Supplementary Appendix

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This appendix has been provided by the authors to give readers additional information about the work.

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S1 Supplemental Materials and Methods

S1.1 Author contributions

All the authors supported the investigations described herein, decided to publish the manuscript, and critically reviewed the manuscript. The first five authors collected and analyzed the data. A subgroup of the authors initially drafted the manuscript. The last author vouches for the accuracy and completeness of the data. There was no commercial funding for this study. Details of author contributions can be found below.

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S1.2 Sample inactivation and RNA extraction for RT-qPCR

Different types of specimens (blood, serum, bronchiolar lavage, nasopharyngeal and oropharyngeal swabs, saliva, urine, semen, conjunctiva) were collected from fatal cases and survivors. Prior to RNA extraction, specimens were inactivated in the highest possible biocontainment laboratory: biosafety level 4 (BSL4) at CDC and BSL3 at CENETROP following CDC's biosafety guidelines (https://www.cdc.gov/safelabs/resources-tools/biosafety-resources-and-tools.html?CDC_AA_refVal=https%3A%2F%2Fwww.cdc.gov%2Fsafelabs%2Fresources-tools.html). A testing algorithm was created that proposes testing patients with suspected VHF for New World arenaviruses first, using enhanced biosafety precautions, before testing for other pathogens with lower biosafety requirements (Figures S2a-S2b).

At CDC, samples were inactivated using 100 µL of the corresponding specimen mixed with 400 µL of MagMax lysis buffer (ThermoFisher Scientific, Waltham, MA, USA) followed by 10 min incubation at room temperature. Inactivated samples were then transferred from BSL4 to a BSL2 laboratory for RNA extraction. RNA was extracted with the MagMAX Pathogen RNA/DNA Kit (Applied Biosystems, Thermo Fisher Sci, Waltham, MA USA) using the BeadRetriever platform (Applied Biosystems, Thermo Fisher Sci, Waltham, MA USA) and following the manufacturer's instructions. In Bolivia, specimens were inactivated in the BSL3 laboratory at CENTROP, with enhanced personal protective equipment for aerosol precautions, like N95 masks or powered air purifying respirators (PAPRs). For RNA extraction, 140 µL of each specimen was placed in 560 µL of lysis buffer (AVL); after 10 min, 560 µL of ethanol were added and 10 min incubation completed the inactivation. Tubes were sealed and decontaminated prior to transfer from BSL3 to BSL2 where RNA extraction was performed with the QIAamp Viral RNA Mini Kit (Qiagen, Germantown, MD, USA) following manufacturer's instructions.

S1.3 CHAPV Bolivia 2019 real-time and conventional RT-PCR diagnostic assays

Quantitative RT-PCR (RT-qPCR) and conventional RT-PCR assays targeting S segment and L segment were developed using Geneious Prime® 2019.2.3 on partial Chapare Bolivia 2019 sequences generated from Patient S1-3. The following conditions were used for the RT-qPCR assays: 5µL of extracted RNA was amplified with either S or L RT-qPCR assays, using Luna Universal Probe One-Step RT-qPCR Kit (New England Biolabs, Ipswich, MA, USA) on the CFX96 Touch Real-Time PCR Detection System (BioRad, Hercules, CA, USA). Alternatively, RT-qPCR assays were also performed using SuperScript™ III Platinum™ One-Step qRT-PCR Kit (Invitrogen, Thermo Fisher Sci, Waltham, MA USA) on the 7500 Real-Time PCR System (Applied Biosystems, Thermo Fisher Sci, Waltham, MA USA). For conventional RT-PCR, 5µL of extracted RNA was amplified using qScript XLT 1-Step RT-PCR Kit (Quantabio) on the C1000 Touch™ Thermal Cycler (BioRad, Hercules, CA, USA)). PCR products were visualized using 2% agarose E-gels (Invitrogen, Thermo Fisher Sci, Waltham, MA USA) and PCR products were sequenced using Sanger sequencing following manufacturer protocols (BigDye™ Terminator v3.1 Cycle Sequencing Kit, 4337457; Applied Biosystems). To evaluate nucleic acid extraction and specimen quality, 5µL of RNA extracted from samples were amplified using Human B2M (Beta-2-Microglobulin) Endogenous Control (Applied Biosystems, Thermo Fisher Sci, Waltham, MA USA). Conventional RT-PCR assays were done at CDC in a subset of initial samples to confirm the amplification of CHAPV by sequencing the amplified RT-PCR product. In August 2019, in collaboration with the Pan-American Health Organization (PAHO), CDC provided the S and L RT-qPCR assays to CENETROP to improve the available diagnostic testing in Bolivia. Those assays have since been used to test human and rodent specimens for CHAPV RNA presence.

The S and L Bol NWA 2019 primers and probes used for the RT-qPCR assays specifically detect the CHAPV 2019 strain but not the older lineage (2003) of the virus. Primer sequences and assay efficiency data are presented below.

CHAPV RT-qPCR Primers and Probes

Name	Sequence (5′–3′)	Length (bases)	PCR product size
S Bol NWA 2019			
Fwd_S492_rtS1	AGTGGATTTTGAGAGCCCTG	20	130 bp
Rev_S621_rtS1	AAATCATGAGACCCTGAGCTG	21	
Prb_S581_rtS1	/56-FAM/TG CGT GTT T/ZEN/C GTT GAC AGT TGA ACT C/3IABkFQ/	26	
L Bol NWA 2019			
Fwd_L3295_rtS1	TTCCATGAGCCCAAGACTTC	20	145 bp
Rev_L3439_rtS1	CGAACCCAAGAGTAGAGCTAAG	22	
Prb_L3371_rtS1	/56-FAM/AA GTT GTC G/ZEN/A AGT GCC TTT TAA CGT TGC /3IABkFQ/	28	

Assay Efficiency

Assay Name	Slope	Efficiency (%)%
S Bol NWA 2019	-3.32	100.1
L Bol NWA 2019	-3.51	92.9

S1.4 Rodent trapping, sample collection, and testing

For the rodent collection work, the Bolivian Ministry of health deployed a team to the municipalities of Caranavi and Guanay, department of north La Paz, where the index case and patient S1-3 lived and worked. The mission started on July 5, 2019 and rodents were captured on July 7, 8 and 9, 2019. GPS coordinates of successful trap sites are provided in Supplemental Results 2.2.

The animals were collected using Sherman live traps (H. B. Sherman Trap Inc., Tallahassee, Florida) baited with oats, peanuts, and vanilla extract. Traps were placed in and around the residence and working crop fields as described above. Captured rodents were weighed,

measured, and humanely euthanized prior to necropsy. Whole blood, spleen, kidney, feces and urine were collected. The RNA was extracted from those samples in Bolivia and the samples were tested by CHAPV RT-qPCR in CENETROP. Extracted RNA from specimens from 32 individual rodents were sent to CDC for NGS. Rodents were identified, initially to genus based on morphological characteristics, and later to species using mitochondrial cytochrome-*b* DNA sequencing.

S1.5 RNA extraction for next-generation sequencing at CDC

For next-generation sequencing (NGS), RNA was extracted from clinical material or virus isolates (100–250 µL of specimen) under BSL4 conditions using Tripure (Roche), followed by phase separation with 1-bromo-3-chloropane (Sigma-Aldrich, St. Louis, MO, USA); RNA was extracted from the upper phase using Clean and Concentrate-25 columns (Zymo, Irvine, CA, USA). Nucleic acids were treated with RNase-free DNase (Roche) and libraries were prepared with NEBNext Ultra II Directional library preparation kit (New England Biolabs). DNA libraries combined with low multiplexing (2–5 samples) and sequenced on an Illumina MiSeq (300 cycles PE v2 kit) or MiniSeq (300 cycles High output).

