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A Review of the Epidemiological, Clinical, and Pathological Aspects of Malignant Catarrhal Fever

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ABSTRACT: **Malignant catarrhal fever is a severe viral disease that affects a variety of domestic and wild ruminants. It is caused by group of Gammaherpes viruses. Each virus is well-adapted to its natural host, and is normally carried asymptomatically in reservoir hosts, but it can cause severe disease in other species. There is no successful treatment for MCF, and the case fatality rate is extremely high. Outbreaks are common in some areas, where cattle are seasonally exposed to the wildebeest associated virus during peak replication periods. Currently, the only effective control measures are to isolate susceptible species from carriers or to breed virus-free reservoir hosts.**

Keywords: MCF, Gamma Herpesviruses, Alcelaphine herpesvirus-1, Ovine herpesvirus-2.

INTRODUCTION

Malignant catarrhal fever is a highly pathogenic and lethal viral disease that mainly affects ruminant species and is caused by a member of the *Gammaherpesvirinae* sub-family of *Rhadinoviruses* (Li *et al*., 2005). In nature, these viruses can be found as in apparent infections in well-adapted ruminants that serve as reservoir hosts (Sood *et al*., 2013). MCF is increasingly being recognised as a source of significant economic losses in many large ruminant species, such as cattle, bison, and deer, as well as a threat to endangered species kept in mixed-species confinement. MCF mostly affects lymphoid organ, elementary, and respiratory tract. After the onset of clinical signs, death may occur within a few days or several weeks (Russell *et al*., 2009). The causative viruses can be existing in nature as subclinical infections in other species that function as carriers and are well-adapted to them. Two major epidemiologic variants of MCF are distinguished by the reservoir ruminant species from which the pathogenic virus emerges. One, known as the African form, is referred to as wildebeest-associated MCF (WA-MCF). The other is referred to as sheep-associated MCF (SA-MCF). In veterinary medicine, MCF has a long history. Maasai pastoralists and South African farmers recognised the connection between wildebeest and MCF in domestic cattle early on, referring to the disease as snotziekte (snotting sickness) (Li *et al*., 2014). MCF has been reported worldwide (Dabakand Bulut, 2003). Clinical events are more intermittent in other parts of the world, and they may appear suddenly in animals who had previously contacted reservoir hosts without ill effects. MCF can be diagnosed using a combination of history, symptoms, histopathology, and identification of viral antibodies or viral DNA in the blood or tissues. No practical, consistently effective treatments and vaccines are available. At present, the only effective control measures are to isolate susceptible species from carriers and maintain hygiene (Ehlers *et al*., 1999).

Etiological agent. Malignant catarrhal fever is caused by viruses belonging to the Macavirus genus of the *Herpesviridae* family (subfamily *Gammaherpesvirinae*) (Davison *et al*., 2009). There are at least 10 members of the MCFVs family, six of which are pathogenic in natural conditions (Li *et al*., 2005). MCF viruses are usually named after their reservoir hosts. The pathogenic viruses include the Alcelaphinae/Hippotraginae group of MCF viruses includes *alcelaphine herpesvirus 1* (AlHV-1), *alcelaphine herpesvirus 2* (AlHV-2), hippotragine

herpesvirus 1 (HiHV-1), and *MCFV-oryx* (Li *et al*., 2014). The Caprinae group includes *ovine herpesvirus 2* (OvHV-2), *caprine herpesvirus 2* (CpHV-2), MCF virus-white tailed deer (MCFV-WTD), MCFV-ibex, MCFV-muskox, and MCFV-aoudad. MCFV-WTD is an outlier in that the virus was named for the infected species rather than the reservoir host, which was unknown at the time. The two most important viruses are OvHV-2, which causes MCF in sheep (SA-MCF), and AlHV-1, which causes MCF in wildebeests (WA-MCF). CpHV-2, MCFV-WTD, MCFV-ibex, and AlHV-2 are among the viruses that have been identified as pathogenic. The AlHV-1 has been isolated and sequenced completely, but OvHV-2 has never been isolated due to the lack of a productive tissue-culture system. However, OvHV-2- infected bovine T-lymphoblastoid cell line BJ1035 that was derived from a clinically affected cow with SA-MCF, has been used for molecular characterization of the virus.

Geographic Distribution. The MCF has a worldwide distribution and can be found in both temperate and tropical climates. The disease has been documented in North and South America, Canada, Australia, New Zealand, Southeast Africa, Brazil, Italy, Europe, Peru, Russia, Asia and among other places (Amoroso *et al*., 2017).WA-MCF is primarily found in Sub-Saharan African countries such as South Africa, Kenya and Tanzania, where wildebeests are important wildlife species (Lankester *et al*., 2015). It causes the loss of approximately 10% of cattle herds each year in Kenya. Cases are becoming more common in South Africa as the game ranching, wildlife, and tourism sectors increase (Orono *et al*., 2019). OvHV-2 is enzootic worldwide in domestic sheep (Albini *et al*., 2003). For the first time in India, Parihar *et al*. (1975) from the Panjab state reported SA-MCF in cattle and buffaloes based on histopathological examination. The disease's epzootiological characteristics in buffalo calves were also studied by Singh *et al*. (1979). Later, cattle were shown to have MCF linked with sheep (Wani *et al*., 2004). OvHV-2 has also been found in sheep and goats in Jammu and Kashmir, as well as in southern India, and has been linked to respiratory illness or fever (Banumathi *et al*., 2008). SA-MCF cases were also found in captive bison in the Karnataka (India) (Sood *et al*., 2012). Kumar *et al*., (2014) detected OvHV-2 in crossbred cattle in Andhra Pradesh (India) during an SA-MCF outbreak. In India, the first molecular evidence and genetic characterization of Ovine Herpesvirus 2 in a multiple animal species was recently reported (Kumar *et al*., 2021). In various Indian states, the incidence of OvHV-2 in sheep ranged from 24.44 % to 85% (Premkrishnan *et al*., 2015). WA-MCF is a constant problem with pastoralists in Eastern and Southern Africa, where wildebeest are commonly found (Bedelian *et al*., 2007). In zoological collections containing wildebeest, WA-MCF has also been a concern (Whitaker *et al*., 2007). Although SA-MCF was first detected in Europe, it has now spread to other regions of the globe where sheep and cattle (or other MCF-susceptible species) are kept together. The first case of MCF was also reported in captive pudu (*Pudupudu*) in Italy (Modesto *et al*., 2015). SA-MCF is currently a major economic and welfare issue in bison in the United States and Bali cattle (*Bos javanicus*) in Indonesia (Berezowski *et al*., 2005). Outside of Africa, the European form of OvHV-2 (SA-MCF), is often found in wildlife domestic animals and captive ruminants. After the first report from Punjab in 1975, SA-MCF was found in Indian cattle and sheep, and it is now listed as an emerging disease in India (Sood *et al*., 2013).

