxide dips; improved water quality and handling	[90]
<i>Branchiomyces</i> spp. Plehn, 1912	Water mold (Oomycete)	Gill tissues	Thrombosis of intravascular tissues of gill blood vessels and necrosis of gill filaments, necrotic gill patches, respiratory distress, surface gasping, lethargy, and high cumulative mortality	No	Improve flow and oxygenation; lower temperature if possible; copper sulfate in pond treatments (where permitted); removal of dead fish	[91,92]

4. Discussion

The taxonomic diversity and geographic spread of pathogens affecting *S. glanis* are shaped by multiple interacting factors. The wide taxonomic range and uneven geographical distribution of pathogens identified in *Silurus glanis* demonstrate the complexity of host–pathogen–environment interactions in freshwater ecosystems. Integrating these elements into a digitally designed conceptual model enables a comprehensive understanding of disease emergence and supports predictive capabilities relevant to both aquaculture and conservation.

Environmental determinants remain primary drivers of pathogen presence and diversity. Temperature influences nearly all pathogen groups, affecting viral replication, bacterial virulence, and the life cycles of protozoans and helminths. In warmer regions or seasons, infections by *Aeromonas* spp., *Flavobacterium columnare*, monogeneans, and oomycetes become more frequent, while thermal instability stimulates the proliferation of ciliates such as *Ichthyophthirius multifiliis*. Water chemistry, including dissolved oxygen, ammonia levels, and organic load, further modifies host susceptibility and pathogen persistence. These variables often act synergistically: eutrophication not only enhances microbial growth but also favors intermediate hosts that support complex parasite life cycles. Consequently, regional environmental gradients directly shape pathogen community structure.

Host-specific factors contribute an additional layer to this complexity. Age-dependent susceptibility is evident, with juveniles experiencing more severe viral and bacterial infections, while adults accumulate long-lived helminths. Physiological stress, driven by high fish density or poor water quality, suppresses immune function and facilitates co-infections, which frequently alter the clinical course of disease. Behavioral traits, such as the benthic feeding habits of *S. glanis*, increase exposure to intermediate hosts of acanthocephalans, nematodes, and trematodes. These host properties must therefore be integrated into any predictive model, aiming to capture realistic infection dynamics.

Wild ecosystems also exert significant influence. Parasites relying on multiple hosts, such as *Diplostomum* (snails and birds), *Eustrongylides* (oligochaetes and birds), and *Pomphorhynchus* (amphipods and fish), respond strongly to changes in biodiversity, water flow, and trophic interactions. Geographical differences in bird migration, mollusk distribution, or fish community composition translate into distinct regional disease profiles. As a result, pathogen patterns in *S. glanis* often reflect broader ecosystem health indicators, making them valuable for ecological monitoring.

Anthropogenic drivers amplify these natural processes. The intensification of aquaculture creates conditions conducive to pathogen transmission and accelerates the spread of infectious agents beyond their native ranges. Fish transport for stocking or trade frequently introduces new pathogens into previously unaffected regions. Insufficient biosecurity, com-

bined with antibiotic misuse, may further promote the establishment of resistant bacterial strains. These human-mediated pathways must be integrated into any digital model that seeks to assess or anticipate pathogen dispersal.

The digital conceptual model, proposed here, synthesizes four fundamental layers: environmental variables, host-related attributes, pathogen traits, and anthropogenic factors. By linking these components, such a model not only visualizes current pathogen distribution but also allows simulations under projected climate conditions, altered aquaculture practices, or changes in wildlife ecology. Predictive outputs may identify high-risk periods or regions, thus supporting targeted surveillance, optimized farm management, and strategic interventions aimed at minimizing disease impact.

Despite considerable progress, several disruptions remain. Molecular characterization of emerging viruses is still limited, and long-term ecological studies on helminth and protozoan communities are insufficient. The role of climate-induced habitat shifts in pathogen emergence warrants further investigation, particularly regarding temperature-sensitive species. Strengthening future research through harmonized diagnostic approaches, expanded geographical sampling, and robust ecological modeling will substantially improve the capacity to manage pathogen risks in *S. glanis* populations, as well as in the water ecosystems at all.

The integration of ecological, environmental, and anthropogenic factors into a digitally structured framework provides new opportunities for understanding pathogen dynamics. Environmental changes, especially temperature rise, eutrophication, and altered hydrological regimes, promote proliferation of opportunistic pathogens such as *Aeromonas*, *Saprolegnia*, monogeneans, etc. A conceptual digital model integrating climatic data, water chemistry, host movement, and pathogen biology can help predict pathogen emergence. Such an approach aligns with One Health principles and offers a solid basis for developing sustainable management strategies that support both aquaculture production and wild ecosystem integrity. A model could incorporate environmental clusters (temperature, dissolved oxygen, nutrient load, etc.); host factors (age, density, stress indicators, etc.); pathogen features (life cycle complexity, intermediate hosts, transmission mode, etc.); anthropogenic impacts (translocations, trade, antibiotic use, etc.). The proposed approach corresponds with the One Health framework and marks connections in fish health in wild and aquaculture aquatic ecosystems, and potential risks to humans.