S1.6 Serological testing for Junin, Machupo, and Chapare viruses

In absence of a CHAPV-specific serological assay at the time of investigation in 2019, the Argentinian National Institute for Viral Hemorrhagic Diseases provided closely related Junin virus (JUNV) enzyme-linked immunosorbent assay (ELISA) to CENETROP, Bolivia. Briefly, the JUNV antigen was a virus cell lysate of Vero-C76 cells infected with JUNV strain XJ Clon 3.[1] Half of the plate was coated with 100 µL/well of the infected cell lysate diluted in phosphate buffered saline (PBS), pH 7.4, and the other half was coated with the normal control antigen (from virus diluent mock-infected cells) and kept overnight at 4°C. 100 µL test sera were added

to duplicate wells (one containing JUNV antigen and the other control antigen) and titrated at serial 4-fold dilutions, beginning at 1:100. After 1 h incubation at 37°C, plates were washed and incubated with anti-human IgG horseradish peroxidase conjugate (Accurate Chemical, Westbury, NY, USA). After another hour, plates were washed and developed with 100 µL of H_2O_2-ABTS substrate system (KPL, Sera Care, Milford, MA, USA). Optical densities were measured at 405 and 450 nm. O.D. corrected values (after subtracting antigen-negative O.D.) greater than 0.2 were considered positive. Samples with titers ≥ 1/400 were considered positive. Immediately following the 2019 investigation, blood and serum samples from the 2019 CHHF confirmed cases were shipped to and tested at CDC using in-house IgM and IgG assays for Machupo virus (MACV), which is also closely related to CHAPV. Most recently, all available blood and serum samples were tested at CDC using novel in-house IgM and IgG assays for CHAPV. Antigen preparations were performed at Viral Special Pathogens Branch BSL4 laboratory using MACV Carvallo strain (NC_005078, NC_005079) or the recently isolated CHAPV 2019 strain. The methods for in-house IgM and IgG ELISAs were previously described for Lassa virus (another arenavirus) and Ebola virus (a filovirus).[2,3] Prior to testing, sera or blood were inactivated in CDC BSL4 laboratory, gamma-irradiated with 2×10^6 rads, and tested in a BSL2 laboratory by serial dilutions as previously described. To detect human IgG, anti-human IgG horseradish peroxidase conjugate (Accurate Chemical, Westbury, NY, USA) and H_2O_2-ABTS substrate system (KPL, Sera Care, Milford, MA, USA) were used. For IgG, an adjusted OD (values between those of positive and negative antigen) of >0.2 was required for each dilution to be considered positive and for a titer to be assigned accordingly. Sera were considered positive if the titer was ≥1:400 and the sum of the adjusted OD was ≥0.95. In IgM capture ELISA, anti-human IgM antibody (KPL, Sera Care, Milford, MA, USA) was used to capture IgM antibodies from the serially diluted samples, followed by the MACV, CHAPV or mock

antigens. Captured antigens were detected using hyperimmune mouse ascitic fluid (HMAF) against MACV or CHAPV available at CDC, followed by a goat anti-mouse horseradish peroxidase conjugate (Thermo Fisher Sci, Waltham, MA USA) and H_2O_2-ABTS substrate system (KPL, Sera Care, Milford, MA, USA). For IgM, adjusted OD of >0.1 was required for each dilution to be considered positive and for a titer to be assigned accordingly. Sera were considered positive if the titer was ≥1:400 and the sum of the adjusted OD was ≥0.45.

S1.7 Virus isolation, immunofluorescence, and electron microscopy

Clinical material from acute and clinically recovered patients (80–100 µL of blood, serum, semen, bronchiolar lavage, urine) was added to sub-confluent (approximately 85%) Vero-E6 cells under BSL4 conditions and allowed to adsorb for 1 h with rocking. After 1 h, fresh Eagle's minimal essential medium (EMEM, Thermo Fisher Sci, Waltham, MA USA) with 2% FBS and penicillin/streptomycin was added to cells and infection proceeded for up to14 days. Cells were checked every 2–3 days for evidence of cytopathic effect (CPE) by comparing to mock (uninfected) cells, and 100 µL of media was collected for CHAPV S and L segment RT-qPCR. Spot slides were prepared from cells on days 7 and 14 and when evidence of CPE was observed. Spot slides were prepared by scraping a portion of the cell flask, centrifuging cells in borate saline at 1200 × g for 10 min, resuspending cell pellets in 0.5 mL of borate saline, and spotting cells onto slides. Slides dried overnight in a biosafety hood, were fixed in 100% acetone, and irradiated with 2×10^6 rads of gamma irradiation prior to staining in the BSL2 laboratory. To confirm evidence of CHAPV infection, cells were stained with anti-CHAPV HMAF diluted 1:100, incubated in a humid chamber for 30 min; after washing 3 times with PBS, anti-mouse FITC (1:40) was used to reveal HMAF binding. CDC in-house-made IFA slides with MACV- and Sabia virus-infected Vero-E6 cells were used as staining controls. Anti-MACV HMAF was also used for initial isolates with negative results; only anti-CHAPV antibodies yielded a

fluorescent signal. For subsequent experiments virus isolation was also monitored by a decrease in the Ct value using the CHAPV RT-qPCR, and by IFA with anti-CHAPV HMAF. To perform electron microscopy, some virus isolates were selected for a second passage in Vero-E6 cells. Seven days post infection, cells were harvested, fixed in a solution of 10% formalin, gamma-irradiated (5×10^6 rads), and transferred to Pathology Branch (CDC, Atlanta, GA, USA) where electron micrographs were obtained showing the typical structure of arenaviruses in the CHAPV isolates

S1.8 References for supplemental laboratory materials and methods

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2. Bausch DG, Rollin PE, Demby AH, et al. Diagnosis and clinical virology of Lassa fever as evaluated by enzyme-linked immunosorbent assay, indirect fluorescent-antibody test, and virus isolation. J Clin Microbiol 2000;38(7):2670-7. DOI: 10.1128/JCM.38.7.2670-2677.2000.
3. Ksiazek TG, West CP, Rollin PE, Jahrling PB, Peters CJ. ELISA for the detection of antibodies to Ebola viruses. J Infect Dis 1999;179 Suppl 1:S192-8. DOI: 10.1086/514313.

S2 Supplemental Results

S2.1 _Additional clinical information of initial 2019 Chapare hemorrhagic fever cases

S2.1.1 Patient S1-1 (fatal)

Patient was a 65-year-old male with no known underlying medical conditions or use of medications. Patient was an agricultural worker in Caranavi Province and in the weeks preceding symptom onset, patient was farming rice in Siliamo, Guanay Municipality. Available clinical information was limited and laboratory values during admission were unavailable for review. Patient remains the probable index case of the 2019 outbreak of Chapare hemorrhagic fever (CHHF).

On April 24, 2019, the patient developed headache, abdominal pain, and nausea. Four days post-symptom onset (April 28), the patient presented to a local hospital (Hospital A) with complaints of persistent symptoms and onset of fever and lumbar pain. The patient was discharged the same day with a suspected diagnosis of dengue. On day 7 post-symptom onset (May 1), the patient's symptoms worsened to include retro-orbital pain and diarrhea, and he presented to a nearby local hospital (Hospital B); he was discharged the same day with suspected dengue. On day 13 (May 7), he presented again to the same hospital and was admitted with severe dehydration, abdominal pain, and gingival hemorrhage, and was treated for suspect dengue with warning signs. The patient remained hospitalized for the next 5 days with worsening clinical signs and died 18 days post-symptom onset (May 12). The suspected cause of his illness remained severe dengue with warning signs and no specimens were collected from the patient for diagnostic testing.

On May 11, the night before the patient died, a medical intern and a family member of the patient had direct contact with him in a hospital in Caranavi; both were later identified as confirmed cases of CHHF (patients S1-2 and S1-3, respectively).

S2.1.2 Patient S1-2 (fatal)

Patient was a 25-year-old female with no history of underlying medical conditions or medication use; patient underwent tonsillectomy 1 year prior to illness onset. Patient was a medical intern at a hospital in Caranavi Municipality. On May 11, she had close contact with the blood and bodily fluids of the patient that would later be identified as the probable index case (patient S1-1) of the initial 2019 CHHF outbreak.

Nine days post exposure (May 20), the patient developed fever, headache, myalgia, arthralgia, and general malaise and called in sick from work. Over the next 3 days, the patient remained symptomatic; on May 23, laboratory testing was notable for anemia (hematocrit 38%, reference range [RR]: 43–58%; hemoglobin 12.5 mg/dL, RR: 14.0–19.8 mg/dL) and leukopenia (WBC 3.3 x 10^9/L; RR: 4.5–11.0 x 10^9/L), with normal platelet and transaminase levels.

On day 7 post-symptom onset (May 27), she was admitted to a local hospital (Hospital A) with fever, abdominal pain, nausea, vomiting, myalgia, and gingival hemorrhage. On physical exam, patient was hemodynamically stable with subjective fever (37°C, with paracetamol use), painful abdomen on palpation, gingival and vaginal (menstruation) hemorrhage, and normal Glasgow coma scale (15/15). Laboratory testing on admission was notable for worsening anemia (hemoglobin 11.8 mg/dL), leukopenia (WBC 2.4 x 10^9/L), and elevated transaminases (AST 1048 U/L, RR: 47–160 U/L; ALT 471 U/L, RR: 11.4–28.8 U/L). The patient was suspected to have dengue with warning signs (critical phase) and received intravenous (IV) fluid resuscitation (Ringer's lactate solution 2 L2L/24 h), omeprazole, and metoclopramide. The morning of day 8 (May 28), the patient's clinical status declined with new onset of lumbar pain, diarrhea, and continued gingival hemorrhage. The same day, labs revealed worsening transaminases; IV fluid therapy continued. In the afternoon, the patient showed evidence of disseminated intravascular

coagulopathy with PT 20 s (RR: 10–13 s-13s), PTTa 90s (RR: 30–40 s-40s); 3 units of fresh frozen plasma were administered.