Host range and susceptibility. MCF can affect a variety of natural hosts in the Artiodactyl families including *Bovidae*, *Cervidae*, *Giraffidae*, and *Suidae* (Russell *et al*., 2009). The subfamilies *Alcelaphinae*, *Caprinae*, and *Hippotraginae*, which include wildebeest, horses, goats, and roan antelope, contain the majority of well-adapted reservoir hosts (Li *et al*., 2001). They shed viruses into the atmosphere and can spread them to clinically susceptible hosts when they come into contact with them directly or indirectly. In general, poorly adapted hosts are often referred to as "dead end hosts" because they do not shed infectious virus. More than 30 ungulate species have been identified as carriers of the disease, including Indian gaur (*Bos gaurus*), domesticated cattle (*Bos indicus*), swamp or water buffalo (*Bubalus bubalis*), Balinese cattle (*Bos javanicus*), American bison (*Bisonbison*), nilgai or blue cattle (*Boselaphus tragocamelus*), red deer (*Cervus elaphus*) (Klieforth *et al*., 2002), sika deer (*Cervus nippon*) (Keel *et al*., 2003), Kudu (*Tragelaphus imberbi; Tragelaphus strepsiceros*), mule deer (*Odocoileus hemionus*), white-tailed deer (*Odocoileus virginianus*) (Kleiboeker *et al*., 2002), axis deer (*Axisaxis*), red brocket deer (*Mazama americana*), Chinese water deer (*Hydropotes inermis*), Père D avid's deer (*Elaphurus davidiansus*), rusa deer (*Cervus timorensis*), elk (*Cervus canadensis*), moose (*Alces alces*), and pigs (*Sus scrofa*) (Alcaraz *et al*., 2009). Rabbits and hamsters are among the laboratory animals that are vulnerable to experimental infection and disease (Gailbreath *et al*., 2008). The susceptibility of different ruminant species to OvHV-2 infection and MCF differ significantly, according to experimental studies in bis on, cattle, and sheep. American bison are approximately 100 times more susceptible to infection and 10,000 times more susceptible to developing disease than are cattle (Taus *et al*., 2006). Bison and domestic sheep are more than six orders of magnitude different in terms of MCF sensitivity (O'toole *et al*., 2007). MCF is normally fatal once clinical signs appear and subclinical infection may occur in bison, cattle, and certain deer species.

Economic impact. MCF has a wide range of economic effects. The losses have never been systematically estimated, because the condition lacks an organised, enforced reporting system, and MCF is significantly under-reported, particularly in mild or unusual instances. It is a sporadic disease, low morbidity occurs in cattle, especially European breeds, with solitary incidences occurring at random intervals. MCF

outbreaks can occur, on rare occasions, reach epidemic proportions and killing a large number of animals in a span of weeks or months (O'Toole *et al*., 2002). It is responsible for significant annual losses to African domestic cow herds, estimated to be over 7% in 1970 (Collery and Foley, 1996). MCF is often lethal in operations involving more susceptible species like bison, banteng, and other antelope species. It is becoming a serious concern for bison breeding and feeding operations in the United States, when MCF-related feedlot losses can reach 10% of animals (Schultheiss *et al*., 2000). MCF has pushed small-scale bison raisers out of business as a result of infected sheep being moved onto neighbouring farms. MCF losses range from sporadic to near-epidemic levels in operations that allow susceptible species to encounter sheep and, to a lesser extent, domestic goats. MCF is responsible for around 40% of the annual death losses in farmed deer in New Zealand (Beatson, 1985). Because of the widespread under-diagnosis of MCF, the true incidence is likely to be substantially higher than widely assumed. In 2003, outbreak in an Idaho bison feedlot resulted in the deaths of over 800 bison, resulting in losses of over a million dollars (Li *et al*., 2006). Clinical MCF diagnostic testing has generally been imprecise, resulting in many misdiagnoses, especially in mild or uncommon diseases. True prevalence is higher than usually thought due to widespread underdiagnosis (O'Toole *et al*., 1997).

Pathogenesis. MCF virus is strongly associated with the cell (Akula *et al*., 2001). The pathogenesis of MCF is thought to be caused by virus-induced T-lymphocyte proliferation and malfunction. Lymphoblastoid cell lines (LCLs) have been derived from SA-MCF-infected animals (cattle and deer), and some of them transmit MCF to susceptible species (Baxter *et al.,* 1993). A typical gammaherpesvirus enters host cells by attaching one or more viral glycoproteins to cellular receptors, followed by the fusion of the viral envelope with the cell membrane of host (Cunha *et al*., 2016). Glycoprotein B (gB) is important in entry of gammaherpesviruses into host cells via heparan sulphate and/or integrin 31 (Akula *et al*., 2002). A group of OvHV-2 glycoproteins, Glycoprotein B (gB), Glycoprotein L (gL), and Glycoprotein H (gH), together known as the core fusion machinery, are necessary for membrane fusion (Aihajri *et al*., 2017). MCF is characterised by significant pathological alterations in the organs of infected animals with little evidence of viral antigen, while viral DNA can be detected using PCR. MCF causes progressive T cell hyperplasia in infected animals, which includes local proliferation and invasion of lymphoid and non-lymphoid organs, as well as severe vasculitis and tissue damage, and epithelial necrosis caused by dysregulated cytotoxic lymphocytes (Saura *et al*., 2018). OvHV-2 associated lesions are more common in visceral lymphoid tissue (e.g., mesenteric lymph nodes), whereas AlHV-1 lesions are more common in peripheral lymph nodes (Swa *et al*., 2001). Furthermore, compared to AlHV-1, OvHV-2-associated lesions contain more areas of necrosis. However, most studies have shown that lymphoid cell infiltrations with both viruses are primarily T cells, with CD8+T cells are the most predominant associated with vascular lesions and lymphoid hyperplasia in tissues, with few CD4+ T cells (Simon *et al*., 2003). Most gamma viruses develop latency in lymphoid tissues in early infection, causing sub-clinical infection in a wide range of ruminant species.

