Veterinary Microbiology 165 (2013) 38–44

Contents lists available at SciVerse ScienceDirect

Veterinary Microbiology

journal homepage: www.elsevier.com/locate/vetmic

Review

Peste des petits ruminants, the next eradicated animal disease?

Emmanuel Albina [a,c,*], Olivier Kwiatek [b,c], Cécile Minet [b,c], Renaud Lancelot [b,c], Renata Servan de Almeida [b,c], Geneviève Libeau [b,c]

[a] CIRAD, UMR CMAEE, F-97170 Petit-Bourg, Guadeloupe, France
[b] CIRAD, UMR CMAEE, F-34398 Montpellier, France
[c] INRA, UMR1309 CMAEE, F-34398 Montpellier, France

ARTICLE INFO

Article history:
Received 1 September 2012
Received in revised form 3 December 2012
Accepted 17 December 2012

Keywords:
Peste des petits ruminants
Morbillivirus
Vaccine
Eradication
Epidemiology

ABSTRACT

Peste des Petits Ruminants (PPR) is a widespread viral disease caused by a *Morbillivirus* (*Paramyxoviridae*). There is a single serotype of PPR virus, but four distinct genetic lineages. Morbidity and mortality are high when occurring in naive sheep and goats populations. Cattle and African buffaloes (*Syncerus caffer*) are asymptomatically infected. Other wild ruminants and camels may express clinical signs and mortality. PPR has recently spread in southern and northern Africa, and in central and far-east Asia. More than one billion sheep and goats worldwide are at risk. PPR is also present in Europe through western Turkey. Because of its clinical incidence and the restrictions on animal movements, PPR is a disease of major economic importance. A live attenuated vaccine was developed in the 1980s, and has been widely used in sheep and goats. Current researches aim (i) to make it more thermotolerant for use in countries with limited cold chain, and (ii) to add a DIVA mark to shorten and reduce the cost of final eradication. Rinderpest virus—another *Morbillivirus*—was the first animal virus to be eradicated from Earth. PPRV has been proposed as the next candidate. Considering its wide distribution and its multiple target host species which have an intense mobility, it will be a long process that cannot exclusively rely on mass vaccination. PPR specific epidemiological features and socio-economic considerations will also have to be taken into account, and sustained international, coordinated, and funded strategy based on a regional approach of PPR control will be the guarantee toward success.

Contents

1. Introduction

Peste des petits ruminants (PPR) is one of the most serious diseases of small ruminants, caused by a *Morbillivirus* that belongs to the *Paramyxoviridae* family. The

* Corresponding author at: UMR CIRAD/INRA CMAEE, Station de Duclos - Prise d'Eau - 97170 Petit-Bourg, Guadeloupe, France.
Tel.: +590 5 90 25 54 44; fax: +590 5 90 94 03 96.
E-mail address: Emmanuel.Albina@antilles.inra.fr (E. Albina).

0378-1135/$ – see front matter © 2012 Elsevier B.V. All rights reserved.
http://dx.doi.org/10.1016/j.vetmic.2012.12.013

infection has long been considered as caused by a variant of rinderpest virus, adapted to small ruminants. The recognition of PPR virus as a novel member of the *Morbillivirus* genus occurred only in the late 70s by using more sensitive laboratory techniques (Gibbs et al., 1979). Because of the dramatic clinical incidence and associated restrictions on animal and product movements, PPR is considered as a disease of major economic impact and has to be notified to the World Animal Health Organization (OIE). A homologous vaccine was made available in the 80s providing life-long immunity after a single injection (Diallo, 2003). Since rinderpest was successfully eradicated from Earth in 2011, there are now growing interests in the eradication of PPR. This goal might be reasonably achievable if lessons learnt from the rinderpest eradication are amended according to the epidemiological specificities of PPR, and organizational/financial issues. In this paper, we present and discuss the scientific bases for PPR surveillance and control, with a special focus on Africa which might be the most challenging continent for PPR eradication, as it was for rinderpest.

1.1. An expanding fatal disease of small ruminants

PPR is one of the most widespread, infectious and contagious diseases of sheep and goats, with mortality rates exceeding 90% in immunologically naive populations. Acute PPR first results in a sudden dullness of infected animals, with high fever and inappetence. One or two days later, congestion of oral, ocular and nasal mucosae leads to serous discharges that later on become more abundant and mucopurulent (Roeder and Obi, 1999; Fig. 1). Bronchopneumonia, revealed by productive cough and dyspnea, and diarrhea usually appears 3 days after the oral lesions. As a consequence of pneumonia and dehydration caused by diarrhea, severely affected animals may die within 5–10 days after the onset of clinical signs (Diallo, 2006). Abortions are often observed during PPR outbreaks, caused by PPRV alone or in combination with other pathogens (Kulkarni et al., 1996; Abubakar et al., 2008). At an early stage of infection, virus excretion is massive in the exhaled air. Nasal and ocular discharges, saliva, and feces also contain large amounts of virus (Abubakar et al., 2012). Since PPRV is quickly inactivated in the environment, its transmission most often occurs by direct contact between infected and healthy animals. However, indirect transmission through recently (within hours) contaminated material cannot be excluded and should be considered in epidemiological models. The high multisystemic virulence of PPRV has been demonstrated in experimental and natural infections (Eligulashvili et al., 1999; Couacy-Hymann et al., 2007b). The virus has a tropism for epithelial and lymphoid cells, thus damaging epithelial cells of the intestinal and respiratory tracts and also circulating and resident lymphocytes. The resulting lesions are directly responsible for the respiratory and digestive disorders and the profound immunosuppression that follow the infection. Despite the immunosuppressive effect of the virus favoring opportunistic pathogens (Kerdiles et al., 2006), recovering animals always develop a strong life-long immunity clearing the virus.