5. Conclusions

S. glanis, the European catfish, hosts a wide array of pathogens reflecting its role as a top predator and a cultured species. This review has highlighted that major pathogen groups—viruses, bacteria, parasites, and oomycetes—all include members capable of infecting wels catfish (Tables 5–9). The biodiversity of *S. glanis* pathogens is noteworthy: from large DNA viruses like ranaviruses and herpesviruses, to microscopic ciliates and metazoan worms, the catfish can be considered an entire ecosystem of parasites. Ecologically, these pathogens illustrate the interconnectedness of the aquatic environment. For instance, the life cycles of catfish helminths involve organisms ranging from mollusks to birds, indicating that changes in ecosystem composition (such as the introduction of new species or loss of predators) can influence catfish disease patterns.

From an aquaculture perspective, understanding these host–pathogen relationships is critical for sustainable catfish production. Viral diseases can cause sudden high losses, so biosecurity and possibly selective breeding for resistance are priorities. Bacterial opportunists demand good water quality and stress reduction, along with prudent use of antibiotics to prevent resistance. Parasite management in catfish farming may involve integrated approaches such as periodic antiparasitic treatments, environmental modifications

(e.g., snail control to break trematode life cycles), and incorporation of cleaner species or low-density polyculture to reduce parasite burdens (taking a cue from traditional practices where catfish themselves helped control carp diseases). Oomycete issues, being secondary, underscore the importance of overall health management.

Importantly, more than 65% of the papers cited in this review are from the last five years (2021–2025), and 30% of all references are published in 2025, reflecting a growing research focus on catfish pathogens. This includes molecular characterization of newly discovered viruses and parasites, as well as investigations into zoonotic aspects of fish pathogens. The One Health concept is highly relevant to *S. glanis* diseases: pathogens such as *Aeromonas* and *Edwardsiella*, and certain parasites, such as *Eustrongylides* and *Contracaecum*, can affect multiple species, including humans. Thus, disease control in catfish aquaculture has benefits beyond just the farm—it helps protect wild fish communities and public health by limiting the spread of infectious agents and antimicrobial resistance genes.

The European catfish faces a broad but manageable spectrum of diseases. Continued research and surveillance are needed to detect emerging pathogens (e.g., novel viruses or drug-resistant bacteria) early. Effective control will rely on a combination of traditional methods (husbandry, hygiene, sound nutrition, etc.) and modern tools (vaccines, probiotics, genetic selection for disease resistance, etc.). Given the ecological versatility of *S. glanis*, a balanced approach that considers environmental health, fish health, and socio-economic factors will be most successful. Protecting European catfish from its pathogens is not only vital for aquaculture productivity but also for conserving this iconic species' role in aquatic ecosystems.

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Abbreviations

The following abbreviations are used in this manuscript:

<i>S. glanis</i>	<i>Silurus glanis</i>
ESV	European sheatfish virus
ECV	European catfish virus
EHNV	Epizootic hematopoietic necrosis virus
SgPV1	<i>Silurus glanis</i> papillomavirus 1
CfCV	<i>Silurus glanis</i> circovirus
<i>A. veronii</i>	<i>Aeromonas veronii</i>
<i>F. columnare</i>	<i>Flavobacterium columnare</i>
<i>S. siluri</i>	<i>Silurotaenia siluri</i>
<i>E. excisus</i>	<i>Eustrongylides excisus</i>
<i>P. laevis</i>	<i>Pomphorhynchus laevis</i>

<i>S. parasitica</i>	<i>Saprolegnia parasitica</i>
<i>S. ferax</i>	<i>Saprolegnia ferax</i>
<i>S. australis</i>	<i>Saprolegnia australis</i>
<i>B. sanguinis</i>	<i>Branchiomyces sanguinis</i>
<i>B. demigrans</i>	<i>Branchiomyces demigrans</i>
MAS	motile <i>Aeromonas</i> septicemia
ESC	enteric septicemia of catfish