On day 9 (May 29), the patient continued to be febrile with gingival hemorrhage and myalgias; echography revealed evidence of hepatic steatosis; vomiting and diarrhea improved. In response to the bleeding, she was transfused with packed red blood cells, crystalloid solution, cryoprecipitates, and fresh frozen plasma. On day 10 (May 30), the patient experienced one generalized tonic-clonic seizure, for which she received diazepam, and was put on mechanical ventilation for airway protection. Labs at the time revealed worsened thrombocytopenia (platelets 55 x 10^9/L, range; RR: 150–450 x 10^9/L), leukopenia (WBC 2.1 x 10^9/L), transaminases (AST 1713 U/L; ALT 450 U/L), and coagulopathy (PT: 25 s; PTTa: 120 s); dengue IgM and NS1 PCR were negative. Over the next 3 days, the patient remained febrile with gingival hemorrhage and remained under mechanical ventilation.

On day 13 (June 2), the patient was transferred by ambulance to the intensive care unit (ICU) of a tertiary care hospital in La Paz (Hospital B) due to her worsening clinical status. During ambulance transfer, a physician had contact with her blood and body fluids and later became identified as patient S1-4. On admission to Hospital B, the patient underwent an emergency upper gastrointestinal (GI) endoscopy due to worsening anemia refractory to transfusions and continued melena; the gastroenterologist performing the endoscopy would later developed symptoms and be identified as patient S1-5. The endoscopy revealed presence of acute erosive gastropathy with diffuse bleeding; the patient subsequently developed hemorrhagic shock and died on day 15 (June 4). The cause of the patient's illness was suspected to be dengue, and it was not until the subsequent cases were identified after her death that an arenavirus infection was suspected in this patient.

S2.1.3 Patient S1-3 (survivor)

Patient was a 25-year-old male no significant medical history, reported underlying conditions, or medications. Patient was an agricultural worker in Caranavi Province and family member of the probable index case (patient S1-1) of the initial 2019 CHHF outbreak. Prior to symptom onset, the patient was farming rice with S1-1 in Siliamo, Guanay Municipality. The patient also lived in the same home as S1-1 while S1-1 was symptomatic prior to hospitalization, and on May 11, S1-3 had direct contact with S1-1 in a hospital in Caranavi the night before S1-1 died.

Nineteen days after final exposure to S1-1 (May 30), the patient developed fever, headache, retro-orbital pain, myalgia, arthralgia, and general malaise. On day 3 post-symptom onset (June 2), his symptoms progressed to include vomiting, abdominal pain, gingival hemorrhage, irritability, psychomotor excitation, disorientation, dehydration, and oliguria.

On day 7 (June 6), patient presented to Hospital A with generalized tonic-clonic seizure, fever (39°C), and difficulty breathing. On admission, he was hypoxic and laboratory testing revealed leukopenia (WBC 2.3 x 10^9/L, RR: 4.5–11.0 x 10^9/L), thrombocytopenia (platelets 105 x 10^9/L, RR: 150–450 x 10^9/L), elevated transaminases (AST 1796 U/L, RR: 47–160 U/L; ALT 1197 U/L, RR: 11.4–28.8 U/L) and coagulopathy (PTTa 65 s, RR: 30–40 s). Intravenous (IV) fluid therapy, anticonvulsants, and sedation were administered, and he was transfused with 3 units of fresh frozen plasma and 7 units of cryoprecipitates. He was transferred the same day to Hospital B with a suspected diagnosis of dengue with warning signs and probable viral encephalitis.

On admission to Hospital B, patient S1-3 was hemodynamically unstable with active hemorrhaging including gingival hemorrhage, epistaxis, gastrointestinal hemorrhage, melena; Glasgow coma scale 8/15 with left hemiparesis; and was mechanically ventilated for airway protection. He was transferred on day 8 (June 7) to the ICU of a nearby private tertiary care hospital (Hospital C) because no ICU beds were available in Hospital B. On arrival to the ICU,

he was febrile and hemodynamically unstable with deteriorating neurologic status and noted to have multiorgan failure with petechiae/ecchymoses, injection-site hemorrhage, hematuria, and active upper gastrointestinal bleeding complicated by hemorrhagic shock and right-sided intracerebral hemorrhage with edema (Figure S3). Labs revealed anemia (hemoglobin 7.3 g/dL, RR: 14–19.8 g/dL) and elevated liver function tests (total bilirubin 3.2 mg/dL, RR: 0.2–1.2 mg/dL; AST 1068 U/L, RR: 47–160 U/L; ALT 712.9 U/L, RR: 11.4–28.8 U/L). He was treated with broad-spectrum antibiotics, antifibrinolytics, calcium gluconate, anticonvulsants, gastric protector, corticosteroids, vasopressors, sedation, and analgesia, and transfused with fresh frozen plasma, platelets, and packed red blood cells.

Over the next 2 days, severe hypoxia and shock continued, but transaminases, bilirubin, thrombocytopenia, and hemorrhagic signs improved. On day 11 (June 10), he progressed to hypoxemic respiratory failure likely secondary to alveolar hemorrhage and ventilator-associated pneumonia (Figure S4, S5). On day 12 (June 11), diagnostic testing for dengue, chikungunya, Zika, leptospirosis, yellow fever, hantavirus, and blood and urine cultures were negative. He remained on mechanical ventilation and antibiotic therapy until day 19 (June 18). Hypoxia and laboratory abnormalities improved; he was extubated on day 22 (June 21) and discharged from ICU on day 25 (June 24). He was discharged from the hospital on day 31 post-symptom onset (June 30) and experienced prolonged neurological deficits including disorientation and confusion that persisted for several months after discharge. See Table S3a for daily account of the patient's clinical evolution and treatment during the first 3 weeks of illness.

S2.1.4 Patient S1-4 (survivor)

Patient was a 48-year-old male with no history of underlying medical conditions or use of medications, and no prior history of travel to tropical areas within or outside Bolivia. On June 2, 2019, the patient, a physician, was involved in the ambulance transfer of S1-2. During the trip,

S1-4 had to aspirate bloody secretions from S1-2's endotracheal tube and oral cavity. He wore gloves and a surgical mask, but a large amount of blood contaminated his clothes.

Sixteen days post exposure (June 18), the patient developed general malaise, fatigue, odynophagia, arthralgia, and myalgia. Over the next 72 h, symptoms progressed to include muscle weakness in the lower extremities and inability to walk. On day 6 post-symptom onset (June 24), he was hospitalized for <24 h in Hospital A. On admission, laboratory testing was notable for: leukopenia (WBC 2.9 x 10^9/L, RR: 5.0–10.0 x 10^9/L); thrombocytopenia (platelets 106 x 10^9/L, RR: 150–400 x 10^9/L), and slightly elevated creatinine (1.7 mg/mL, RR: 0.7–1.5 mg/mL).

In the next 24 h, the patient's clinical status worsened, and he developed generalized muscle weakness, predominantly in the lower extremities. On day 8 (June 26), he was transferred to Hospital B for elevated level of care due to his worsening clinical picture.

In the first 48 h of hospitalization at Hospital B, the patient developed worsening paraparesis (muscle strength 4/5), with intact reflexes and sensitivity and without evidence of meningeal irritation. Laboratory testing was notable for worsened leukopenia (WBC 2.1 x 10^9/L) and thrombocytopenia (platelets 96 x 10^9/L), rising creatinine (3 mg/dL), and worsening liver function tests (total bilirubin 2.4 mg/dL, RR: <1.2 mg/dL; AST 229 U/L, RR: 47–160 U/L; ALT 291 U/L, RR: 11.4–28.8 U/L; ALP 345 U/L, RR: 44–147 U/L). Proteinuria and leukocyturia (30–60 per field) were also noted on urinalysis.

On day 9 (June 27), 24 h after admission to hospital B, a lumbar puncture was performed showing 0 nucleated cells, protein 37 mg/dL, and glucose 90 mg/dL. A nerve conduction velocity test and electromyography revealed a mild myopathic pattern without evidence of polyradiculopathy. Due to suspicion of viral myopathy vs. Guillian Barre syndrome, treatment

with intravenous immunoglobulin at a dose of 2 g/kg infused for 5 days was started without corticosteroids.

On day 10 (June 28), the patient's epidemiologic link to an unknown VHF related to contact with a patient transferred from Caranavi (patient S1-2) was established. The same day, the patient developed gingival hemorrhage, melena, disorientation, and confusion, and was transferred to the ICU of Hospital B. His laboratory findings were notable for elevated creatine phosphokinase (1390 U/L, RR: 55–170 U/L), leukopenia (2.1 x 10^9/L), fibrinogen (375 mg/dL, RR: 200–400 mg/dL), creatinine (1.6 mg/dL), prothrombin time (13 s), activated partial thromboplastin time (38.1 s INR 1, and worsening liver function tests (total bilirubin 3.88 mg/dL; RR: 0.2–1.2 mg/dL;; ALT 595 U/L, RR: 11.4–28.8 U/L; lactate dehydrogenase 3307 U/L, RR: 140–280 U/L).. He was managed with broad-spectrum antibiotics (meropenem) and acyclovir in addition to intense transfusion support (fresh frozen plasma and platelet concentrates), at the time, no hemodynamic or respiratory support was required.