Fig. 1. Goat from Senegal infected with PPR virus and showing apathy and sero-mucopurulent nasal discharges resulting from bacterial superinfection (courtesy of Habib Salami).

PPRV mostly affects sheep and goats although goats are often more severely affected than sheep (Lefèvre and Diallo, 1990). However, variable seroprevalence has been observed in sheep and goats after an outbreak (Özkul et al., 2002; Abraham et al., 2005; Swai et al., 2009; Ayari-Fakhfakh et al., 2011). Many factors may explained these differences like the management practices, host density, strain virulence (Couacy-Hymann et al., 2007a), as well as host species and breed (Diop et al., 2005). For instance, Sahelian goats are considered as more resistant than Guinean dwarf goats, while Alpine goats have been found very sensitive after experimental infections (Hammouchi et al., 2012). PPRV is not considered as pathogenic to cattle, domestic and wild African buffaloes (*Syncerus caffer*) although 10% of them may seroconvert when exposed to PPRV in enzootic regions (Özkul et al., 2002; Abraham et al., 2005; Couacy-Hymann et al., 2005). However, high case fatality rates (96%) have been reported and the disease experimentally reproduced in domestic Indian buffaloes (*Bubalus bubalis*) in India (Govindarajan et al., 1997). Additionally, PPR is now recognized as an emerging disease in camelids. A respiratory syndrome was the main sign in Ethiopia and Sudan (Roger et al., 2000; Khalafalla et al., 2010). Whether PPRV-infected and sick buffaloes and camels are source of infection for small ruminants remains unclear. Other wild ruminants, including representatives of the *Gazellinae*, *Tragelaphinae* and *Caprinae* subfamilies, may express a serious illness and mortality. In specific conditions, wildlife may have played an important role in PPR epidemiology in the Arabian Peninsula (Kinne et al., 2010). In areas where PPRV has been present for a long time, we have some evidences that PPRV might represent a threat for wildlife. As for rinderpest, wildlife is more likely to be a victim rather than a reservoir for the PPRV (Anderson, 1995; Couacy-Hymann et al., 2005). However, this is a domain where knowledge remains scarce and should deserve more attention since PPR is progressing southward in Africa where wild ruminant density, as well as sheep and goat density, are high. In particular, little is known regarding virus excretion in infected camels, cattle and wildlife, as well as PPRV persistence in the

environment. It is even the case in goats in which PPRV might be excreted in the feces during at least 2 month after natural infection (Ezeibe et al., 2008; Abubakar et al., 2012). This topic should deserve more attention and would be useful to improve PPRV transmission models and risk analysis for the introduction of PPRV in disease-free areas (livestock trade, transhumance).

Initially described in 1942 (Gargadennec and Lalanne, 1942), PPR has long been considered as confined to West Africa. The first PPR observation outside West Africa was in Sudan, between 1970 and 1972 (El Hag Ali and Taylor, 1984). In 1983, it was confirmed in the Arabic Peninsula and subsequently in Asia (Taylor et al., 1990; Maillard et al., 2008). In recent years, field data and laboratory findings have confirmed the dramatic spread of PPR toward the south of Africa, affecting Gabon, Democratic Republic of Congo, Somalia, Kenya and Tanzania (Swai et al., 2009). In October 2012, PPR has been reported for the first time in Angola (OIE notification). The risk of PPR introduction is now high for neighboring countries with major sheep and goat populations like Mozambique or Zambia. Apart from Egypt which is infected at least since 1989 (Ismail and House, 1990), the Moroccan outbreak in 2008 was the first PPR incursion in North Africa. PPR has now been identified in Tunisia (Ayari-Fakhfakh et al., 2011) and Algeria (De Nardi et al., 2012). The current distribution of the disease is shown in Fig. 3.

1.2. A pleomorphic causative agent

Like the closely related measles, rinderpest, carnivore and marine mammal morbilliviruses, PPRV is an enveloped and pleomorphic RNA virus with a size of 150–700 nm (Barrett, 1999). Its genome consists of 15,948 nucleotides assembled in a unique negative single strand RNA molecule and encodes 6 structural proteins (Fig. 2): the major nucleoprotein (N), the phosphoprotein (P), the matrix protein (M), the large protein (L), and the two external fusion (F) and haemagglutinin (H) glycoproteins that elicit protection against the disease in infected or vaccinated animals (Diallo et al., 2002; Berhe et al., 2003). The viral genomic RNA and N, P and L proteins form the ribonucleoprotein (RNP), which is the minimal essential structure for viral replication in cells (Bailey et al., 2007).