References

- Lindell, N. Habitat Preference and Activity Pattern of European Catfish (*Silurus glanis*) at Its Northernmost Distribution Area. Ph.D. Thesis, Linköping University, Linköping, Sweden, 2021.
- Říha, M.; Rabaneda-Bueno, R.; Jarić, I.; Souza, A.T.; Vejřík, L.; Draštík, V.; Blabolil, P.; Holubová, M.; Jůza, T.; Gjelland, O.K.; et al. Seasonal habitat use of three predatory fishes in a freshwater ecosystem. *Hydrobiologia* **2022**, *849*, 3351–3371. [\[CrossRef\]](#)
- Encina, L.; Rodríguez-Ruiz, A.; Orduna, C.; Cid, J.R.; Granado-Lorencio, C. Impact of invasive catfish (*Silurus glanis*) on the fish community of Torrejón reservoir (Central Spain) during a 11-year monitoring study. *Biol. Invasions* **2024**, *26*, 745–756. [\[CrossRef\]](#)
- Sajdlová, Z.; Jůza, T.; Blabolil, P.; Draštík, V.; Čech, M. Pelagic distribution and night foraging of early juvenile European catfish (*Silurus glanis* L.). *J. Fish Biol.* **2025**, *106*, 1893–1898. [\[CrossRef\]](#) [\[PubMed\]](#)
- Adakebaike, Z.; Wang, Z.; Anasi, H.; He, J.; Zhai, X.; Shi, C.; Nie, Z. Study on the morphological development timeline and growth model of embryos and larvae of European catfish (*Silurus glanis*). *Animals* **2025**, *15*, 2478. [\[CrossRef\]](#)
- Hegedűs, B.; Bagi, Z.; Kusza, S. Navigating the genetic landscape: Investigating the opportunities and risks of cross-species SNP array application in catfish. *Genes* **2025**, *16*, 717. [\[CrossRef\]](#) [\[PubMed\]](#)
- Koprucu, K.; Yonar, S.M.; Koprucu, S.; Yonar, M.E. Effect of different stocking densities on growth, oxidative stress, antioxidant enzymes, and hematological and immunological values of European catfish (*Silurus glanis* L.; 1758). *Aquac. Int.* **2024**, *32*, 2957–2973. [\[CrossRef\]](#)
- Gisbert, E.; Luz, R.K.; Fernandez, I.; Pradhan, P.K.; Salhi, M.; Mozanzadeh, M.T.; Kumar, A.; Kotzamanis, Y.; Castro-Ruiz, D.; Bessonart, M.; et al. Development, nutrition, and rearing practices of relevant catfish species (Siluriformes) at early stages. *Rev. Aquac.* **2022**, *14*, 73–105. [\[CrossRef\]](#)
- Saleh, M.; Sellyei, B.; Kovács, G.; Székely, C. Viruses infecting the European catfish (*Silurus glanis*). *Viruses* **2021**, *13*, 1865. [\[CrossRef\]](#)
- Saleh, M.; Abdel-Baki, A.-A.; Dkhil, M.A.; El-Matbouli, M.; Al-Quraishy, S. Antiprotozoal effects of metal nanoparticles against *Ichthyophthirius multifiliis*. *Parasitology* **2017**, *144*, 1802–1810. [\[CrossRef\]](#)
- Roohi, D.J.; Sattari, M.; Asgharnia, M.; Rufchaei, R. Occurrence and intensity of parasites in European catfish, *Silurus glanis* L., 1758 from the Anzali Wetland, southwest of the Caspian Sea, Iran. *Croat. J. Fish.* **2014**, *72*, 25–31. [\[CrossRef\]](#)
- Mauad, J.R.C.; da Silva, M.C.; Araújo, C.M.C.; Silva, R.M.M.F.; Coleman, S.M.D.Q.; Russo, M.R. Zoonotic agents in farmed fish: A systematic review from the interdisciplinary perspective of the One Health concept. *Vet. Sci.* **2025**, *12*, 437. [\[CrossRef\]](#) [\[PubMed\]](#)
- Baer, J.; Hüsken, S.; Fromherz, M.; Geist, J.; Brinker, A. Drivers of the range expansion of the European catfish (*Silurus glanis*) within its native distribution. *J. Fish Biol.* **2025**, *107*, 1030–1044. [\[CrossRef\]](#)
- Rees, A. The Impact of Introduced European Catfish (*Silurus glanis* L.) in UK Waters: A Three Pond Study. Ph.D. Thesis, University of Hertfordshire, Hatfield, UK, 2020.
- Ziarati, M.; Zorriehzahra, M.J.; Hassantabar, F.; Mehrabi, Z.; Dhawan, M.; Sharun, K.; Shamsi, S. Zoonotic diseases of fish and their prevention and control. *Vet. Q.* **2022**, *42*, 95–118. [\[CrossRef\]](#)
- Bass, D.; Baron, S.; Travers, A.; Longshaw, M.; Haenen, O. One Health in fish and shellfish. *Bull. Eur. Assoc. Fish Pathol.* **2025**, *45*, 124752. [\[CrossRef\]](#)
- Page, M.J.; McKenzie, J.E.; Bossuyt, P.M.; Boutron, I.; Hoffmann, T.C.; Mulrow, C.D.; Shamseer, L.; Tetzlaff, J.M.; Akl, E.A.; Brennan, S.E.; et al. The PRISMA 2020 statement: An updated guideline for reporting systematic reviews. *BMJ* **2021**, *372*, n71. [\[CrossRef\]](#)
- Boon, J.H.; Huisman, E.A. Viral, bacterial and fungal diseases of Siluroidei, cultured for human consumption. *Aquat. Living Resour.* **1996**, *9*, S153–S164. [\[CrossRef\]](#)
- Costa, V.A.; Holmes, E.C. Diversity, evolution, and emergence of fish viruses. *J. Virol.* **2024**, *98*, e00118–24. [\[CrossRef\]](#)
- Tarján, Z.L.; Doszpoly, A.; Eszterbauer, E.; Benko, M. Partial genetic characterisation of a novel alloherpesvirus detected by PCR in a farmed Wels catfish (*Silurus glanis*). *Acta Vet. Hung.* **2022**, *70*, 321–327. [\[CrossRef\]](#) [\[PubMed\]](#)
- Abonyi, F.; Varga, A.; Sellyei, B.; Eszterbauer, E.; Doszpoly, A. Juvenile Wels catfish (*Silurus glanis*) display age-related mortality to European catfish virus (ECV) under experimental conditions. *Viruses* **2022**, *14*, 1832. [\[CrossRef\]](#)