Epidemiology. For AlHV-1 and OvHV-2, the epidemiology of MCF has been relatively well described in terms of virus transmission patterns from reservoir animals to clinically vulnerable hosts. Both viruses are spread into the environment by their reservoirs through nasal and possibly ocular secretions (Li *et al*., 2004). In general, effective transmission via infected secretions of a reservoir host to a clinically susceptible host is favoured by intimate contact and a cold, wet environment (Jacobsen *et al*., 2007). MCFV cannot be passed from one clinically vulnerable host to another through natural means; infected animals are therefore regarded as dead-end hosts (Zakharova *et al*., 2020). Almost all reservoir hosts are infected with their own strain of MCFV; but, under certain circumstances, a dual infection can occur. However, MCF-like disease has been described in goats and sheep but the infection in reservoir hosts is usually mild (Klieforth *et al*., 2002). MCFVs, like other herpesviruses, are relatively unstable in the environment; for example, AlHV-1 can lose 99.9% of its infectivity in about 3 hours in dry and hot weather (Mushi *et al*., 1981**)**. MCFV is present in clinically vulnerable hosts in the vicinity of inadvertent wildlife carriers worldwide.

Transmission. There could be various routes of transmission of MCFVs like inhalation, injection, direct contact, and in-utero. MCF occurs in all seasons. Nasal and ocular secretions are the primary sources of free virus in wildebeest. The occurrence and severity of disease are also influenced by the immune status and age of host. Transmission usually occurs when susceptible host contact with reservoir host. Both AlHV-1 and OvHV-2 appear to be spread via contact or aerosol, mostly among wildebeest calves (AlHV-1) and lambs (OvHV-2) under one year of age (Russell *et al*., 2009). Both horizontal and vertical transmissions are important of the transmission of disease. The virus is maintained in similar but not identical patterns in sheep and wildebeest population. The transmission pattern of AlHV-1 in wildebeests is differed significantly from that of OvHV-2 in sheep (Li *et al*., 1998). AlHV-1 transmission is very effective within free-living wildebeest populations. AlHV-1 can be seen in both cell-free and cell-associated forms in wildebeest. Cell-free AlHV-1 is contagious, whereas the cell-associated virus is only occasionally transmitted to other species. Wildebeests shed cell-free virus in nasal and ocular secretions for a brief period of time after becoming infected. All wildebeest calves become infected with MCFV within their first few months of life, either through in utero infection and direct contact, or aerosol transmission. Wildebeest calves can transmit the virus

in nasal and ocular secretions, often in the cell-free form. Although close contact is typically needed. Transmissions of the virus over one hundred metres have been recorded (Mushi *et al*., 1981). Contamination of pastures, as well as fomites, can also play a role in transmission. Transmission by wildebeest calves occurs most often between the ages of 1-2 months. Virus shedding usually peaks in aged 3 to 4 months. Transmission after the age of 6 months, AlHV-1 is mostly cell-associated, and these wildebeest shed very little virus or uncommon due to the production of neutralising antibodies. OvHV-2 is primarily transmitted by the respiratory tract, most likely by aerosols. In instance, lambs between the ages of 6 and 9 months shed a virus in their nasal and ocular secretions on an irregular basis. When lambs are 3–6 months old, they become infected through aerosol transfer from other members of the flock, and they begin actively shedding the virus at about 6–9 months. Shedding decreases at about 10 months, with adults shed at a far slower rate than adolescents. A percentage of lambs are infected in utero, with the majority being infected perinatally. However, the infection does not occur until after 3 months of age in some cases, possibly because of maternal antibody. While susceptible species must be in close proximity to sheep, transmission from sheep to cattle has been shown at distances of up to 5.1 km in bison and at least 70 metres in cattle from a lamb feedlot (Li *et al*., 2008). The presence of viral nucleic acids in the sperm of rams has also been observed, though the relevance of this study is still unknown (Taus *et al*., 2015). CpHV-2 appears to be transmitted similarly to OvHV-2 in goats. Most other MCF viruses have little or no information about how they spread in their reservoirs. MCFVs-susceptible species are usually dead-end hosts, and they do not transmit the virus to other animals; this has the beneficial effect of limiting or controlling disease spread, particularly during outbreaks. This is because of the virus remains in a cell-associated manner in these species, and a cell-free virus is not produced.

Clinical signs. MCF in cattle has been identified as having several overlapping but distinct clinical disease patterns, including mild, per-acute, alimentary, cutaneous, neurological, and head & eye (OIE, 2004). Typical symptoms include fever, ocular and nasal discharge, inappetence, diarrhoea, lesions of the buccal cavity, and depression (Zemljic *et al*., 2012). The clinical signs vary depending on the species infected, the virus, the degree of exposure to the disease, and the length of time the animal lives after the clinical signs appear. Per-acute MCF is most common in cervids, and is preceded by symptoms such as depression, weakness, diarrhoea, and dysentery occurring 12-24 hours before death. Depression and fever are the most common clinical symptoms in the first 1 to 3 days of sickness, accompanied by oculo-nasal discharge (Brown *et al*., 2007). Cattle, on the other hand, can survive for a week or more. The head and eye form are the commonest type of expression in cattle characterised by opacity of the cornea, agalactia, high fever, rapid pulse rate, anorexia, blepharospasm, lymphadenopathy and congestion of scleral vessels (Yus *et al*., 1999). Bilateral mucoid nasal discharge develops in the animals, which can become mucopurulent, profuse, and haemorrhagic in some cases. Affected animals can also be dyspnoeic due to nasal cavity obstruction and open-mouthed breathing. Severe bilateral keratitis, characterised by corneal opacity, excessive tearing, and hypopyon, is common, resulting in photophobia and partial blindness. Nervous signs such as in co-ordination, tremor, and a demented appearance might occur early stage, whereas head pressing paralysis and nystagmus are common in the latter stages. In nervous forms, nervous signs are evident, like hyperaesthesia, incoordination, nystagmus, weakness in one limb, and muscle tremor. The alimentary form has many of the same effects as the head and eye form, except for mild ocular changes and severe diarrhoea. In cutaneous form, skin ulceration and necrosis restricted to the udder and teats. The mild form is often seen in experimental animals. Chronic cases have been recorded in cattle and bison, whereas MCF recovery has been reported in livestock that remained infected for a long time. The clinical course is shorter in MCF-susceptible animals, such as American bison and many cervid species than in cattle, and sudden death is possible. In bison and deer, nasal discharge, muzzle crusting, and corneal opacity are less noticeable than cattle (Palmer *et al*., 2013). MCF appears similar in other species in general, but there are some differences. Corneal opacity in water buffalo has been reported to be inconsistent, whereas mild to moderate conjunctivitis appears to be normal. While some animals have typical nasal, ocular, gastrointestinal, and/or neurological symptoms, typically, American bison (Bison bison) die quickly without showing purulent rhinitis or corneal opacity. In comparison to cattle, haematuria and haemorrhagic enteritis, enlarged lymph nodes are more common in bison. Inhalation pneumonia is also a common terminal stage of disease. The most consistent symptoms in pigs with acute MCF are fever, dyspnoea, foul-smelling nasal discharges, crumbling rhinitis, corneal oedema, erosions of the oral and nasalmucosa, uveitis, haematuria, reddened foci on the skin, reproductive losses (e.g., stillbirths, abortions), and neurological symptoms were also seen in some outbreaks. In pigs, the disease appears to be acute or per acute, but chronic form of disease lasting several weeks have also been observed (Brown *et al*., 2007).