Phylogenetic studies on the N or F gene sequences have shown that PPRV strains can be split into four different lineages (Shaila et al., 1996; Kwiatek et al., 2011). The three first lineages were historically settled in Africa according to the apparent spread of the virus from West to East Africa (Banyard et al., 2010). The fourth lineage was until recently confined to Asia, including Turkey and the Arabic peninsula. Within a remarkably short time, it spread to a large part of the African continent. It is now found from Sudan to northern Africa (Algeria, Morocco), as well as central Africa and the Gulf of Guinea (Khalafalla et al., 2010; Kwiatek et al., 2011; Fig. 3). In Senegal and Mauritania, a similar scenario has occurred with lineage II, originating from Central Africa and moving to West Africa whereas in the 1980s, lineage I was the dominant, if not the single lineage found there.

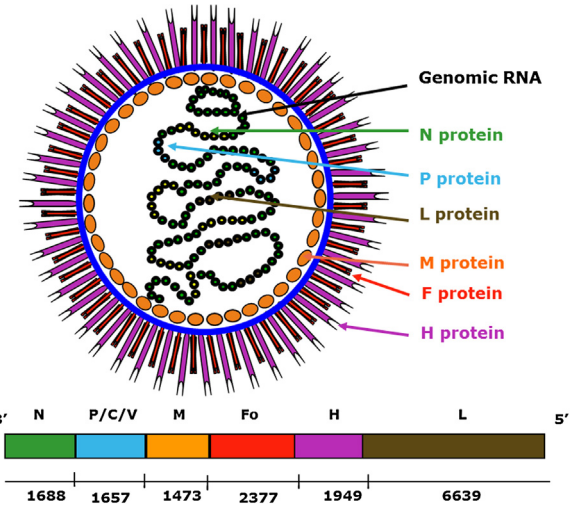

Fig. 2. Schematic representation of the PPR *morbillivirus* (courtesy of Djénéba Keita).

1.3. State of the art in diagnosis and control strategies

PPRV is routinely identified on the basis of clinical examination, gross pathology and histological findings. Clinical signs and lesions can be misleading for PPR diagnosis since other diseases, including pasteurellosis or contagious caprine pleuropneumonia, have similar consequences. Fortunately, several rapid, specific and sensitive laboratory methods are available for confirmation. The most popular techniques for virus detection are based on molecular biology. Conventional reverse transcription polymerase chain reaction (RT-PCR) is routinely used in most laboratories due to its very high specificity and sensitivity. More recently, one-step real-time RT-PCRs have been developed and shown to be the most sensitive techniques for PPRV genome detection (Bao et al., 2008; Kwiatek et al., 2010). When PCR machines are not accessible, immunocapture ELISA (ICE) can be used. It is rapid, specific and rather sensitive for PPRV antigen detection in sick animals (Libeau et al., 1994). When confirmation of the first outbreak or further characterization of the virus outbreak is required, the virus must be isolated and grown in cell cultures. Since the virus is circulating and excreted for approximately 10 days after the onset of fever, samples, including blood, body fluids (lachrymal and nasal discharges) and damaged organs and tissues, must be collected during the acute phase of the disease.

Virus isolation is time consuming and cumbersome. Moreover, the preservation of samples collected under field conditions is not always adequate for successful laboratory results. African green monkey kidney cells (Vero) have been for a long time the cells of choice for the isolation and propagation of PPRV. However, some isolates may not grow well in these cells. Recently, transformed monkey cells expressing sheep/goat signaling lymphocytic activation molecules (SLAM or CD150), the virus cellular receptors, have been shown to possess increased sensitivity (Adombi et al., 2011).