On day 13 (July 1), the possibility of arenavirus infection was suggested, and oral ribavirin was started for 5 days, in addition to a unit of anti-MACV convalescent plasma. In the following days, mucosal and gastrointestinal bleeding persisted, and transfusion support continued. The patient had evidence of worsening liver injury with progressive elevation of bilirubin and transaminases, in addition to worsening leukopenia and thrombocytopenia.

On day 18 (July 6), the patient developed hypoxemic respiratory failure secondary to transfusion-related acute lung injury, requiring mechanical ventilation. In the next 48 h, he developed worsening acute kidney injury and started renal replacement therapy with intermittent hemodialysis on day 22 (July 10), which was maintained for 2 sessions. Broad-spectrum antibiotics (meropenem and cotrimoxazole) were administered for suspected hospital-acquired

pneumonia due to the presence of *Burkholderia cepacia* in culture of tracheal secretions. In the following weeks, mechanical ventilation was maintained; patient was tracheostomized on day 34 (July 22). The patient developed mechanical ventilation-associated pneumonia which was managed with piperacillin/tazobactam, and probable candidemia, which was treated with a course of anidulafungin.

On day 55 (August 12), mechanical ventilation was withdrawn, and the tracheostomy tube was removed on August 15. On day 66 (August 23), patient was discharged from the ICU to a general private hospital room. In the following weeks, he remained hospitalized with no new nosocomial infections occurred; however, his stay was prolonged out of precaution due to persistent CHAPV RNA detection in blood samples and bodily fluids. Muscular and neurological deficits related to prolonged critical care persisted, including paresis and episodes of delirium, improving over the following weeks of hospitalization. On day 161 post-symptom onset (November 26), patient was discharged from Hospital B for follow-up by outpatient consultation. The patient continued to have difficulty walking throughout his recovery and required physical therapy to regain function of his lower limbs. See Table S3b for daily account of patient's clinical evolution and treatment during the first 2 weeks of illness.

S2.1.5 Patient S1-5 (fatal)

Patient was a 42-year-old male with no known significant medical history. He was a gastroenterologist in La Paz, and on June 4, 2019, performed an endoscopy S1-2.

Sixteen days post exposure (June 20), the patient presented and was admitted to Hospital A with a 2-day history of fever and severe headache refractory to NSAIDs and diarrhea that resolved after 24 h. On admission, he was afebrile and hemodynamically stable with no notable neurological deficits (Glasgow coma scale: 15/15). Laboratory testing on admission was notable

for leukopenia (WBC 3.1 x 10^9/L; RR: 4.5–11.0 x 10^9/L) and mildly elevated creatinine (1.5 mg/mL; RR: 0.6–1.2 mg/mL).

Symptoms worsened during hospitalization to include severe arthralgia and myalgia, anorexia, and fever (up to 38.5°C). On day 4 post symptom onset (June 22), laboratory testing revealed worsening leukopenia (WBC 2.3 x 10^9/L), rising creatinine (2.2 mg/mL), and new onset thrombocytopenia (platelets 145 x 10^9/L; RR:150–450 x 10^9/L). On day 7 (June 25), his clinical status briefly improved, with resolution of fever and improvement of arthralgia and myalgia for a period of 12 h. However, laboratory testing revealed persistent leukopenia (WBC 2.05 x 10^9/L), worsening thrombocytopenia (platelets 59.0 x 10^9/L), elevated liver function tests (AST 196 U/L, RR: 47–160 U/L; ALT 155 U/L, RR: 11.4–28.8 U/L), elevated creatine kinase-MB (570 µg/L; range: 0–7 µg/L), and elevated coagulation factors (PT 15 s, RR: 10–13 s; INR 1.27, RR: ≤ 1.1; D-dimer 147 mg/L, RR: ≤ 0.5 mg/L).

On day 8 (June 26), the patient became confused and agitated and developed hyporeflexia of the lower limbs and elevated creatine phosphokinase (2285 U/L, RR: 55–170 U/L). Electromyography was performed and revealed a mixed neurogenic-myogenic pattern. Due to worsening clinical status and acute neurologic decompensation, the patient was transferred to Hospital B the same day. Diagnostic testing performed on admission included a viral panel (cytomegalovirus, Epstein-Barr virus, herpes simplex virus, human immunodeficiency virus, and parvovirus B19), blood cultures, and immunological profile, which were all negative. Following transfer, the patient continued to be confused and agitated and had one self-limited, generalized tonic-clonic seizure. The initial diagnosis of the internal medicine service was post-infectious encephalomyelitis and polymyositis, for which methylprednisolone (single dose of 250mg; discontinued due to severe hyperglycemia) and intravenous immune globulin were administered.

On day 9 (June 27), the patient's mental status continued to deteriorate, and he developed massive gingival hemorrhage with a drop in hemoglobin from 14.4 to 8.6 g/dL (RR: 14.0–19.8 g/dL). The patient was transferred to the ICU, where he required mechanical ventilation for airway protection. Hehe was transfused with 4 units of packed red blood cells, 6 units 6Uof platelets (2 by apheresis), and tranexamic acid. On day 12 (June 30), following ICU transfer, the patient's exposure to a case of hemorrhagic fever of unknown etiology (later confirmed as CHHF patient S1-2) was established. He received 2 units of anti-MACV convalescent plasma and ribavirin (2g IV loading dose followed by 1g IV every 6 h for 4 days, then 500 mg every 8 h) for suspected arenavirus infection. Ribavirin was discontinued on July 8 due to worsening liver function.

On day 13 (July 1), patient developed ventilator-associated pneumonia due to *Acinetobacter baumannii*, for which broad-spectrum antibiotic therapy (meropenem and polymyxin) was started. Beginning on day 14 (July 2), patient developed scleral and dermal icterus and worsening hemorrhagic signs including melena, hematemesis, injection-site hemorrhage, and petechiae/ecchymoses. Laboratory testing revealed persistent leukopenia (WBC 3.5 x 10^9/L), anemia (hemoglobin 9.9 g/dL), thrombocytopenia (platelets 50.0 x 10^9/L), elevated liver function tests (AST 456 U/L; ALT 122 U/L; total bilirubin 3.0 mg/dL, RR: 0.2–1.2 mg/dL) and worsening renal function (creatinine 7.9 mg/mL). The patient's clinical status continued to deteriorate, and he developed multiorgan failure and hemorrhagic shock, persistent acute kidney injury requiring continuous renal replacement therapy, and increasing vasopressor and oxygen requirements. During hospitalization, the patient received 24 units of packed red blood cells, 23 units of fresh frozen plasma, 36 units of cryoprecipitates, 6 units of platelet concentrates, and 5 units of platelet concentrates by apheresis. The patient died on day 22 post-symptom onset (July

10). See Table S3c for daily account of the patient's clinical evolution and treatment throughout hospitalization.

S2.1.6 Patient S2-1 (fatal)

Patient was a 29-year-old pregnant female at 16 weeks gestation with no known underlying medical conditions or medications. Patient was an agricultural worker in Palos Blancos Municipality with no known epidemiologic links to the first cluster of CHHF cases in 2019 or any other subsequent CHHF cases.

On July 9, 2019, the patient developed fever and diarrhea. Six days later (July 15), she presented to a local hospital with complaints of fever, headache, myalgia, retro-orbital pain, abdominal pain, vomiting, lethargy, and gingival hemorrhage. After 2 h, the patient was transferred for more intensive care to a larger hospital (Hospital B) in the same municipality and admitted for 3 days under suspicion of dengue with warning signs. Laboratory testing revealed notable anemia (hemoglobin 8 g/dL; RR: 14–19.8 g/dL) and elevated transaminases (AST 962 U/L, RR: 47–160 U/L; ALT 261 U/L, RR: 11.4–28.8 U/L). On July 17, 8 days after symptom onset, the patient was transferred to a referral hospital (Hospital C) with continued gingival hemorrhage. On arrival in Hospital C, she was hemodynamically unstable and hypoxic (SpO_2 84%), with notable icterus, cervical lymphadenopathy, and respiratory distress. Chest radiograph revealed bilateral pleural effusion with infiltrates. The same day, labs were notable for thrombocytopenia (platelets 130 x 10^9/L, RR: 150–450 x 10^9/L), worsened transaminase levels (AST 1137 U/L; ALT 279 U/L), elevated clotting time (24 min, RR: 4–10 min), with slightly improved anemia (hemoglobin 9.9 g/dL). The same evening, she was transferred by ambulance to a tertiary care hospital in La Paz (Hospital D) due to her deteriorating clinical status. During ambulance transfer, the patient had a cardiac arrest and received cardiopulmonary resuscitation. She died the

following morning, 9 days post-symptom onset (July 18). No nosocomial transmission was associated with this case.

S2.2 Rodent trapping and CHAPV RT-qPCR testing

Rodent captures were completed on July 7, 8 and 9, 2019. Captures were successful in 2 of 3 trapping sites (Alto Sabaya, GPS coordinates: 15°41'08,1" S ; 67°32'21,3" W and Siliamo (Rice Fields), GPS coordinates: 15°35'23" S; 67°47'18" W); see table S4 for a summary of the trapping effort for each site. Of the 48 rodents captured in Caranavi and Guanay Municipalities, 52% (n=25) belonged to genus *Oligoryzomys,* 42% (n=20) to *Oryzomys*, 4% (n=2) to *Rattus*, and 2% (n=1) to *Marmosa*. One distinctive picture of each genus is presented below. See Table S5 for additional detailed data on rodent trapping and CHAPV RT-qPCR testing.