Gross lesion. The gross lesions are generally widespread and involve most of the organs. Inflammation is often seen in the nasal and oral mucosa, with either focal or diffuse necrosis, erosion, or ulceration. Petechial haemorrhage, catarrhal exudate, and diphtheritic membranes are often observed in the respiratory tract (Sharma *et al*., 2019). Punctate or large ulcers are usually seen on the palate, gums, tips of the oral papillae, oesophagus, rumen, abomasum, and intestine. Lymph nodes may become engorged to the point of becoming unusable. Conjunctivitis and corneal oedema are caused by purulent inflammation of the

entire eye. In cattle and bison, urinary tract lesions characterised by multifocal areas of haemorrhage of the epithelial lining of the urinary bladder and swelling. Multiple raised pale foci (1-5 mm) on kidney surface are also present and these may extend into the cortex, but the degree varies within an animal.

Histopathological findings. The basic pathological feature of MCF is an interstitial accumulation of lymphocytes, and other mononuclear cells are found in a range of tissues (Liggittand DeMartini, 1980). The pathognomonic lesions are necrotic vasculitis with lymphoblast and macrophage infiltration of the tunica media and adventitia. These lesions may difficult to locate, especially in the more rapidly fatal cases. Presence of inclusion bodies are generally uncommon. Perivascular cuffing with lymphocyte is seen in most of the organs, and a characteristic vascular lesion is obliterate arteriopathy (Radostits *et al*., 2007). A non-suppurative meningoencephalitis with lymphocytic perivascular cuffing and a marked increase in the cellularity of the cerebrospinal fluid may also be present in the brain. Widespread vacuities is less common in deer, water buffalo, and bison (O'toole *et al*., 2002).

Diagnosis. MCF can be diagnosed through a combination of clinical manifestations, histopathology examination, and the presence of virus-specific antibodies or DNA in blood or tissue samples. Diagnosis of MCF is difficult and poses significant challenges to veterinarians because of the involvement of multiple systems and symptomatic similarity to other diseases in the field. Rinder pest, Bovine viral diarrhoea or mucosal disease, and bovine rhinotracheitis are the most common differential diagnoses. MCF can mimic rabies and tick-borne encephalitis when the CNS is intimately involved. While a history of interaction with a carrier species (wildebeest, sheep, and goats) is helpful. MCF viruses inactivate easily in dead animals, so samples should be taken as quickly as possible. *Alcelaphine Herpesvirus-1*(AlHV-1) can be isolated from the bovine thyroid turbinate cell line, co-cultured with peripheral blood leukocytes or disaggregated cells from affected tissues in bovine/ovine monolayer cell culture and can detected by immune fluorescence or immune cytochemistry (Russell *et al*., 2014). For isolation of virus, 10-20 ml anti-coagulated blood (EDTA) in live animals or a portion of organs (e.g., lung, spleen, lymph node, brain, adrenal gland, and intestinal wall), can be collect after death. The characteristic cytopathological effects (Cowdry type A intranuclear inclusion bodies) may develop after several passages with fresh susceptible cells (Hristov and Peshev, 2016). While OvHV-2 and CpHV-2 cannot be isolated in cell culture, but it has been possible to generate lymphoblastoid cell lines from infected deer and cattle. Thus, the diagnosis of OvHV-1 infection has previously relied on histopathological examination or the detection of antibodies that cross-react with AlHV-1 via serological tests. For tentative diagnosis of MCF, the age of the in-contact carrier species, grazing patterns, its food habitat, calving, type of rearing, and other factors is relevant. In practice, MCF is frequently confirmed by histopathologic evidence of multi-system lymphoid infiltration, degenerative epithelial lesions and disseminated vasculitis. Tissue samples to be collected for histopathology examination in cattle include, skin, liver, lung, brain, lymph nodes, kidney, adrenal gland, oesophagus, eye, oral epithelium, urinary bladder, thyroid, and heart muscles (Sharma *et al*., 2019). Intestinal and urogenital tissues are important, in bison. Because some MCF viruses cannot be isolated. PCR is preferred clinical diagnostic test to detect viral genome. However, a low amount of viral DNA in samples from sub-clinically infected animals can result in false negative results. The most tested tissues for PCR testing are peripheral anticoagulated (EDTA) blood, lymph node, kidney, intestinal wall, and brain. PCR can detect both OvHV-2 and AHV-1 viral DNA. Despite the fact that virus isolation has not been successful in OvHV-2 infections. Many studies have used this PCR assay to detect latent OvHV-2 infections in sheep. The primer (#556) binds to a region of low homology between OvHV-2 and AlHV-1, resulting in OvHV-2 specificity. When performed on infected sheep and other animals with clinical disease, a nested PCR targeting an ORF 75 of OvHV-2, which codes for the viral tegument protein, has shown promising results. It has been authorised by the World Organization for Animal Health (OIE) as a diagnostic test for OvHV-2 infection. The test has been widely used in veterinary diagnostic laboratories for many years, and it is considered to be the gold standard (Muller *et al*., 1998). However, its frequent usage in diagnostic laboratories to identify OvHV-2 DNA for confirmation of clinical SA-MCF may be troublesome because to the high likelihood of carryover amplicon contamination, which could result in false positive results. Furthermore, it can only be utilised as a qualitative method for establishing infection in the in vivo model, and not as a quantitative method. It is possible to confirm the diagnosis of MCF by PCR detection of the viral genome in PBL and tissues (Traul *et al*., 2007). Serological tests are more helpful in infected cattle and bison than in extremely vulnerable species such as deer, which frequently die before antibodies formed in their blood. These tests should be used in combination with histopathology and clinical results since stable incidental hosts may have antibodies to MCF viruses (Li *et al*., 2011). Serological tests for MCF viruses include immunofluorescence or immunoperoxidase tests (IPT), virus neutralization test (VNT), ELISA, and immunoblotting. For the screening of infection in susceptible animals, competitive ELISA is the test of choice. Although a positive serological test is indicative of infection, but it does not necessarily indicate the presence of clinical disease. It has been developed as an OvHV-2–specific test, using a glycoprotein antigen as the competitive inhibition (CI)-ELISA as the antigen (Alhajri *et al*., 2018). Because distinct herpesviruses, such as BHV-1 and BHV-4, share antigens, non-specificity in ELISA, IFA, and IPT tests may be observed in the results (Reid, 2004). Virus neutralising test detects neutralising antibodies in