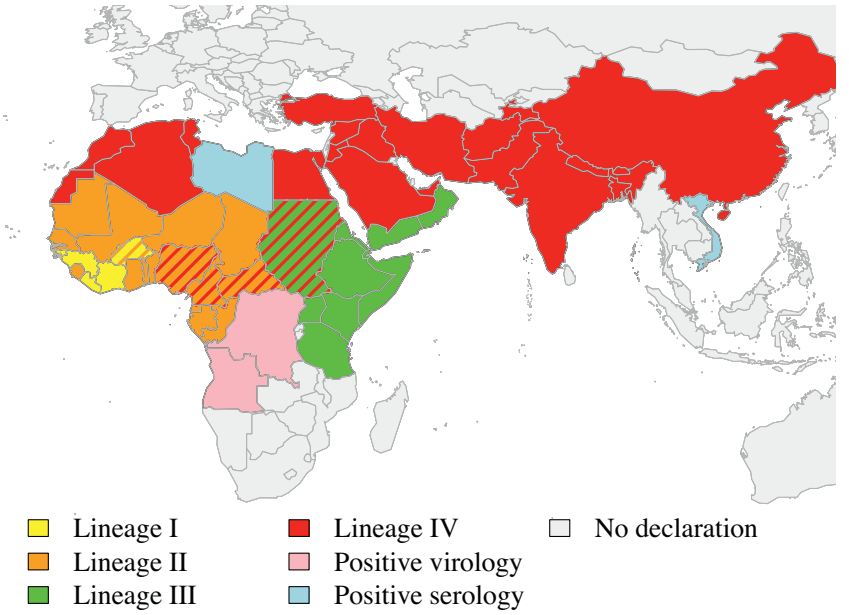

Fig. 3. Worldwide cumulative distribution of the four PPR virus lineages. Different colors show different lineages and hached bars represent the last identified lineage in the corresponding country. The pink color indicates virological evidence of PPR infection. The blue color indicates serological evidence of PPR infection but no virus isolated. The grey color indicates missing information or disease never reported.

The host antibody response is detectable around 10 days after the first contact with the virus and usually persists for the rest of the economic life. The most reliable and rapid test for antibody detection is a competition ELISA based on a monoclonal antibody directed against the virus nucleoprotein (Libeau et al., 1995). The virus neutralization test (VNT) is also an OIE prescribed reference method. Because VNT necessitates cell and virus cultures, and thus takes several days before result outcome, it is not used as a routine test. It is mostly used in reference laboratories to confirm unclear results.

Once confirmed, the most effective way to control PPR in a given area is mass immunization of small ruminants. Very efficient commercial vaccines are available. They confer a life-long immunity after a single administration. A homologous vaccine has been developed in the 1980s after the successful attenuation of the Nigeria 75/1 strain through multiple passages on Vero cells (Diallo, 2003). This vaccine provides a life-long immunity against PPR after a single shot and is used worldwide. However, it has a low thermal stability: half-life of 2–6 h at 37 °C after reconstitution (Diallo, 2004). To overcome this, the vaccine strain has been mixed with cryo-protectant mixture containing trehalose. Thus, preservation of sufficient virus titer could be extended to 5–14 days at 45 °C in the lyophilized form, and 21 h at 37 °C after reconstitution (Worrall et al., 2000; Silva et al., 2011). These thermostabilizing additives are compatible with the shipment of the vaccine to remote areas without the need for a cold chain. Alternative thermotolerant PPR-recombinant poxvirus vaccines have been engineered in the past (Jones et al., 1993; Diallo et al., 2002, 2007; Berhe et al., 2003), but none of them have yet been launched in the market and used in the field. To improve the control of PPR in the field, it might be interesting to develop a DIVA vaccine (differentiating infected from vaccinated animals). Such a DIVA vaccine associated to a performing DIVA diagnosis may only be important when PPR becomes close to elimination, to reveal a low-noise PPRV transmission and trigger the reinforcement of vaccination campaigns and disease surveillance based for instance on a test-and-removal strategy (elimination of animals tested with infection antibodies) to speed up the eradication. It could also be a worthwhile tool in case of re-emerging outbreaks in a free-zone since an emergency vaccination plan and a parallel test-and-removal control strategy could rapidly blow out a new epizootics. However, the DIVA vaccine is not an urgent issue because the existing vaccines, possibly improved for better thermotolerance, are fully efficient for the early stages of PPR control. Recombinant poxviruses are theoretically DIVA vaccines since the antigens not included in the vaccines can serve in the serological tests. Another interesting option is to modify the existing and widely used attenuated vaccine. Several research groups are working on this possibility by using reverse genetics (Minet et al., 2009; Hu et al., 2012). Again, the time before a commercial vaccine will become available may cover several years. Several African and Middle East laboratories have the capacity to produce and deliver tens of millions of PPR vaccine doses within months, like it was the case in Morocco after PPR emergence in 2008. A major point is the necessity of reliable quality controls before vaccine batch release. With this respect, the PANVAC laboratory based in Addis Ababa is an outstanding tool to ensure the quality of PPR vaccines used in the vaccination campaigns. The number of vaccine doses per vial will also need to be adapted to field situations. For instance, vials of >100 doses should be avoided in areas where sheep and goat

flocks are tiny. Given adequate manufacturing process, most of the eradication program can be achieved using the current vaccines.

In developing countries that are endemically infected with PPRV, vaccination is often not consistently applied during the inter-epidemic waves. Emergency vaccination alone does not prevent heavy economic losses arising from animal mortality and morbidity since several days are required for a protective immune response. Antivirals as emergency tools against PPR are not yet available, but promising results have been recently generated based on the use of RNA interference. In vitro, 99.99% of PPRV replication could be inhibited by small interfering RNA molecules (Servan de Almeida et al., 2007; Keita et al., 2008). The next challenge for such strategies will be the development of efficient and cheap in vivo delivery systems.