22. Surján, A.; Fónagy, E.; Eszterbauer, E.; Harrach, B.; Doszpoly, A. Complete genome sequence of a novel fish papillomavirus detected in farmed Wels catfish (*Silurus glanis*). *Arch. Virol.* **2021**, *166*, 2603–2606. [[CrossRef](#)] [[PubMed](#)]
23. Doszpoly, A. Establishment and partial characterization of three novel permanent cell lines originating from European freshwater fish species. *Pathogens* **2025**, *14*, 531. [[CrossRef](#)]
24. Békési, L.; Kovacs-Gayer, E.; Ratz, F.; Turkovics, O. Skin infection of the sheatfish (*Silurus glanis* L.) caused by a herpes virus. In *Fish, Pathogens, and Environment in European Polyculture*; Akadémiai Kiadó: Budapest, Hungary, 1984; pp. 25–30.
25. Shi, H.; Zhou, T.; Wang, X.; Yang, Y.; Wu, C.; Liu, S.; Bao, L.; Li, N.; Yuan, Z.; Jin, Y.; et al. Genome-wide association analysis of intra-specific QTL associated with the resistance for enteric septicemia of catfish. *Mol. Genet. Genom.* **2018**, *293*, 1365–1378. [[CrossRef](#)]
26. Abass, N.Y.; Ye, Z.; Alsaqufi, A.; Dunham, R.A. Comparison of growth performance among channel–blue hybrid catfish, ccGH transgenic channel catfish, and channel catfish in a tank culture system. *Sci. Rep.* **2022**, *12*, 740. [[CrossRef](#)] [[PubMed](#)]
27. Ahne, W.; Schlotfeldt, H.J.; Thomsen, I. Fish viruses: Isolation of an icosahedral cytoplasmic deoxyribovirus from sheatfish (*Silurus glanis*). *J. Vet. Med. Ser. B* **1989**, *36*, 333–336. [[CrossRef](#)] [[PubMed](#)]
28. Martín, V.; Mavian, C.; Lopez Bueno, A.; de Molina, A.; Díaz, E.; Andrés, G.; Alejo, A. Establishment of a zebrafish infection model for the study of wild-type and recombinant European sheatfish virus. *J. Virol.* **2015**, *89*, 10702–10706. [[CrossRef](#)]
29. Leimbach, S.; Schütze, H.; Bergmann, S.M. Susceptibility of European sheatfish *Silurus glanis* to a panel of ranaviruses. *J. Appl. Ichthyol.* **2014**, *30*, 93–101. [[CrossRef](#)]
30. Siwicki, A.K.; Pozet, F.; Morand, M.; Volatier, C.; Terech-Majewska, E. Effects of iridovirus-like agent on the cell-mediated immunity in sheatfish (*Silurus glanis*)—An in vitro study. *Virus Res.* **1999**, *63*, 115–119. [[CrossRef](#)]
31. Mancheva, K.; Danova, S.; Vilhelmova-Ilieva, N.; Dobрева, L.; Kostova, K.; Simeonova, L.; Atanasov, G. Viral pathogens with economic impact in aquaculture. *Acta Microbiol. Bulg.* **2021**, *37*, 111–121.
32. Pozet, F.; Morand, M.; Moussa, A.; Torhy, C.; De Kinkelin, P. Isolation and preliminary characterization of a pathogenic icosahedral deoxyribovirus from the catfish *Ictalurus melas*. *Dis. Aquat. Org.* **1992**, *14*, 35–42. [[CrossRef](#)]
33. Bovo, G.; Comuzi, M.; Cescia, G.; Giorgetti, G.; Giacometti, P.; Cappellozza, E. Isolamento di un agente virale irido-like da pescegatto (*Ictalurus melas*) dall'allevamento. *Boll. Soc. Ital. Patol. Ittica* **1993**, *11*, 3–10.
34. Bigarré, L.; Cabon, J.; Baud, M.; Pozet, F.; Castric, J. Ranaviruses associated with high mortalities in catfish in France. *Bull. Eur. Assoc. Fish Pathol.* **2008**, *28*, 163–168.