animals infected with AlHV-1 or other related viruses in the Alcelaphine/Hippotraginae group, but it is of limited use in detecting antibodies against OvHV-2 or other related viruses in the Caprinae group due to low or no cross-reactivity of OvHV-2 neutralising antibodies to AlHV-1 in the Caprinae group (Taus *et al*., 2015).

Prevention and control. No practical, consistently effective treatments are available. Corticosteroids, antibiotics, antivirals, vitamins, and other supportive medications have all been mentioned as possible treatments (Penny, 1998). There have been reports of cattle recovering after treatment, usually with corticosteroids. However, the treatment's role has yet to be determined, as large numbers of cattle recover without treatment. At present, the only method of prevention is the proper management of susceptible species. Avoid co-farming of cattle with sheep and goat species. Avoid use of same mangers for both cattle and sheep. The best way to keep disease from spreading in vulnerable hosts is to keep them away from known carrier animals. Separation conditions may be influenced by the degree of susceptibility. Although cattle such as *Bos taurus* and *Bos indicus* should not be allowed near wildebeest, sheep associated MCF is uncommon in these animals, and there may be little to no morbidity if they come into contact with sheep (Aielloand Moses, 2016). The minimum distance required to avoid airborne transmission is yet unknown. In addition to the number of reservoir hosts shedding viruses, the amount of virus they shed, and environmental factors such as temperature and humidity, as well as the vulnerability of the incidental host, are all likely to have an effect. Fomites should be sterilised before coming into contact with susceptible incidental hosts, and they should not be allowed on pastures where reservoir hosts have recently grazed. The feed used by ewes and lambs should not be allowed to cattle. Control in zoos and wild animal parks is complicated by a large number of potentially susceptible species and MCF virus carriers, who are often unknown. An operation that depends upon mixing species, such as petting zoos, may wish to consider producing their own virus-free hosts for a zoo or wildlife parts could be viable (Li *et al*., 1999). Susceptible animals should be isolated from reservoir hosts or their environment as soon as possible during an outbreak to avoid further cases. Sick animals do not need to be culled or isolated, according to current opinion, because transmission from them is unlikely or uncommon. In sub-clinically infected or mildly affected animals, good husbandry, including reduced exposure to stressors, could be beneficial in reducing the number of cases, particularly in the more susceptible species. Recovered animals are usually immune to forthcoming infection. Still, no effective vaccine or immunization methods have been discovered. Attempts have been made using inactivated AlHV-1 virus, inactivated cell-free AlHV-1 virus in FCA, inactivated cell cultures of AlHV-1 (WCII strain in Freund's Complete Adjuvant (FCA), and most recently inactivated virus strain C500 from serially pass aged cell culture with emulgisen adjuvant and CpGoligodeoxy nucleotides (TLR9 agonist) (Parameswaran *et al*., 2014).

CONCLUSIONS

MCF is a fascinating and important disease with many unanswered questions about transmission, sporadic incidence, and pathogenesis. Since there is currently no effective treatment for MCF, disease management is solely based on prevention and control. The only effective strategy is to limit contact between MCF-susceptible species and natural hosts of the viruses, which is being made almost impossible by encroachment and settlement of wildlife areas. Vaccine production, efficient and prompt confirmatory diagnosis and genetic studies of WA-MCF may all be part of a three-pronged approach to integrated control of WA-MCF. Both wildebeest and sheep of any age should be considered possible sources of infection. Even though the mode of transmission is not well known, it is still best to avoid direct contact or indirect contact by personnel or fomites. Separation from sheep is important in the case of highly susceptible species like Bali cattle. It is necessary to conduct systematic and ongoing surveillance and monitoring of small ruminants. Diagnostic tests are time-consuming, require experience, and are insufficient to support active field surveillance, especially in hotspots. Since most of the infected animals die within two weeks, there is no time for lengthy diagnostic tests, sensitive and quick detection methods in the field are needed.

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REFERENCES

Aiello, S. E., & Moses, A. M. (2016). Ocular Neoplasia in Cattle.The Merck Veterinary Manual. (11th) Ed. Merck & Co. *In., Kenilworth, NJ, USA*.

Akula, S. M., Pramod, N. P., Wang, F. Z., & Chandran, B. (2001). Human herpesvirus 8 envelope-associated glycoprotein B interacts with heparansulfate-like moieties. *Virology*, *284*(2): 235-249.

Akula, S. M., Pramod, N. P., Wang, F. Z., & Chandran, B. (2002). Integrin α3β1 (CD 49c/29) is a cellular receptor for Kaposi's sarcoma-associated herpesvirus (KSHV/HHV-8) entry into the target cells. *Cell*, *108*(3): 407-419.