Rattus

Oligoryzomys

Oryzomys

Marmosa

S2.3 Bioinformatics and phylogenetic analysis

CHAPV-like reads from patient S1-3 were identified using EDGE (v2.4.0 dev) and vPipe (v1.0.0). A partial viral genome was produced by mapping reads to a CHAPV reference from 2003 (NC_010562-3) using an in-house pipeline (github.com/evk3/Nipah_phylogenetics) (consisting of adaptor removal (cutadapt), quality trimming (prinseq-lite -min_qual_mean 25 -trim_qual_right 20 -min_len 50), read mapping (bwa mem) and basecalling with Geneious/v 11.1.2, consensus threshold = 0%, majority or iVar). Reads did not map to Apore virus (MF317490-1) or Sabia virus (NC_006313,7). A complete viral genome was constructed from the viral isolate by mapping reads to a Chapare/2003 (NC_010562-3) reference using in-house scripts and using viral-ngs (version 1.22.1, Broad Institute) with a custom LASTAL database containing references from New World Arenaviruses clade B and C for a guided *de novo* assembly. Viral isolate consensus genomes were identical, differing 5′- (7 bp) and 3′-most (180 bp) viral termini (negative-sense). Consensus genomes from the first viral isolate (201901267-8) and rodents were generated using viral-ngs. All subsequent patient and viral isolate reads were mapped to the CHAPV Caranavi viral isolate sequence using Geneious or in-house scripts. Phylogenetic analysis was performed using mafft (v 7.450) aligner (-auto) and raxml (v. 8.2.12-PTHREAD) (-m GTRGAMMAI -p $RANDOM -f a -x $RANDOM -N 1000). Genomes can be accessed through Genbank at MZ772903-23 and raw reads through SRA at PRJNA753830. Phylogenetic analyses demonstrate that two unique CHAPV strains were circulating in Caranavi and Alto Beni/Palos Blancos regions from May-December 2019 (Figure 4). The analysis also supports CHAPV human-to-human transmission in the first cluster and additional introductions to humans later. There was no documented relationship between the first cluster of cases from Caranavi (S1) in May, the cases from Palos Blancos (S2) in July, or Alto Beni (S3) and Caranavi (S4) in December of 2019. The S4 virus sequence is closer to the rodent than to the S1 sequences. It is not possible to exclude the possibility of human-to-human transmission in S2,

S3, or S4, but the data suggests four different spillover events that likely occurred during agricultural work in the valley of North La Paz in 2019.

S2.4 Serological testing for Junin, Machupo, and Chapare viruses

Nine samples from five patients were tested for anti-Junin virus IgG serology. Only patient, S1-3, developed showed a significant increase in IgG titers in a sample collected 51 days after symptom onset compared to samples collected 8- and 14-days post-symptom onset. All other samples were negative for anti-JUNV IgG. A subset of samples tested negative using MACV IgM and IgG ELISA assays at CDC (*unpublished data*). Prior to this report, no C HAPV CHAPV serological assays were available. With the 2019 CHAPV isolates and specimens, CDC's Viral Special Pathogens Branch was able to develop in-house anti-CHAPV IgM and IgG detection assays. All serum and blood samples from CHAPV 2019-confirmed cases for which the CDC had sufficient sample volume were inactivated and tested with these assays as described in the supplemental materials and methods section (section 1.6). The results are summarized in Table S1 below. Despite some limitations of available samples, seroconversion (change from absent to positive titers of anti-CHAPV IgM and IgG antibodies when comparing samples collected during acute disease to later samples) was observed in survivor S1-3, S1-4, and S3-1. We noticed that the presence of those antibodies did not preclude the detection of CHAPV RNA in S1-3 and S1-4. Interestingly, positive IgM and IgG antibody titers were detectable in survivor S1-3 and S1-4 up to 190- and 170-days post-symptom onset, respectively. Only one convalescent sample (24 days post symptom onset) was available from S1-3, and in this specimen, the IgM (1:6400) was higher than the IgG (1:1600) titer. Unfortunately, we did not have enough serum specimens to perform neutralization tests. Further investigations of survivors are needed to determine their current antibody response status and whether their antibodies to CHAPV are neutralizing.

S3 Supplemental Tables and Figures

S3.1 Table S1: RT-qPCR, virus isolation, and serology results for CHAPV-positive specimens from 2019 CHHF patients

Patient # -gender, age *(outcome)*	Specimen type – collection date	No. of days post-symptom onset	RT-qPCR Ct values (S/L)*	anti-CHAPV † IgM titer	anti-CHAPV † IgG titer	Viral isolate (Specimen/Date) ‡
S1-2 - Female, 25 y *(fatal)*	Serum –24 May	**4**	29/32	NA	NA	Y (Serum / 24 May)
	Serum – 28 May	8	29/32	NA	NA	
S1-3 - Male, 21 y *(survivor)*	Whole blood - 6 Jun	8	32/34	1:50	1:50	Y (Semen / 23 Aug)
	Serum – 6 Jun	8	ND/ND	1:50	1:50	
	Serum – 12 Jun	14	ND/ND	1:50	1:50	
	Urine – 7 Jul	40	38/ND			
	Whole blood – 19 Jul	51	31/34	**1:6400**	**1:6400**	
	Whole blood – 14 Aug	77	32/35	**1:400**	**1:6400**	
	Semen– 23 Aug	**86**	29/30			
	Whole blood – 2 Sep	96	ND/ND	**1:400**	**1:6400**	
	Whole blood – 19 Sep	113	ND/ND	**1:1600**	**1:6400**	
	Semen - 19 Sep	113	32/34			
	Whole blood – 5 Dec	190	ND/ND	**1:400**	**1:6400**	
S1-4 - Male, 48 y *(survivor)*	Whole blood – 27 Jun	9	26/29	1:50	1:50	Y (Serum / 27 Jun)
	Serum – 27 Jun	**9**	30/32	1:50	1:50	
	Blood Clot – 27 Jun	9	28/31	1:50	1:50	
	Bronchoalveolar lavage – 27 Jun	9	28/30			
	Bronchoalveolar lavage – 12 Ju1	24	25/29			
	Whole blood – 2 Aug	45	21/22	**1:6400**	**1:6400**	
	Whole blood – 13 Aug	56	23/24	**1:6400**	**1:6400**	
	NP swab – 13 Aug	56	29/29			
	Urine – 13 Aug	56	33/38			
	Serum – 23 Aug	66	30/34	**1:6400**	**1:6400**	
	NP swab – 23 Aug	66	38/40			
	Urine – 23 Aug	66	33/36			
	Conjunctiva – 23 Aug	66	34/36			
	Whole blood – 2 Sep	76	23/24	**1 :6400**	**1 :6400**	
	Serum – 2 Sep	76	28/31	**1 :6400**	**1:6400**	
	Semen – 2 Sep	76	24/28			
	Whole blood – 19 Sep	93	24/25	**1 :1600**	**1:6400**	
	Urine – 19 Sep	93	35/38			
	Whole blood – 5 Dec	170	36/ND	**1 :1600**	**1:6400**	
	Semen – 5 Dec	170	33/36			
S1-5 - Male, 42 y *(fatal)*	Whole blood – 28 Jun	**10**	25/27	1:50	1:50	Y (Blood / 28 Jun)
	Serum - 28 Jun	10	27/29	NA	NA	
S2-1 - Female, 29y *(fatal)*	Serum – 15 Jul	7	28/33	1:50	1:50	Y (Serum 17 Jul)
	Serum – 17 Jul	**9**	28/30	NA	NA	
S3-1 - Male, 47 y *(survivor)*	Whole blood – 13 Dec	10	27/28	1:50	1:50	
	NP swab– 13 Dec	10	27/31			
	OP swab– 13 Dec	10	26/30			
	Bronchoalveolar lavage – 13 Dec	10	25/30			
	Urine– 13 Dec	10	40/ND			
	Whole blood – 27 Dec	24	31/ND	**1:6400**	**1:1600**	
	Urine– 27 Dec	24	35/38			
S4-1 - Female, 27 y *(survivor)*	Whole blood – 31 Dec	26	28/30	1:50	1:50	
	NP swab – 31 Dec	26	31/30			
	Bronchoalveolar lavage – 31 Dec	26	31/30			
	Urine – 31 Dec	26	35/36			
S4-2 - Male, 7 y *(survivor)*	Serum – 31 Dec	8	38/ND	1:50	1:50	

* For RTq-PCR results, ND = not detected. Ct values for S and L targets are presented as an indication of CHAPV RNA detection with the 2019 CHAPV RT-qPCR assays developed using sequence information of initial samples from the 2019 outbreak.

† Serology results are presented for all serum and blood samples from CHAPV 2019 outbreak patients that were available at CDC; NA = sample not available. For IgM and IgG, a titer <1:400 is considered negative; a titer ≥1:400 is considered positive and is indicated in **bold**.