1.4. Is PPR eradicable?

In June 2011, OIE has officially declared a world free of rinderpest. It is only the second mammal disease after smallpox in 1980 but the first animal disease that has been eradicated in the world. The eradication process took more than five decades and required at least five consecutive large international programs: (1) the Joint Program 15 (JP15, 1962–1976), (2) the Pan African Rinderpest Campaign (PARC, 1986–1999), (3) the West Asia Rinderpest Eradication Campaign (WAREC, 1989–1994), (4) the Pan African Program for the Control of Epizootics (PACE) 1999–2006, and (5) the Global Rinderpest Eradication Program (GREP, 1980–2010). This long time before reaching rinderpest eradication was the consequence of various factors, including insufficient mobilization of international authorities, political instability in affected countries and the lack of consolidation measures after JP15. For instance, weak disease surveillance resulted in the persistence of unnoticed endemic rinderpest foci and in massive resurgence of the disease in the 1980s. Such lessons should be considered in future plans for global PPR eradication.

PPR has also specific features with respect to rinderpest. Because of its quick spread in immunologically naïve flocks, the virus can only maintain itself in large populations if new susceptible hosts (newborn, transhumant, or purchased animals) are available (Anderson, 1995). This is indeed the case for sheep and goat populations which have a quick turnover (at least 30%/year vs. 10% in cattle), and high mobility (local and regional trade, transhumance). PPR is also a seasonal disease occurring mostly during the cool, dry season in most endemic areas of Africa (Lancelot et al., 2002; Abubakar et al., 2009). To decrease PPR incidence and the risk of its (re-) introduction in other areas, initial steps of a PPR control program should start in highly infected regions, preferentially at the beginning of the dry season in Africa, and primarily in areas where intense animal contacts may occur: borders, large livestock markets, pastoral areas, and so on.

A better knowledge of sheep and goat population dynamics, herd management practices (including offtake rates) and animal movements (trade, transhumance) will be a critical condition for success. Another issue is the local management of the PPR control program by farmers, community based animal health workers, veterinary professionals and services, and research organizations. More than two decades ago, the management performances of many African countries were insufficient to support the needs for the development of livestock productions (Anteneh, 1989). Since then, progresses have been made, notably thanks to the PVS tools provided by OIE for the evaluation of veterinary services and the support for capacity building (Schneider, 2011). However, there are still some improvements to achieve in the governance of the surveillance systems and veterinary services in order to better coordinate the private and public actors of animal health management. In addition, veterinary services related to the surveillance and control of transboundary diseases must be considered as public goods and should consecutively remain under public management (Holden et al., 1996). They should receive attention and sustained resources and benefit from regional and international coordination. In this context, it is the role of international organizations for funding and capacity building to create synergies for the deployment of national human and financial resources to support animal health surveillance systems and veterinary services. The experience of rinderpest eradication has shown that a strong coordination of all actors at the international level is necessary to achieve the results. It is now widely recognized that the Global Framework for the progressive control of transboundary animal diseases (GF-TADs), which is a joint FAO/OIE initiative combining the strengths of both organizations, would be the best canal for this international coordination. GF-TADs is a facilitating mechanism which endeavors to empower regional alliances in the fight against transboundary animal diseases (TADs), to provide capacity building and to assist in establishing programs for the specific control of certain TADs based on regional priorities. As discussed and agreed in several regional and international conferences, GF-TADs is an ideal forum where PPR control strategies can be elaborated and decided in collaboration with national veterinary services. For Africa, the Inter-African Bureau of Animal Resources of the African Union (AU-IBAR) has also a major role to play, as a GF-TADs stakeholder, as well as a regional organization strongly involved in the coordination and implementation of PPR control.

Though a good PPR vaccine is available, the definition and implementation of a relevant vaccination strategy and vaccination monitoring might be tricky. Among others, the questions of the quick turnover in sheep and goat populations, accessibility of these populations for vaccination and monitoring, intensity of small ruminant trade and transhumances, have to be considered. In addition, spatial and temporal heterogeneities in sheep and goat population density and dynamics, as well as differences in breed receptivity and sensitivity to PPR virus, make it difficult to define a priori the most efficient vaccination strategy. The possible existence of wildlife reservoirs and the role of camels in PPR epidemiology make the situation even more complicated. Epidemiological modeling might be useful in these circumstances, to help deciding on the choice of the

best strategy including vaccination frequency, spatial setting of vaccination, and target species. However, realistic models taking population heterogeneities and seasonal changes into account still require further research efforts in epidemiology and mathematics. For instance, we lack models combining PPRV establishment (e.g., SEIR models: see Anderson and May (1991)) together with spread models (e.g., metapopulation models like in Ezanno and Lesnoff (2009)) to assess cost/benefit ratios of vaccination strategies that might be implemented in sub-regions of Africa.

Another major gap for the success of PPR control is the lack of economical assessment of control strategies, and even of PPR cost. Such information would be useful to help veterinary services in convincing governments and international organizations to support and fund PPR control. Economic models might be built on top of epidemiological model (and possibly sheep and goat population dynamics models), thus allowing ex-ante economic assessment of PPR control strategies. All these are actually the conditions of success for PPR control, and perhaps eradication. Though many lessons can be learnt from rinderpest eradication, appropriate design and practical implementation of PPR control strategies will have to find their own way.

Acknowledgements

The authors thank Thierry Lefrançois, Damien Meyer, Dominique Martinez and Denise Bastron for helpful reviewing and editing of this article.