Albini, S., Zimmermann, W., Neff, F., Ehlers, B., Häni, H., Li, H., & Ackermann, M. (2003). Porcine malignant catarrhal fever: diagnostic findings and first detection of the pathogenic agent in diseased swine in Switzerland. *Schweizer Archiv Fur Tierheilkunde*, *145*(2): 61-68.

Alcaraz, A., Warren, A., Jackson, C., Gold, J., McCoy, M., Cheong, S. H., & Li, H. (2009). Naturally occurring sheep-associated malignant catarrhal fever in North American pigs. *Journal of Veterinary Diagnostic Investigation*, *21*(2): 250-253.

Alhajri, S. M., Cunha, C. W., Knowles, D. P., Li, H., & Taus, N. S. (2018). Evaluation of glycoprotein Ov8 as a potential antigen for an OvHV-2-specific diagnostic assay. *Plos One*, *13*(7): e0200130.

Alhajri, S. M., Cunha, C. W., Nicola, A. V., Aguilar, H. C., Li, H., & Taus, N. S. (2017). Ovine herpesvirus 2 glycoproteins B, H, and L are sufficient for, and viral glycoprotein Ov8 can enhance, cell-cell membrane fusion. *Journal of Virology*, *91*(6): e02454-16.

Amoroso, M.G., Galiero, G., & Fusco, G. (2017). Genetic characterization of ovine herpesvirus 2 strains involved in water buffaloes malignant catarrhal fever outbreaks in Southern Italy. *Veterinary Microbiology*, *199*: 31-35.

Banumathi, N., Sood, R., Pativ, S. S., Subramanlan, M., & Pradhaw, H. K. (2008). Genomic detection of ovine herpesvirus-2 in South Indian sheep and goat. *Indian Journal of Animal Sciences* (India), *78*: 13–16.

Baxter, S. I. F., Pow, I., Bridgen, A., & Reid, H. W. (1993). PCR detection of the sheep-associated agent of malignant catarrhal fever. *Archives of Virology*, *132*(1-2): 145-159.

Beatson, N. S. (1985). Field observations of malignant catarrhal fever in red deer in New Zealand. In Biology of deer production. Proceedings of an *International Conference held at Dunedin*, New Zealand, 13-18 February 1983 (pp. 135-137). Royal Society of New Zealand.

Bedelian, C., Nkedianye, D., & Herrero, M. (2007). Maasai perception of the impact and incidence of malignant catarrhal fever (MCF) in southern Kenya. *Preventive Veterinary Medicine*, *78*(3-4): 296-316.

Berezowski, J. A., Appleyard, G. D., Crawford, T. B., Haigh, J., Li, H., Middleton, D. M., & Woodbury, M. (2005). An outbreak of sheep-associated malignant catarrhal fever in bison (Bison bison) after exposure to sheep at a public auction sale. *Journal of Veterinary Diagnostic Investigation*, *17*(1): 55-58.

Brown, C. C., Baker, D. C., Barker, I. K. (2007). Alimentary system. In: Maxie M. G. Ed. Pathology of Domestic Animals. Vol 2. 5th ed. Edin-burgh, UK: Saunders Elsevier; 2007: 152–159.

Collery, P., & Foley, A. (1996).An outbreak of malignant catarrhal fever in cattle in the Republic of Ireland. *Veterinary Record*, *139*(1): 16-17.

Cunha, C. W., Taus, N. S., Dewals, B. G., Vanderplasschen, A., Knowles, D. P., & Li, H. (2016). Replacement of glycoprotein B in alcelaphine herpesvirus 1 by its ovine herpesvirus 2 homolog: implications in vaccine development for sheep-associated malignant catarrhal fever. *Msphere*, *1*(4): e00108-16.

Dabak, M., & H. Bulut (2003). Outbreak of malignant catarrhal fever in cattle in Turkey. The Veterinary Record *152*(8): 240-241.

Davison, A. J., Eberle, R., Ehlers, B., Hayward, G. S., McGeoch, D. J., Minson, A. C., & Thiry, E. (2009). The order herpesvirales. *Archives of Virology*, *154*(1): 171-177.

Ehlers, B., Borchers, K., Grund, C., Fro, K. & Ludwig, H. (1999). Detection of new DNA polymerase genes of known and potentially novel herpesviruses by PCR with degenerate and deoxyinosine-substituted primers. *Virus Genes*, *18*(3): 211-220.

Gailbreath, K. L., Taus, N. S., Cunha, C. W., Knowles, D. P. & Li, H. (2008). Experimental infection of rabbits with ovine herpesvirus 2 from sheep nasal secretions. *Veterinary Microbiology*, *132*(1-2): 65-73.

Hristov, M. V. & Peshev, R. D. (2016). Isolation and identification of malignant catarrhal fever virus in cell cultures. *Bulgarian Journal of Veterinary Medicine*, *19*(4): 263-273

Jacobsen, B., Thies, K., von Altrock, A., Förster, C., König, M., & Baumgärtner, W. (2007). Malignant catarrhal fever-like lesions associated with ovine herpesvirus-2 infection in three goats. *Veterinary Microbiology*, *124*(3-4): 353-357.

Keel, M. K., Gage, P. J., Noon, T. H., Bradley, G. A., & Collins, J. K. (2003). Caprine herpesvirus-2 in association with naturally occurring malignant catarrhal fever in captive sika deer (*Cervus nippon*). *Journal of Veterinary Diagnostic Investigation*, *15*(2): 179-183.

Kleiboeker, S. B., Miller, M. A., Schommer, S. K., Ramos-Vara, J. A., Boucher, M., & Turnquist, S. E. (2002). Detection and multigenic characterization of a herpesvirus associated with malignant catarrhal fever in white-tailed deer (*Odocoileus virginianus*) from Missouri. *Journal of Clinical Microbiology*, *40*(4): 1311-1318.

Klieforth, R., Maalouf, G., Stalis, I., Terio, K., Janssen, D. & Schrenzel, M. (2002). Malignant catarrhal fever-like disease in Barbary red deer (*Cervuselaphus barbarus*) naturally infected with a virus resembling alcelaphine herpesvirus 2. *Journal of Clinical Microbiology*, *40*(9): 3381-3390.