‡ Multiple specimens were tested for virus isolation but only selected positive samples are reported in this table. Y = yes, an isolate was obtained; for samples from which an isolate was obtained, the number of days post-symptom onset is **bolded and underlined** in the corresponding column.

S3.2 Table S2. Summary of next generation sequencing mapping metrics

Accession Number	Specimen Identifier	Tree Label	Segment	Sequence Length	Percent of Ns	Percent Abiguious bases	Percent Genome Coverage	Included in Trees?	Number of Mapped Reads	Average Fold Coverage
MZ772917	202003462	Case S3-1, Alto Beni, 13-Dec-2019 (202003462)	S	3350	0.2	0.0	99.8	Yes	303	13.0
MZ772918			L	7053	10.2	0.1	89.7	Yes	231	4.8
MZ772915	202003458 viral isolate	Case S2-1, Palos Blancos, 17-Jul-2019 (202003458_vi)	S	3350	0.1	0.0	99.8	Yes	1249	104.9
MZ772916			L	7083	0.0	0.0	100.0	Yes	779	22.0
MZ772913	202003458	Case S2-1, Palos Blancos, 17-Jul-2019 (202003458)	S	3352	0.1	0.0	99.9	Yes	721	31.7
MZ772914			L	7053	0.0	0.0	100.0	Yes	823	17.1
MZ772920	Bolivia-27S	O. microtis 27S, Caranavi, Jul-2019	S	3350	0.0	0.0	100.0	Yes	3907	175.4
MZ772921			L	7048	0.0	0.0	100.0	Yes	7636	163.3
MZ772922	Bolivia-30O	O. microtis 30O, Caranavi, Jul-2019	S	3350	0.4	0.0	99.5	Yes	286	13.7
MZ772923			L	7074	0.5	0.0	99.4	Yes	258	5.7
MZ772919	202003467	Case S4-1, Caranavi, 31-Dec-2019 (202003467)	S	3350	1.4	0.1	98.5	Yes	159	6.8
			L	7047	37.1	0.1	62.8	No	114	2.3
MZ772907	201901324	Case S1-5, La Paz, 28-Jun-2019 (201901324)	S	3350	9.7	0.2	90.1	Yes	119	5.1
			L	7047	65.5	0.0	34.4	No	60	1.2
MZ772912	202003440	Case S1-4, La Paz, 2-Sep-2019 (202003440)	S	3356	9.2	0.3	90.6	Yes	130	5.4
			L	7056	60.9	0.1	39.0	No	65	1.2
MZ772905	201901267-8 viral isolate	Case S1-2, Caranavi, 24/28-May-2019 (201901267-8_vi)	S	3370	0.6	0.0	99.4	Yes	100204	4633.8
MZ772906			L	7104	0.8	0.0	99.2	Yes	56880	1207.9
MZ772903	201901267-8	Case S1-2, Caranavi, 24/28-May-2019 (201901267-8)	S	3350	0.0	0.0	100.0	Yes	1673	74.8
MZ772904			L	7062	0.0	0.1	99.9	Yes	1109	23.5
MZ772911	202003410 viral isolate	Case S1-3, Caranavi, 23-Aug-2019 (202003410_vi)	S	3350	1.1	0.0	98.9	Yes	2397	53.6
			L	7047	24.7	0.1	75.2	No	1067	15.8
MZ772909	201901328	Case S1-4, La Paz, 27-Jun-2019 (201901328)	S	3350	0.3	0.0	99.7	Yes	1281	56.8
MZ772910			L	7086	0.1	0.0	99.9	Yes	758	16.0
MZ772908	201901326	Case S1-4, La Paz, 28-Jun-2019 (201901326)	S	3351	1.9	0.1	98.0	Yes	130	5.6
			L	7047	51.8	0.0	48.1	No	76	1.5

S3.3 Table S3a. Patient S1-3 (survivor): Clinical evolution and treatment throughout first 3 weeks of illness

		Date in May - June 2019 (Date post-symptom onset)																			
		5/30 (0)	5/31 (1)	6/1 (2)	6/2 (3)	6/3 (4)	6/4 (5)	6/5 (6)	6/6* (7)	6/7 (8)	6/8 (9)	6/9 (10)	6/10 (11)	6/11 (12)	6/12 (13)	6/13 (14)	6/14 (15)	6/15 (16)	6/16 (17)	6/17 (18)	6/18 (19)	6/19‡ (20)
Signs and Symptoms																					
Fever		x	x	x	x	x	x	x	x	x	x	x									
Headache		x	x	x	x	x	x	x	x	†											
Retro-orbital pain		x	x	x	x	x	x	x	x	†											
Myalgia		x	x	x	x	x	x	x	x	†											
Arthralgia		x	x	x	x	x	x	x	x	†											
Malaise		x	x	x	x	x	x	x	x	†											
Abdominal pain					x	x	x	x	x	†											
Vomiting					x	x	x	x	x	†											
Diarrhea										†											
Respiratory difficulty									x	†											
Confusion/agitation					x	x	x	x	x	†											
Generalized seizure									x												
Petechiae/ecchymoses										x	x	x									
Hemorrhagic signs					x	x	x	x	x	x	x	x									
Shock										x	x	x	x	x	x	x					
Heart rate (bpm)										110											
Mean arterial pressure (mmHg)										84											
SpO2 (84-95%)										93	83										
Glasgow Coma Scale (3-15; -4 = intubated)											8										
Laboratory test	**Reference range**																				
Leukocytes	4.5-11 x 10^9/L								2.3	3.5	7.0		12.1							12.1	
Hemoglobin	14-19.8 g/dL								16	10.5	7.3		8.9							8.9	
Hematocrit	43-58%								49	33	23		28							28	
Platelets	150-450 x 10^9/L								105		198		314							314	
Total bilirubin	0.2-1.2 mg/dL									2.3	3.2		2							2	
Direct bilirubin	0-0.3 mg/dL									1.5	2.7		0.9							0.9	
AST	47-160 U/L								1796		1068		148							148	
ALT	11.4 - 28.8 U/L								1197		712.9		119							119	
Creatinine	.6-1.2 mg/mL										0.9		0.7							0.7	
Sodium	135 – 146 mmol/L									132											
Potassium	3.5 - 5.5 mmol/L									3.5											
PTTa	30-40 seconds								65												
INR	≤1.1								1.6												
Treatment																					
Vasopressor									x	x	x	x	x	x	x	x					
Mechanical ventilation									x	x	x	x	x	x	x	x	x	x	x	x	x
Packed red blood cells											x										
Fresh frozen plasma									x		x										
Platelet concentrates											x										
Cryoprecipitates									x												
Broad spectrum antimicrobials											x	x	x	x	x	x	x	x	x	x	

* Day of admission to local hospital in Caranavi; transferred same day to tertiary care hospital in La Paz.

† Not assessed after patient began mechanical ventilation.

‡ Patient extubated on June 21 (day 22 post-symptom onset); discharged from ICU on June 24 (day 25) and discharged from hospital on June 30 (day 31). Neurological deficits including disorientation and confusion persisted for months after discharge.

S3.4 Table S3b. Patient S1-4 (survivor): Clinical evolution and treatment throughout first 2 weeks of illness

		Date in June – July 2019 (Date post-symptom onset)														
		6/18 (0)	6/19 (1)	6/20 (2)	6/21 (3)	6/22 (4)	6/23 (5)	6/24‡ (6)	6/25 (7)	6/26‡ (8)	6/27 (9)	6/28 (10)	6/29 (11)	6/30 (12)	7/1 (13)	7/2§ (14)
Signs and Symptoms																
Fever*																
Headache		x	x	x	x	x	x	x	x	x	x	x	x	x	x	x
Myalgia		x	x	x	x	x	x	x	x	x	x	x	x	x	x	x
Arthralgia		x	x	x	x	x	x	x	x	x	x	x	x	x	x	x
Malaise		x	x	x	x	x	x	x	x	x	x	x	x	x	x	x
Arthralgia							x	x x	x x	x x x	x x x	x x x	x x x	x x x	x x x	x x
Muscle weakness †																
Abdominal pain																
Nausea																
Vomiting																
Diarrhea																
Confusion/agitation †												x	x x	x x	x x	x x x
Generalized seizure																
Petechiae/ecchymoses																
Hemorrhagic signs †												x	xx	xx	xx	x
Shock																
Laboratory test	**Reference range**															
Leukocytes	4.5-11 x 10^9/L							2.9		2.1		2.1		1.02	.97	
Hemoglobin	14-19.8 g/dL							17.1		16.5		15.8		14.2	12.9	
Hematocrit	43-58%							49.7		51		48		43.4	38.9	
Platelets	150-450 x 10^9/L							106		96		53		34	35	
Albumin	3.5-5.5 g/dL										3.5			3.3	1.9	
Total bilirubin	0.2-1.2 mg/dL										2.4	3.81	5.8	4.2	4.8	6.26
AST	47-160 U/L										291	145	1750	811	398	1670
ALT	11.4 - 28.8 U/L										229	595	743	662	389	531
ALP	44-147 U/L										345.1	314				
Creatinine	.6-1.2 mg/mL							1.7		3	2.3	1.6	1.5	1.6	1.7	1.5
BUN	7-20 mg/dL									77.67	62.44	48.28	38.62			27.89
Lactate dehydrogenase	140 - 280 U/L											3307				
Glucose	70-80 mg/dL							107								
Sodium	135 – 146 mmol/L							140		140						
Potassium	3.5 - 5.5 mmol/L							4.1		4.2						
Troponin																
CPK-MB	0-7 ug/L															
CPK Total	55-170 U/L											1390				
C-reactive protein	< 6 mg/dL							normal		normal						high
PT	10-13 seconds											13				
PTTa	30-40 seconds											35.2	63			
INR	≤1.1									1		1	1	1	1	
Fibrinogen activity	200-400 mg/dL											375.5				
D Dimer	≤0.5 mg/L															
Treatment																
Fresh frozen plasma (U)												x	x	x	x	x
Platelet concentrates (U)												x	x	x	x	x
IV immunoglobulin (g/kg)										2	2	2	2	2		
Ribavirin^															x	x
Anti-Machupo convalescent plasma (U)															1	
Meropenem												x	x	x	x	x
Acyclovir												x	x	x	x	x