35. Eaton, H.; Metcalf, J.; Penny, E.; Tcherepanov, V.; Upton, C.; Brunetti, C.R. Comparative genomic analysis of the family Iridoviridae: Re-annotating and defining the core set of iridovirus genes. *Virol. J.* **2007**, *4*, 11. [[CrossRef](#)] [[PubMed](#)]
36. Marsh, I.B.; Whittington, R.J.; O'Rourke, B.; Hyatt, A.D.; Chisholm, O. Rapid differentiation of Australian, European and American ranaviruses based on variation in major capsid protein gene sequence. *Mol. Cell. Probes* **2002**, *16*, 137–151. [[CrossRef](#)] [[PubMed](#)]
37. Choi, H.J.; Hur, J.W.; Cho, J.B.; Park, K.H.; Jung, H.J.; Kang, Y.J. Introduction of bacterial and viral pathogens from imported ornamental finfish in South Korea. *Fish. Aquat. Sci.* **2019**, *22*, 5. [[CrossRef](#)]
38. Adriaenssens, E.; Dutilh, B.; Harrach, B.; Junglen, S.; Kropinski, A.; Krupovic, M.; Kuhn, J.; Mushegian, A.; Postler, T.; Rubino, L.; et al. Modify the International Code of Virus Classification and Nomenclature (ICVCN) to prospectively mandate a uniform genus-species type virus species naming format. *Int. Comm. Taxon. Viruses. Approv. Propos.* **2018**, 2018.001G. [[CrossRef](#)]
39. Van Doorslaer, K. Revisiting papillomavirus taxonomy: A proposal for updating the current classification in line with evolutionary evidence. *Viruses* **2022**, *14*, 2308. [[CrossRef](#)]
40. Lőrincz, M.; Dán, Á.; Láng, M.; Csaba, G.; Tóth, G.Á.; Székely, C.; Cságola, A.; Tuboly, T. Novel circovirus in European catfish (*Silurus glanis*). *Arch. Virol.* **2012**, *157*, 1173–1176. [[CrossRef](#)]
41. Rusiñol, M.; Martínez-Puchol, S.; Ribeiro, D.; Verdager, J.; Torrejón, O.; Itarte, M.; Fernandez-Cassi, X. *Livestock Aggregated Samples for Monitoring Viruses Infecting Animals and Potentially Zoonotic Viral Pathogens*; SSRN Elsevier Inc.: New York, NY, USA, 2025; p. 5432750.
42. Varsani, A.; Abd-Alla, A.M.; Arnberg, N.; Bateman, K.S.; Benkő, M.; Bézier, A.; Biagini, P.; Bojko, J.; Butkovic, A.; Canuti, M.; et al. Summary of taxonomy changes ratified by the International Committee on Taxonomy of Viruses (ICTV) from the Animal DNA Viruses and Retroviruses Subcommittee. *J. Gen. Virol.* **2025**, *106*, 002113. [[CrossRef](#)]
43. Fijan, N.; Matasin, Z.; Jeney, Z.; Oláh, J.; Zwillenberg, L.O. Isolation of Rhabdovirus carpio from sheatfish (*Silurus glanis*). In *Fish, Pathogens, and Environment in European Polyculture*; Akadémiai Kiadó: Budapest, Hungary, 1981.
44. Békési, L.; Pálfi, V.; Csontos, L.; Kovács-Gayer, É.; Csaba, G.; Horváth, I. Epizootiology of the red disease of silure (*Silurus glanis* L.) and study of the isolated rhabdovirus. *Magy. Allatorv. Lapja* **1987**, *42*, 418–422.
45. Caruso, D.; Schlumberger, O.; Dahm, C.; Proteau, J.P. Plasma lysozyme levels in sheatfish *Silurus glanis* (L.) subjected to stress and experimental infection with *Edwardsiella tarda*. *Aquac. Res.* **2002**, *33*, 999–1008. [[CrossRef](#)]
46. Xiucui, H.; Xiaoxue, L.; Aijun, L.; Jingfeng, S.; Yajiao, S. Characterization and pathology of *Aeromonas veronii* biovar *sobria* from diseased sheatfish *Silurus glanis* in China. *Isr. J. Aquac.-Bamidgeh* **2019**, *71*.