Kumar, N., Sood, R., Pateriya, A.K., Venkatesakumar, E., Ramprabhu, R., Dixit, R., & Singh, V. P. (2021). First molecular evidence and genetic characterization of Ovine Herpesvirus 2 in multiple animal species in India. *Frontiers in Veterinary Science*, *8*, 44.

Lankester, F., Lugelo, A., Mnyambwa, N., Ndabigaye, A., Keyyu, J., Kazwala, R., & Russell, G. C. (2015). Alcelaphine herpesvirus-1 (*malignant catarrhal fever virus*) in wildebeest placenta: genetic variation of ORF50 and A9. 5 alleles. *Plos One*, *10*(5): e0124121.

Li, H., Cunha, C. W., Taus, N. S., & Knowles, D. P. (2014). Malignant catarrhal fever: inching toward understanding. *Annu. Rev. Anim. Biosci.*, *2*(1): 209-233.

Li, H., Cunha, C. W., & Taus, N. S. (2011). Malignant catarrhal fever: understanding molecular diagnostics in context of epidemiology. *International Journal of Molecular Sciences*, *12*(10): 6881-6893.

Li, H., Gailbreath, K., Flach, E. J., Taus, N. S., Cooley, J., Keller, J. & Crawford, T. B. (2005). A novel subgroup of rhadinoviruses in ruminants. *Journal of General Virology*, *86*(11): 3021-3026.

Li, H., Karney, G., O'Toole, D. & Crawford, T. B. (2008). Long distance spread of malignant catarrhal fever virus from feedlot lambs to ranch bison. *The Canadian Veterinary Journal*, *49*(2): 183.

Li, H., Keller, J., Knowles, D. P. & Crawford, T. B. (2001). Recognition of another member of the malignant catarrhal fever virus group: an endemic gammaherpesvirus in domestic goats. *Journal of General Virology*, *82*(1): 227-232.

Li, H., O'Toole, D., Kim, O., Oaks, J. L., & Crawford, T. B. (2005). Malignant catarrhal fever-like disease in sheep after intranasal inoculation with ovine herpesvirus-2. *Journal of Veterinary Diagnostic Investigation*, *17*(2): 171-175.

Li, H., Snowder, G., O'Toole, D., & Crawford, T. B. (1998). Transmission of ovine herpesvirus 2 in lambs. *Journal of Clinical Microbiology*, *36*(1): 223-226.

Li, H., Taus, N. S., Jones, C., Murphy, B., Evermann, J. F., & Crawford, T. B. (2006). A devastating outbreak of malignant catarrhal fever in a bison feedlot. *Journal of Veterinary Diagnostic Investigation*, *18*(1): 119-123.

Li, H., Taus, N. S., Lewis, G. S., Kim, O., Traul, D. L., & Crawford, T. B. (2004). Shedding of ovine herpesvirus 2 in sheep nasal secretions: the predominant mode for transmission. *Journal of Clinical Microbiology*, *42*(12): 5558-5564.

Li, H., Westover, W. C., & Crawford, T. B. (1999). Sheep-associated malignant catarrhal fever in a petting zoo. *Journal of Zoo and Wildlife Medicine*, 408-412.

Liggitt, H. D., & DeMartini, J. C. (1980). The Pathomorphology of Malignant Catarrhal Fever: II. Multisystemic Epithelial Lesions. *Veterinary Pathology*, *17*(1): 73-83.

Modesto, P., Grattarola, C., Biolatti, C., Varello, K., Casalone, C., Mandola, M. L., & Acutis, P. L. (2015). First report of malignant catarrhal fever in a captive pudu (*Pudupuda*). *Research in Veterinary Science*, *99*: 212-214.

Müller-Doblies, U. U., Li, H., Hauser, B., Adler, H., & Ackermann, M. (1998). Field validation of laboratory tests for clinical diagnosis of sheep-associated malignant catarrhal fever. *Journal of Clinical Microbiology*, *36*(10): 2970-2972.

Mushi, E. Z., Rossiter, P.B., Jessett, D., & Karstad, L. (1981). Isolation and characterization of a herpesvirus from topi (*Damaliscus korrigum, Ogilby*). *Journal of Comparative Pathology*, *91*(1): 63-68.

Mushi, E. Z., Rurangirwa, F. R., & Karstad, L. (1981). Shedding of malignant catarrhal fever virus by wildebeest calves. *Veterinary Microbiology*, *6*(4): 281-286.

OIE (2004). Malignant catarrhal fever. In: OIE Manual of Diagnostic Tests and Vaccines for Terrestrial Animal, fifth ed., France, pp. 570–579.

Orono, S. A., Gitao, G. C., Mpatswenumugabo, J. P., Chepkwony, M., Mutisya, C., Okoth, E., & Cook, E. A. J. (2019). Field Validation of clinical and laboratory diagnosis of wildebeest associated malignant catarrhal fever in cattle. *BMC Vety. Res.*, *15*(1): 1-10.

O'Toole, D., Li, H., Miller, D., Williams, W. R., & Crawford, T. B. (1997). Chronic and recovered cases of sheep associated malignant catarrhal fever in cattle. *Veterinary Record*, *140*(20): 519-524.

O'toole, D., Li, H., Sourk, C., Montgomery, D. L. & Crawford, T. B. (2002). Malignant catarrhal fever in a bison (*Bison bison*) feedlot, 1993–2000. *Journal of Veterinary Diagnostic Investigation*, *14*(3): 183-193.

O'toole, D., Taus, N. S., Montgomery, D. L., Oaks, J. L., Crawford, T. B., & Li, H. (2007). Intra-nasal inoculation of American bison (*Bison bison*) with ovine herpesvirus-2 (OvHV-2) reliably reproduces malignant catarrhal fever. *Veterinary Pathology*, *44*(5): 655-662.

Palmer, M. V., Thacker, T. C., Madison, R. J., Koster, L. G., Swenson, S. L. & Li, H. (2013). Active and latent ovine herpesvirus-2 (OvHV-2) infection in a herd of captive white-tailed deer (*Odocoileus virginianus*). *Journal of Comparative Pathology*, *149*(3): 162-166.