* Patient remained afebrile throughout illness.

† **x** = Mild; **xx** = Moderate; **xxx** = Severe

‡ Admitted to private tertiary care hospital in La Paz on June 24; transferred to nearby tertiary care hospital on June 26.

^ On July 1 (day 13 post-symptom onset, possibility of arenavirus infection is established; oral ribavirin is administered and continued for 5 days.

§ On July 6 (day 18 post-symptom onset) patient developed hypoxemic respiratory failure and required mechanical ventilation that was maintained until August 12 (day 55). On August 23 (day 66) he was discharged from ICU to private hospital room, but remained hospitalized until November 26 (day 161) out of precaution related to persistent Chapare virus RNA detection in blood samples. He continued to have difficulty walking in the months following discharge and required physical therapy to regain function of his lower limbs.

S3.5 Table S3c. Patient S1-5 (fatal): Clinical evolution and treatment throughout course of hospitalization

		Date in June – July 2019 (Date post-symptom onset)																				
		6/20†	6/21	6/22	6/23	6/24	6/25	6/26†	6/27	6/28	6/29	6/30	7/1	7/2	7/3	7/4	7/5	7/6	7/7	7/8	7/9	7/10§
		(2)	(3)	(4)	(5)	(6)	(7)	(8)	(9)	(10)	(11)	(12)	(13)	(14)	(15)	(16)	(17)	(18)	(19)	(20)	(21)	(22)
Signs and Symptoms																						
Fever		x*	x	x*	x*	x*	x‡	x*	x*	x*	x	x	x	x	x	x	x	x	x*	x*	x*	x*
Headache		x	x	x	x	x	x	x	^													
Myalgia			x	x	x	x	x‡	x	^													
Arthralgia			x	x	x	x	x‡	x	^													
Abdominal pain									^													
Nausea			x	x	x	x	x	x	^													
Vomiting									^													
Diarrhea									^													
Confusion/agitation								x	x^													
Generalized seizure								x														
Petechiae/ecchymoses														x	x	x	x	x	x	x	x	x
Hemorrhagic signs									x	x	x	x	x	x	x	x	x	x	x	x	x	x
Shock										x	x	x	x	x	x	x	x	x	x	x	x	x
Heart rate (bpm)		84	74	89	64	68	90	64	92	77	82	105	118	136	146	136	141	143	108	110	77	140
Respiratory rate (brpm)		20	18	20	18	20	24	17	22	24	22	24	22	31	20	26	30	20	27	26	26	28
Mean arterial pressure (mmHg)		93	83	90	83	73	83	70	70	71	71	59	66	57	49	56	58	61	49	54	84	60
SpO2 (84-95%)		88	89	84	84	83	84	83	87	90	96	88	93	92	92	91	85	92	90	89	90	87
Glasgow Coma Scale (3-15; -4 = intubated)		15	15	15	15	14	14	14	-4	-4	-4	-4	-4	-4	-4	-4	-4	-4	-4	-4	3	3
Estimated blood loss (mL)										1650		1980	460	930	1055	1080	260	550	100	300	200	300
Laboratory test	**Reference range**																					
Leukocytes	4.5-11 x 10^9/L	3.1	2.4	2.3		2.0	2.1	2.0	1.8	5.0	1.5	8.3	4.0	3.5	4.7	4.3	3.9	4.1	4.2	5.0	4.2	5.3
Hemoglobin	14-19.8 g/dL	17.9	17.2	15.9				18	16.9	14	14.4	8.6	8	9.9	8.6	11	11.6	9.3	11.3	10	10	10
Hematocrit	43-58%	56	52	43				54	53.4	42	45	26	24	30	26	33	35	28	34	30	30	30
Platelets	150-450 x 10^9/L	207	174	145		96	59	78	89	69	78	50	65	50	42	48	33	40	47	50	50	48
Total bilirubin	0.2-1.2 mg/dL		1				1.5	2.5	3.6	3.1		2.8	2.8	3	4.2	5.7	5.2	5.3	11.2	14.8	14.8	15
AST	47-160 U/L		28				196	704	194		510	424	450	456	547	366	360	160	2420		7.98	798
ALT	11.4 - 28.8 U/L		29				155	414	499		148	150	148	122	210	244	250	419	540		518	518
Creatinine	.6-1.2 mg/mL	1.5	2.2	2.2			2	1.7	1.9	2.6	2.3	4.4	7.17	7.9	7.6	7.2	6.7	6.6	6.2	6	6	5.6
BUN	7-20 mg/dL	13		20			19	15	36	40	36	38	95.2	108	108	106	100	107	100	113	114	124
Lactate dehydrogenase	140 - 280 U/L						995		3476			2019	2824	2279				2300				87
Glucose	70-80 mg/dL	122	98					215	456	276	466	346	576	656	694	638	256	320	420	292	160	316
Sodium	135 – 146 mmol/L		139				140	135	145	148	146	146	145	138	134	129	139	129	128	130	138	127
Potassium	3.5 - 5.5 mmol/L		4.3				4.2	3.75	5.1	5	5.30	5.1	6.37	6.64	6.68	6	5	5	6.27	6.12	4.49	5.96
Troponin												neg	neg	neg			pos	pos		pos	pos	pos
CPK-MB	0-7 ug/L						570					54	500	253		252	210	312			315	318
CPK Total	55-170 U/L							2285	2600			1804	800	376			neg	2016		2018	2080	1272
PT	10-13 seconds						15	16	14	14.5	12	12.9	12.5	15	15.1	15.4	17	16	15.8	15.8	15	17
PTTa	30-40 seconds						33									171	0	0	0	67.3	15	
INR	≤1.1						1.27	1.44	1.23	1.26	1	1.1	1.15	1.31	1.31	1.34	1.57	1.41	1.43	1.40	1.4	1.5
Fibrinogen activity	200-400 mg/dL						80	57	70	67.7	95.9	84	87.7	64.4	63.8	62	53.3	40.3	58	60.3	30	51
D Dimer	≤0.5 mg/L						147									12	243		399	332	332	
Treatment																						
Vasopressor										x	x	x	x	x	x	x	x	x	x	x	x	x
Mechanical ventilation									x	x	x	x	x	x	x	x	x	x	x	x	x	x
Ultrafiltration (mL)														2000	2500	1500	2500	1500	2000	4000	2000	0
Renal replacement therapy														x	x	x	x	x	x	x	x	x
Packed red blood cells (U)												4		8		4		4		2	2	
Fresh frozen plasma (U)									4	4	9							4		2		
Platelet concentrates (U)											2	4										
Platelets by apharesis (U)												2					1	2				
Cryoprecipitates (U)													2			12	4	4	4	12		
Tranexamic acid												1	1	1	1	1	1	1	1	1		
Anti-Machupo convalescent plasma (U)												2										
Ribavirin													x	x	x	x	x	x	x	x		
Meropenem													x	x	x	x	x	x	x	x		
Polymyxin													x	x	x	x	x	x	x	x		
Acyclovir										x	x	x										
Oseltamivir									x	x												

* Subjective fever; anti-pyretics were used throughout hospitalization to control fever.

† Admitted to community hospital in La Paz on June 20; transferred to tertiary care hospital on June 26 due to worsening clinical status.

‡ On June 25 (day 7 post-symptom onset), clinical picture improved for a period of 12 hours, including resolution of fever an improved arthralgia and myalgia.

^ Not assessed after patient began mechanical ventilation.

§ Patient died on July 10, 22 days post-symptom onset.