References

Abraham, G., Sintayehu, A., Libeau, G., Albina, E., Roger, F., Laekemariam, Y., Abayneh, D., Awoke, K.M., 2005. Antibody seroprevalences against peste des petits ruminants (PPR) virus in camels, cattle, goats and sheep in Ethiopia. Prev. Vet. Med. 70, 51–57.

Abubakar, M., Ali, Q., Khan, H.A., 2008. Prevalence and mortality rate of peste des petits ruminant (PPR): possible association with abortion in goat. Trop. Anim. Health Prod. 40, 317–321.

Abubakar, M., Jamal, S.M., Arshed, M.J., Hussain, M., Ali, Q., 2009. Peste des petits ruminants virus (PPRV) infection; its association with species, seasonal variations and geography. Trop. Anim. Health Prod. 41, 1197–1202.

Abubakar, M., Arshed, M.J., Zahur, A.B., Ali, Q., Banyard, A.C., 2012. Natural infection with peste des petits ruminants virus: a pre and post vaccinal assessment following an outbreak scenario. Virus Res. 167, 43–47.

Adombi, C.M., Lelenta, M., Lamien, C.E., Shamaki, D., Koffi, Y.M., Traoré, A., Silber, R., Couacy-Hymann, E., Bodjo, S.C., Djaman, J.A., Luckins, A.G., Diallo, A., 2011. Monkey CV1 cell line expressing the sheep-goat SLAM protein: a highly sensitive cell line for the isolation of peste des petits ruminants virus from pathological specimens. J. Virol. Methods 173, 306–313.

Anderson, E.C., 1995. *Morbillivirus* infections in wildlife (in relation to their population biology and disease control in domestic animals). Vet. Microbiol. 44, 319–332.

Anderson, R., May, R., 1991. Infectious Diseases of Humans: Dynamics and Control. Oxford University Press, New York, 757 p.

Anteneh, A., 1989. Livestock and food production in Africa: challenge and opportunity for the national livestock and veterinary services. Rev. Sci. Tech. Off. Int. Epiz. 8, 591–606.

Ayari-Fakhfakh, E., Ghram, A., Bouattour, A., Larbi, I., Gribâa-Dridi, L., Kwiatek, O., Bouloy, M., Libeau, G., Albina, E., Cêtre-Sossah, C., 2011. First serological investigation of peste-des-petits-ruminants and Rift Valley fever in Tunisia. Vet. J. 187, 402–404.

Banyard, A.C., Parida, S., Batten, C., Oura, C., Kwiatek, O., Libeau, G., 2010. Global distribution of peste des petits ruminants virus and prospects for improved diagnosis and control. J. Gen. Virol. 91, 2885–2897.

Barrett, T., 1999. *Morbillivirus* infections, with special emphasis on morbilliviruses of carnivores. Vet. Microbiol. 69, 3–13.

Bailey, D., Chard, L.S., Dash, P., Barrett, T., Banyard, A.C., 2007. Reverse genetics for peste-des-petits-ruminants virus (PPRV): promoter and protein specificities. Virus Res. 126, 250–255.

Bao, J., Li, L., Wang, Z., Barrett, T., Suo, L., Zhao, W., Liu, Y., Liu, C., Li, J., 2008. Development of one-step real-time RT-PCR assay for detection and quantitation of peste des petits ruminants virus. J. Virol. Methods 148, 232–236.

Berhe, G., Minet, C., Le Goff, C., Barrett, T., Ngangnou, A., Grillet, C., Libeau, G., Fleming, M., Black, D.N., Diallo, A., 2003. Development of a dual recombinant vaccine to protect small ruminants against peste-des-petits-ruminants virus and capripoxvirus infections. J. Virol. 77, 1571–1577.

Couacy-Hymann, E., Bodjo, C., Danho, T., Libeau, G., Diallo, A., 2005. Surveillance of wildlife as a tool for monitoring rinderpest and peste des petits ruminants in West Africa. Rev. Sci. Tech. 24, 869–877.

Couacy-Hymann, E., Bodjo, C., Danho, T., Libeau, G., Diallo, A., 2007a. Evaluation of the virulence of some strains of peste-des-petits-ruminants virus (PPRV) in experimentally infected West African dwarf goats. Vet. J. 173, 178–183.

Couacy-Hymann, E., Bodjo, S.C., Danho, T., Koffi, M.Y., Libeau, G., Diallo, A., 2007b. Early detection of viral excretion from experimentally infected goats with peste-des-petits ruminants virus. Prev. Vet. Med. 78, 85–88.

De Nardi, M., Lamin Saleh, S.M., Batten, C., Oura, C., Di Nardo, A., Rossi, D., 2012. First evidence of peste des petits ruminants (PPR) virus circulation in Algeria (Sahrawi territories): outbreak investigation and virus lineage identification. Transbound. Emerg. Dis. 59, 214–222.

Diallo, A., 2003. Control of peste des petits ruminants: classical and new generation vaccines. Dev. Biol. (Basel) 114, 113–119.