47. Miryala, K.R.; Swain, B. Advances and challenges in *Aeromonas hydrophila* vaccine development: Immunological insights and future perspectives. *Vaccines* **2025**, *13*, 202. [\[CrossRef\]](#)
48. Tayyab, M.; Islam, W.; Waqas, W.; Zhang, Y. Probiotic–vaccine synergy in fish aquaculture: Exploring microbiome-immune interactions for enhanced vaccine efficacy. *Biology* **2025**, *14*, 629. [\[CrossRef\]](#)
49. Okasha, L.A.; Farag, E.A.; Sayed-ElAhl, R.M.; Sherif, A.H. Presence of virulent *Edwardsiella tarda* in farmed Nile tilapia and striped catfish. *BMC Microbiol.* **2025**, *25*, 572. [\[CrossRef\]](#) [\[PubMed\]](#)
50. Janda, J.M.; Duman, M. Expanding the spectrum of diseases and disease associations caused by *Edwardsiella tarda* and related species. *Microorganisms* **2024**, *12*, 1031. [\[CrossRef\]](#)
51. Ribeiro, V.P.; Bastos, J.K.; Harries, M.D.; Page, P.N.; Techen, N.; Meepagala, K.M. Constituents from Brazilian propolis against *Edwardsiella ictaluri* and *Flavobacterium covaie*, two bacteria affecting channel catfish (*Ictalurus punctatus*). *J. Fish Dis.* **2025**, *48*, e14030. [\[CrossRef\]](#)
52. Wang, Q.; Shen, L.; Guo, M.; Wang, B.; Liu, H.; Xiao, K.; Shi, Z.; Ji, W. Blood-brain barrier permeability and brain immunity in yellow catfish (*Pelteobagrus fulvidraco*) responding to *Edwardsiella ictaluri* infection. *Aquaculture* **2025**, *599*, 742179. [\[CrossRef\]](#)
53. Chen, R.; Shi, Z.; Han, Q.; Wang, Q.; Wang, B.; Guo, M.; Ji, W.; Shen, L. Expression profiles of NOD1 and NOD2 and pathological changes in gills during *Flavobacterium columnare* infection in yellow catfish, *Tachysurus fulvidraco*. *J. Fish Biol.* **2025**, *106*, 1766–1776. [\[CrossRef\]](#)
54. Wang, H.; Sheng, L.; Yazdi, Z.; Li, X.; Liu, Z.; Canakapalli, S.; Zhou, Y.; Liao, C.; Emami, S.; Kelly, A.M.; et al. The impact of florfenicol treatment on the microbial populations present in the gill, intestine, and skin of channel catfish (*Ictalurus punctatus*). *Anim. Microbiome* **2025**, *7*, 68. [\[CrossRef\]](#) [\[PubMed\]](#)
55. Kumar, S.; Paul, T.; Sarkar, P.; Kumar, K. *Management of Fish Diseases*; Springer Nature Singapore: Singapore, 2025.
56. Yang, H.; Tu, X.; Xiao, J.; Hu, J.; Gu, Z. Investigations on white spot disease reveal high genetic diversity of the fish parasite, *Ichthyophthirius multifiliis* (Fouquet, 1876) in China. *Aquaculture* **2023**, *562*, 738804. [\[CrossRef\]](#)
57. Ahmet, Ö.Z.E.R. Wild and cultured fish parasites in Türkiye: An updated list of species, hosts, microhabitat and zoogeographical distribution since 2020. *Bull. Univ. Agric. Sci. Vet. Med. Cluj-Napoca Vet. Med.* **2025**, *82*, 12–68.
58. Marana, M.H.; Al-Jubury, A.; Mathiessen, H.; Buchmann, K. Lipopeptide surfactant killing of *Ichthyophthirius multifiliis*: Mode of action. *Aquac. Rep.* **2023**, *30*, 101562. [\[CrossRef\]](#)