Parameswaran, N., Russell, G. C., Bartley, K., Grant, D. M., Deane, D., Todd, H., & Haig, D. M. (2014). The effect of the TLR9 ligand CpG-oligodeoxynucleotide on the protective immune response to alcelaphine herpesvirus-1-mediated malignant catarrhal fever in cattle. *Veterinary Research*, *45*(1): 1-11.

Parihar, N. S., Rajya, B. S., & Gill, B. S. (1975). Occurrence of malignant catarrhal fever in India. *Indian Vet. J.*, *52*: 857-859.

Penny, C. (1998). Recovery of cattle from malignant catarrhal fever. *Veterinary Record*, *142*(9).

Premkrishnan, G. N., Sood, R., Hemadri, D., Chanu, K.V., Khandia, R., Bhat, S., & Bhatia, S. (2015). Cross-sectional study indicates nearly a quarter of sheep population in Karnataka state of India is infected with ovine herpesvirus 2. *Virus Disease*, *26*(3): 180-188.

Radostits, O. M., Gay, C. C., Hinchcliff, K. W., & Constable, P. D. (2007). Veterinary Medicine: A Textbook of the Diseases of Cattle, Horses, Sheep, Pigs and Goats. 10th ed. Edinburgh, UK: Saunders Elsevier. 1245–1248.

Reid, H. W., & Van Vuuren, M. (2004). Malignant catarrhal fever. In: Coetzer JAW, Tustin RC (Eds). Infectious Diseases of Livestock. 2nd ed. Oxford, UK: Oxford University Press; 895–908.

Russell, G. C., Scholes, S. F., Twomey, D. F., Courtenay, A. E., Grant, D. M., Lamond, B., Norris, D., Willoughby, K., Haig, D. M., & Stewart, J. P. (2014). Analysis of the genetic diversity of ovine herpesvirus 2 in samples from livestock with malignant catarrhal fever. *Vet. Microbiol.*, *172*(1-2): 63-71.

Russell, G. C., Stewart, J. P., & Haig, D. M. (2009). Malignant catarrhal fever: a review. *The Veterinary Journal*, *179*(3): 324-335.

Saura, H., Al-Saadi, M., Stewart, J. P., & Kipar, A. (2018). New Insights into the Pathogenesis of Vasculitis in Malignant Catarrhal Fever. *Journal of Comparative Pathology: 158*, 98.

Schultheiss, P. C., Collins, J. K., Spraker, T. R., & DeMartini, J. C. (2000). Epizootic malignant catarrhal fever in three bison herds: differences from cattle and association with ovine herpesvirus 2. *Journal of Veterinary Diagnostic Investigation*, *12*(6): 497-502.

Sharma, B., Parul, S., Basak, G. & Mishra, R. (2019). Malignant catarrhal fever (MCF): An emerging threat. *Journal of Entomology and Zoology Studies*, *7*: 26-32.

Simon, S., Li, H., O'Toole, D., Crawford, T. B., & Oaks, J. L. (2003). The vascular lesions of a cow and bison with sheep-associated malignant catarrhal fever contain ovine herpesvirus 2-infected CD8+ T lymphocytes. *Journal of General Virology*, *84*(8): 2009-2013.

Singh, G., Singh, B., Gupta, P. P., & Hothi, D. S. (1979). Epizootiological observations on malignant catarrhal fever and transmission of the disease in buffalo calves (*Bubalus bubalis*). *Acta Veterinaria Brno*, *48*(1-4): 95-103.

Sood, R., Hemadri, D., & Bhatia, S. (2013). Sheep associated malignant catarrhal fever: an emerging disease of bovids in India. *Indian Journal of Virology*, *24*(3): 321-331.

Sood, R., Manoj, K., Bhatia, S., Pateriya, A. K., Khandia, R., Siddiqui, A., & Venkatesha, M. D. (2012). Ovine herpesvirus type 2 infection in captive bison in India. *Veterinary Record*, *170*(25): 654.

Swa, S., Wright, H., Thomson, J., Reid, H., & Haig, D. (2001). Constitutive activation of Lck and Fyn tyrosine kinases in large granular lymphocytes infected with the γ-herpesvirus agents of malignant catarrhal fever. *Immunology*, *102*(1): 44-52.

Taus, N. S., Cunha, C. W., Marquard, J., O'Toole, D., & Li, H. (2015). Cross-reactivity of neutralizing antibodies among malignant catarrhal fever viruses. *PloS one*, *10*(12): e0145073.

Taus, N. S., Oaks, J. L., Gailbreath, K., Traul, D. L., O'Toole, D., & Li, H. (2006). Experimental aerosol infection of cattle (*Bos taurus*) with ovine herpesvirus 2 using

nasal secretions from infected sheep. *Veterinary Microbiology*, *116*(1-3): 29-36.

Traul, D. L., Taus, N. S., Oaks, J. L., Toole, D. O., Rurangirwa, F. R., Baszler, T. V. & Li, H. (2007). Validation of non-nested and real-time PCR for diagnosis of sheep-associated malignant catarrhal fever in clinical samples. *Journal of Veterinary Diagnostic Investigation*, *19*(4): 405-408.

Wani, S. A., Bhat, M. A., Samanta, I., Buchoo, B. A., Ishaq, S. M., Pandit, F., & Buchh, A. S. (2004). Clinical, serological, and molecular evidence of sheep-associated malignant catarrhal fever in India. *The Veterinary Record*, *155*(8): 242-244.

Whitaker, K. A., Wessels, M. E., Campbell, I. & Russell, G. C. (2007). Outbreak of wildebeest-associated malignant catarrhal fever in Ankole cattle. *Veterinary Record*, *161*(20): 692-695.

Yus, E., Guitan, J., Diaz, A., & Sanjuan, M. L. (1999). Outbreak of malignant catarrhal fever in cattle in Spain. *Vet. Rec.*, *145*: 466–467.

Zakharova, O., Toropova, N., Burova, O., Titov, I., Meltsov, I., & Blokhin, A. (2020). Malignant catarrhal fever in cattle in the irkutsk region. *Journal of Veterinary Research*, *64*(2): 215.

Zemljič, T., Pot, S. A., Haessig, M., & Spiess, B. M. (2012). Clinical ocular findings in cows with malignant catarrhal fever: ocular disease progression and outcome in 25 cases (2007–2010). *Veterinary Ophthalmology*, *15*(1), 46-52.

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