Diallo, A., 2004 Peste des Petits Ruminants. In: Manual of Diagnostic Tests and Vaccines for Terrestrial Animals, OIE Terrestrial Manual, fifth ed. ISBN 92-9044-622-6.

Diallo, A., 2006. Control of peste des petits ruminants and poverty alleviation? J. Vet. Med. B Infect. Dis. Vet. Public Health 53, 11–13.

Diallo, A., Minet, C., Berhe, G., Le Goff, C., Black, D.N., Fleming, M., Barrett, T., Grillet, C., Libeau, G., 2002. Goat immune response to capripox vaccine expressing the hemagglutinin protein of peste des petits ruminants. Ann. N. Y. Acad. Sci. 969, 88–91.

Diallo, A., Minet, C., Le Goff, C., Berhe, G., Albina, E., Libeau, G., Barrett, T., 2007. The threat of peste des petits ruminants: progress in vaccine development for disease control. Vaccine 25, 5591–5597.

Diop, M., Sarr, J., Libeau, G., 2005. Evaluation of novel diagnostic tools for peste des petits ruminants virus in naturally infected goat herds. Epidemiol. Infect. 133, 711–717.

El Hag Ali, B., Taylor, W.P., 1984. Isolation of peste des petits ruminants virus from Sudan. Res. vet. Sci. 36, 1–4.

Eligulashvili, R., Perl, S., Stram, Y., Friedgut, O., Scheichat, N., Samina, I., Trainin, Z., 1999. Immunohistochemical detection of peste des petits ruminants viral antigen in formalin-fixed, paraffin-embedded tissues from cases of naturally occurring infection. J. Vet. Diagn. Invest. 11, 286–288.

Ezanno, P., Lesnoff, M., 2009. A metapopulation model for the spread and persistence of contagious bovine pleuropneumonia (CBPP) in African sedentary mixed crop-livestock systems. J. Theor. Biol. 256, 493–503.

Ezeibe, M.C.O., Okoroafor, O.N., Ngene, A.A., Eze, J.I., Eze, I.C., Ugonabo, J.A.C., 2008. Persistent detection of peste de petits ruminants antigen in the faeces of recovered goats. Trop. Anim. Health Prod. 40, 517–519.

Gargadennec, L., Lalanne, A., 1942. La peste des petits ruminants. Bull. Serv. Zoo AOF 5, 16–21.

Gibbs, E.P., Taylor, W.P., Lawman, M.J., Bryant, J., 1979. Classification of peste des petits ruminants virus as the fourth member of the genus *Morbillivirus*. Intervirology 11, 268–274.

Govindarajan, R., Koteeswaran, A., Venugopalan, A.T., Shyam, G., Shaouna, S., Shaila, M.S., Ramachandran, S., 1997. Isolation of pestes des petits ruminants virus from an outbreak in Indian buffalo (*Bubalus bubalis*). Vet. Rec. 141, 573–574.

Hammouchi, M., Loutfi, C., Sebbar, G., Touil, N., Chaffai, N., Batten, C., Harif, B., Oura, C., El Harrak, M., 2012. Experimental infection of alpine goats with a Moroccan strain of peste des petits ruminants virus (PPRV). Vet. Microbiol. http://dx.doi.org/10.1016/j.vetmic.2012.04.043.

Holden, S., Ashley, S., Bazeley, P., 1996. Improving the delivery of animal health services in developing countries: a literature review. A report to the overseas development administration of the United Kingdom. In: Gordon, S., Hebbert, A., (Eds.), Livestock in Development, pp. 1–96. ISBN 095280610X.

Hu, Q., Chen, W., Huang, K., Baron, M.D., Bu, Z., 2012. Rescue of recombinant peste des petits ruminants virus: creation of a GFP-expressing