59. Özil, Ö. Antiparasitic activity of medicinal plants against protozoan fish parasite *Ichthyophthirius multifiliis*. *Isr. J. Aquac.-Bamidgeh* **2023**, *75*, 1–9. [\[CrossRef\]](#)
60. Sajiri, W.M.H.W.; Székely, C.; Czeglédi, I.; Buchmann, K.; Sellyei, B. Comparative in vitro effects of novel and conventional parasitocides on *Thaparocleidus vistulensis* (Siwak, 1932) (Monopisthocotyla) parasitizing European catfish (*Silurus glanis*). *Aquac. Rep.* **2025**, *43*, 102952. [\[CrossRef\]](#)
61. Gul, Y.R. Assessment of the Morphological Investigations of Some Freshwater Fish Parasites in Greater Zab River, North of Iraq. Master's Thesis, Graduate School of Natural and Applied Sciences, Harran University, Şanlıurfa, Türkiye, 2020.
62. Arapi, E.A.; Cable, J. Survey-based insights into treatments of infectious diseases in freshwater ornamental fish in the UK. *Acad. Biol.* **2025**, *3*, 3. [\[CrossRef\]](#)
63. Farmer, B.D.; Straus, D.L.; Beck, B.H. *Spironucleus* and *Hexamita* spp. In *Clinical Guide to Fish Medicine*; John Wiley & Sons, Inc.: Hoboken, NJ, USA, 2021; Volume 20, p. 506.
64. Rogers, W. 3.2.1 Ichthyobodiasis. In *Fish Health Section—Blue Book*; American Fisheries Society: Bethesda, MD, USA, 1994.
65. Mhaisen, F.T. Checklists of blood parasites of fishes of Iraq. *Aalb. Acad. J. Pure Sci.* **2020**, *1*, 1–12.
66. Barzegar, M.; Raissy, M.; Shamsi, S. Protozoan parasites of Iranian freshwater fishes: Review, composition, classification, and modeling distribution. *Pathogens* **2023**, *12*, 651. [\[CrossRef\]](#)
67. Kosymbetovich, S.E. Parasitofauna of productive fish: Lower area of the Amudarya River. *Am. J. Appl. Sci. Technol.* **2025**, *5*, 26–27. [\[CrossRef\]](#)
68. Atanasov, G. Fauna, Morphology and Biology on the Endohelminths of Fish from Bulgarian Part of the Danube River. Ph.D. Thesis, IEMPAM-BAS, Sofia, Bulgaria, 2012.
69. Mancheva, K.; Karaivanova, E.; Atanasov, G.; Stojanovski, S.; Nedeva, I. Analysis of the influence of the host body size on morphometrical characteristics of *Ancylo-discoides siluri* and *Ancylo-discoides vistulensis*. *Biotechnol. Biotechnol. Equip.* **2009**, *23*, 735–741. [\[CrossRef\]](#)
70. Gallina, E.; Strona, G.; Galli, P. *Thaparocleidus siluri*, monogenoidean parasite of *Silurus glanis*: A new record from Italy. *Parassitologia* **2009**, *51*, 69–70.
71. Sajiri, W.M.H.W.; Székely, C.; Molnár, K.; Buchmann, K.; Sellyei, B. Pathological effects of *Thaparocleidus vistulensis* (Siwak, 1932) infection on the gills of *Silurus glanis* Linnaeus, 1758. *Acta Vet. Hung.* **2025**, *73*, 56–63. [\[CrossRef\]](#) [\[PubMed\]](#)
72. Sajiri, W.M.H.W.; Székely, C.; Sellyei, B. Influences of light–dark cycle and water temperature on in vitro egg laying, hatching, and survival rate of *Thaparocleidus vistulensis* (Dactylogyridea: Ancylo-discoididae). *Parasitol. Res.* **2025**, *124*, 4. [\[CrossRef\]](#)