virus and application in rapid virus neutralization test. Vet. Res. 43, 48.
Ismail, I.M., House, J., 1990. Evidence of identification of peste des petits ruminants from goats in Egypt. Arch. Exp. Vet. Med. 44, 471–474.
Jones, L., Giavedoni, L., Saliki, J.T., Brown, C., Mebus, C., Yilma, T., 1993. Protection of goats against peste des petits ruminants with a vaccinia virus double recombinant expressing the F and H genes of rinderpest virus. Vaccine 11, 961–964.
Keita, D., Servan de Almeida, R., Libeau, G., Albina, E., 2008. Identification and mapping of a region on the mRNA of *Morbillivirus* nucleoprotein susceptible to RNA interference. Antivir. Res. 80, 158–167.
Kerdiles, Y.M., Cherif, B., Marie, J.C., Tremillon, N., Blanquier, B., Libeau, G., Diallo, A., Wild, T.F., Villiers, M.B., Horvat, B., 2006. Immunomodulatory properties of *morbillivirus* nucleoproteins. Viral Immunol. 19, 324–334.
Khalafalla, A.I., Saeed, I.K., Ali, Y.H., Abdurrahman, M.B., Kwiatek, O., Libeau, G., Obeida, A.A., Abbas, Z., 2010. An outbreak of peste des petits ruminants (PPR) in camels in the Sudan. Acta Trop. 116, 161–165.
Kinne, J., Kreutzer, R., Kreutzer, M., Wernery, U., Wohlsein, P., 2010. Peste des petits ruminants in Arabian wildlife. Epidemiol. Infect. 138, 1211–1214.
Kulkarni, D.D., Bhikane, M.S., Shaila, M.S., Varalakshmi, P., Apte, M.P., Narladkar, B.W., 1996. Peste des petits ruminants in goats India. Vet. Rec. 138, 187–188.
Kwiatek, O., Keita, D., Gil, P., Fernández-Pinero, J., Jimenez Clavero, M.A., Albina, E., Libeau, G., 2010. Quantitative one-step real-time RT-PCR for the fast detection of the four genotypes of PPRV. J. Virol. Methods 165, 168–177.
Kwiatek, O., Ali, Y.H., Saeed, I.K., Khalafalla, A.I., Mohamed, O.I., Obeida, A.A., Abdelrahman, M.B., Osman, H.M., Taha, K.M., Abbas, Z., El Harrak, M., Lhor, Y., Diallo, A., Lancelot, R., Albina, E., Libeau, G., 2011. Asian lineage of peste des petits ruminants virus, Africa. Emerg. Infect. Dis. 17, 1223–1231.
Lancelot, R., Lesnoff, M., McDermott, J.J., 2002. Use of Akaike information criteria for model selection and inference. An application to assess prevention of gastrointestinal parasitism and respiratory mortality of Guinean goats in Kolda, Senegal. Prev. Vet. Med. 55, 217–240.
Lefèvre, P.C., Diallo, A., 1990. Peste des petits ruminants. Rev. Sci. Tech. 9, 935–981.
Libeau, G., Diallo, A., Colas, F., Guerre, L., 1994. Rapid differential diagnosis of rinderpest and peste des petits ruminants using an immunocapture ELISA. Vet. Rec. 134, 300–304.
Libeau, G., Préhaud, C., Lancelot, R., Colas, F., Guerre, L., Bishop, D.H., Diallo, A., 1995. Development of a competitive ELISA for detecting antibodies to the peste des petits ruminants virus using a recombinant nucleoprotein. Res. Vet. Sci. 58, 50–55.
Maillard, J.C., Van, K.P., Nguyen, T., Van, T.N., Berthouly, C., Libeau, G., Kwiatek, O., 2008. Examples of probable host-pathogen co-adaptation/co-evolution in isolated farmed animal populations in the mountainous regions of North Vietnam. Ann. N.Y. Acad. Sci. 1149, 259–262.
Minet, C., Yami, M., Egzabhier, B., Gil, P., Tangy, F., Bremont, M., Libeau, G., Diallo, A., Albina, E., 2009. Sequence analysis of the large (L) polymerase gene and trailer of the peste des petits ruminants virus vaccine strain Nigeria 75/1: expression and use of the L protein in reverse genetics. Virus Res. 145, 9–17.
Özkul, A., Akca, Y., Alkan, F., Barrett, T., Karaoglu, T., Dagalp, S.B., Anderson, J., Yesilbag, K., Cokcaliskan, C., Gencay, A., Burgu, I., 2002. Distribution, and host range of peste des petits ruminants virus, Turkey. Emerg. Infect. Dis. 8, 709–712.
Roeder, P., Obi, T. 1999. Recognizing peste des petits ruminants. A field manual. 28 p.
Roger, F., Diallo, A., Yigezu, L.M., Hurard, C., Libeau, G., Mebratu, G.Y., Faye, B., 2000. Investigation of a new pathological condition of camels in Ethiopia. J. Camel Pract. Res. 2, 163–166.
Schneider, H., 2011. Good governance of national veterinary services. Rev. Sci. Tech. 30, 325–338.
Servan de Almeida, R., Keita, D., Libeau, G., Albina, E., 2007. Control of ruminant *morbillivirus* replication by small interfering RNA. J. Gen. Virol. 88, 2307–882311.
Shaila, M.S., Shamaki, D., Forsyth, M.A., Diallo, A., Goatley, L., Kitching, R.P., Barrett, T., 1996. Geographic distribution and epidemiology of peste des petits ruminants virus. Virus Res. 43, 149–153.
Silva, A.C., Carrondo, M.J., Alves, P.M., 2011. Strategies for improved stability of peste des petits ruminants vaccine. Vaccine 29, 4983–4991.
Swai, E.S., Kapaga, A., Kivaria, F., Tinuga, D., Joshua, G., Sanka, P., 2009. Prevalence and distribution of Peste des petits ruminants virus antibodies in various districts of Tanzania. Vet. Res. Commun. 33, 927–936.
Taylor, W.P., Al Busaidy, S., Barrett, T., 1990. The epidemiology of peste des petits ruminants in the sultanate of Oman. Vet. Microbiol. 22, 341–352.
Worrall, E.E., Litamoi, J.K., Seck, B.M., Ayelet, G., 2000. Xerovac: an ultra rapid method for the dehydration and preservation of live attenuated rinderpest and peste des petits ruminants vaccines. Vaccine 19, 834–839.