73. Bognár, A.; Borkhanuddin, M.H.; Nagase, S.; Sellyei, B. Biopsy-based normalizations of gill monogenean-infected European catfish (*Silurus glanis* L., 1758) stocks for laboratory-based experiments. *PeerJ* **2024**, *12*, e18288. [\[CrossRef\]](#) [\[PubMed\]](#)
74. Sajiri, W.M.H.W.; Székely, C.; Molnár, K.; Buchmann, K.; Sellyei, B. Reproductive strategies of the parasitic flatworm *Thaparcleidus vistulensis* (Siwak, 1932) (Platyhelminthes, Monogenea) infecting the European catfish *Silurus glanis* Linnaeus, 1758. *Int. J. Parasitol. Parasites Wildl.* **2023**, *22*, 113–120. [\[CrossRef\]](#)
75. Normann, V.W. Drivers of Fish Parasite Infracomunities in a High Latitude System—A Dive into Host and Parasite Traits Shaping Parasite Diversity. Master's Thesis, UiT—The Arctic University of Norway, Tromsø, Norway, 2025.
76. Yakhchali, M.; Tehrani, A.A.; Ghoreishi, M. The occurrence of helminth parasites in the gastrointestinal of catfish (*Silurus glanis* Linnaeus 1758) from the Zarrine-roud river, Iran. *Vet. Res. Forum* **2012**, *3*, 143.
77. Frolova, T.V.; Izvekov, E.I.; Izvekova, G.I. First insights into the activity of major digestive enzymes in the intestine of the European catfish *Silurus glanis* and protective anti-enzymatic potential of its gut parasite *Silurotaenia siluri*. *J. Fish Biol.* **2023**, *103*, 985–993. [\[CrossRef\]](#)
78. Molnár, K.; Székely, C. Helminth parasites of fishes from the Hungarian section of River Danube. *Halászat-Tudomány (Fish. Sci.)* **2024**, *10*, 3–20. [\[CrossRef\]](#)
79. Ibrahimova, N.E. Systematic review of catfish (*Silurus glanis* L., 1758) parasites within their habitat. *Nematoda. Adv. Biol. Earth Sci.* **2020**, *5*, 76–83.
80. Kurbanova, A. Fish parasitofauna of the Amudarya river delta lakes. *BIO Web Conf.* **2024**, *141*, 03010. [\[CrossRef\]](#)
81. Kudlai, O.; Rakauskas, V.; Baker, N.J.; Pantoja, C.; Lisitsyna, O.; Binkienė, R. Helminth parasites of invasive freshwater fish in Lithuania. *Animals* **2024**, *14*, 3293. [\[CrossRef\]](#) [\[PubMed\]](#)
82. Mhaisen, F.T.; Abdul-Ameer, K.N. Checklist of fish hosts of species of *Contracaecum* Railliet & Henry, 1912 (Nematoda: Ascaridida: Anisakidae) in Iraq. *Biol. Appl. Environ. Res.* **2021**, *5*, 281–306. [\[CrossRef\]](#)
83. Kirin, D.; Chunchukova, M. Helminths and helminth communities of *Silurus glanis* (Linnaeus, 1758) from the Tundja river, Bulgaria. *Sci. Pap. Ser. D Anim. Sci.* **2021**, *64*, 42–86.
84. Buchmann, K.; Karami, A.M. Fish acanthocephalans as potential human pathogens. *Curr. Clin. Microbiol. Rep.* **2024**, *11*, 99–106. [\[CrossRef\]](#)
85. Soose, L.J.; Krauss, M.; Landripet, M.; Laier, M.; Brack, W.; Hollert, H.; Klimpel, S.; Oehlmann, J.; Jourdan, J. Acanthocephalans as pollutant sinks? Higher pollutant accumulation in parasites may relieve their crustacean host. *Sci. Total Environ.* **2025**, *958*, 177998. [\[CrossRef\]](#)
86. Mijošek, T.; Šariri, S.; Kljaković-Gašpić, Z.; Fiket, Ž.; Marijić, V.F. Interrelation between environmental conditions, acanthocephalan infection and metal(loid) accumulation in fish intestine: An in-depth study. *Environ. Pollut.* **2024**, *356*, 124358. [\[CrossRef\]](#) [\[PubMed\]](#)
87. Hassanine, R.; Al-Hasawi, Z. Acanthocephalan worms mitigate the harmful impacts of heavy metal pollution on their fish hosts. *Fishes* **2021**, *6*, 49. [\[CrossRef\]](#)
88. Stroe, M.D.; Guriencu, R.C.; Athanosoupolos, L.; Ion, G.; Coman, E.; Mocanu, E.E. Health profile of some freshwater fishes collected from Danube River sector (km 169–197) in relation to water quality indicators. *Sci. Pap. Ser. D Anim. Sci.* **2022**, *65*, 654–663.
89. Esposito, A.; Foata, J.; Quilichini, Y. Parasitic helminths and freshwater fish introduction in Europe: A systematic review of dynamic interactions. *Fishes* **2023**, *8*, 450. [\[CrossRef\]](#)
90. Eszterbauer, E.; Erdei, N.; Hardy, T.; Kovács, A.; Verebélyi, V.; Hoitsy, G.; Katics, M.; Bernáth, G.; Lang, Z.; Kaján, G.L. Distribution and environmental preferences of *Saprolegnia* and *Leptolegnia* (Oomycota) species in fish hatcheries in Hungary. *Aquaculture* **2025**, *596*, 741737. [\[CrossRef\]](#)
91. Sarkar, P.; Raju, V.S.; Kuppusamy, G.; Rahman, M.A.; Elumalai, P.; Harikrishnan, R.; Arshad, A.; Arockiaraj, J. Pathogenic fungi affecting fishes through their virulence molecules. *Aquaculture* **2022**, *548*, 737553. [\[CrossRef\]](#)
92. FAO. *Summary Report: Regional Workshop on Fish Health Management*; FAO: Ankara, Türkiye, 2023.
93. Sahu, A.K.; Kumar, V.; Udgate, S.K.; Das, B.K. Diagnostic techniques for fish fungal diseases. In *Laboratory Techniques for Fish Disease Diagnosis*; Springer Nature Singapore: Singapore, 2025; pp. 227–258.

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