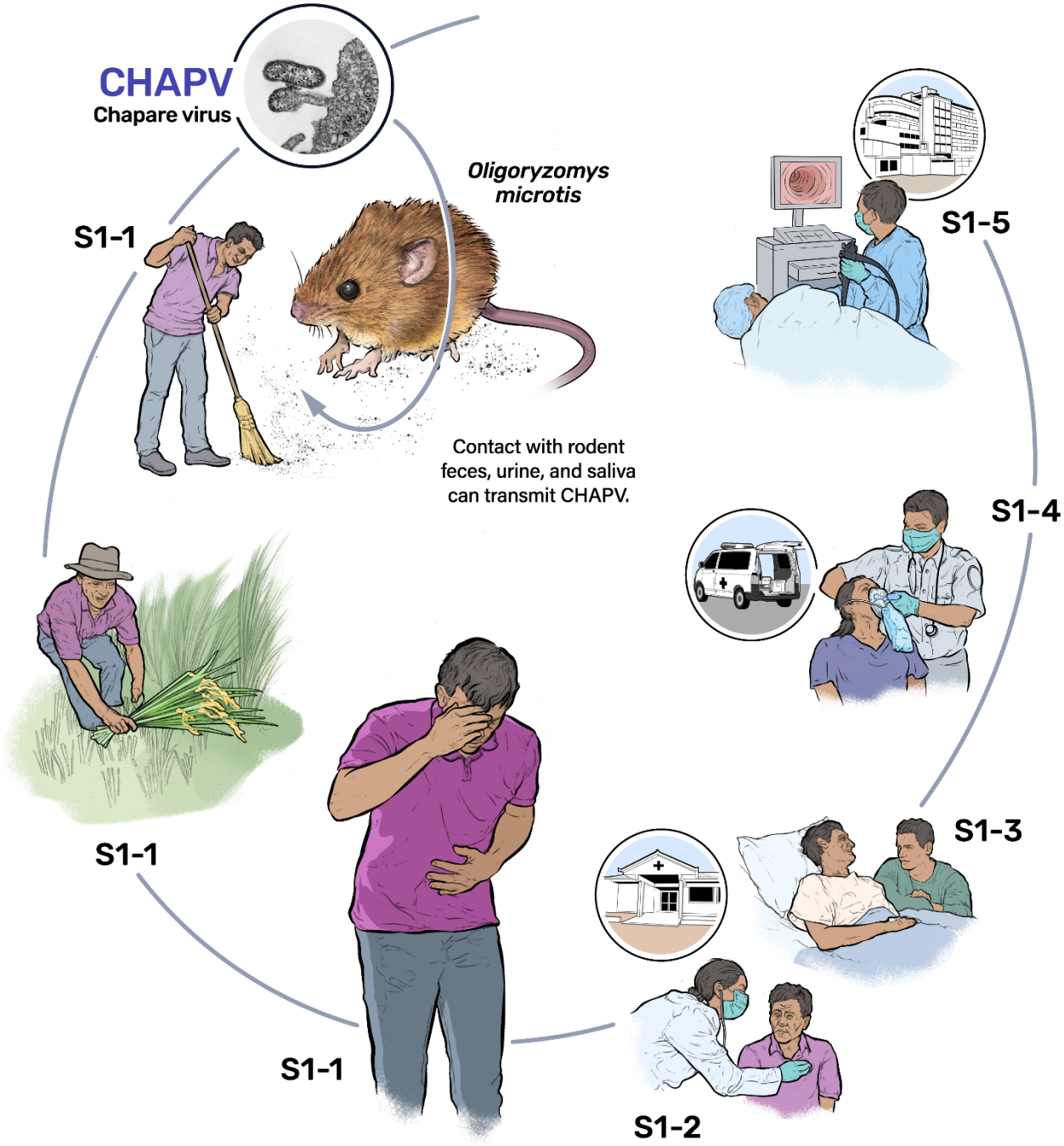
S3.6 Table S4. Summary of rodent trapping effort between July 7-9, 2019

Department	Municipality	Community*	Sector	#Traps	#Successful traps	Trap Index	Genus	Quantity
LaPaz	Caranavi	Alto Sabaya	Peridomicile index case	100	2	2	*Oryzomys*	2
		Huayna Pata	Peridomicile patient S1-3	100	0	0	-	0
	Guanay	Siliamo	Intradomicile Houses in the sector	7	2	29	*Rattus*	2
			Temporal storage rice bags	100	12	12	*Oligoryzomys*	11
							Oryzomys	1
			Rice fields	86	11	13	*Oligoryzomys*	4
							Oryzomys	7
			Intra and peridomicilie Index case	20	5	25	*Oligoryzomys*	3
							Oryzomys	1
							Marmosa	1
			Temporal storage rice bags	10	4	40	*Oligoryzomys*	2
							Oryzomys	2
			Bushes at the edge of rice fields	20	3	15	*Oligoryzomys*	1
							Oryzomys	2
			Temporal storage rice bags	28	10	36	*Oligoryzomys*	5
							Oryzomys	5
						Total		**49**

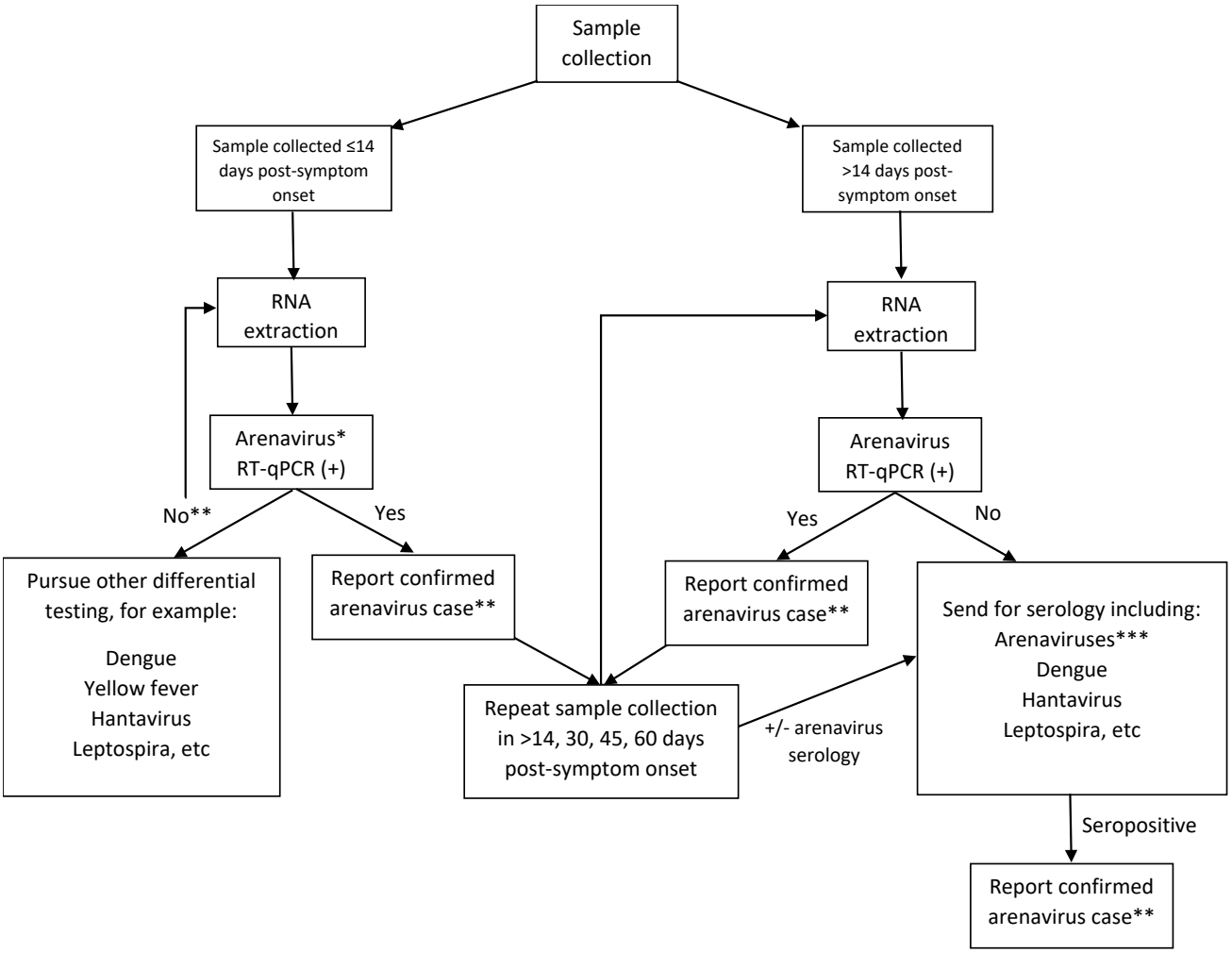
S3.7 Table S5. Detailed data on rodent trapping and CHAPV RT-qPCR testing

General information				Morphometric measurements									Biological specimens					Observations	CHAPV RT-qPCR
Collection date	Site	Code	Trap #	Genus	Sex	Weight (gr)	Total length (cm)	Body (cm)	Tail (cm)	Ear (cm)	Leg (cm)	# Embryos	Blood	Spleen (cm)	Kidney	Feces	Urine		
07/07/2019	Alto Sabaya	AS1	1	*Oryzomys*	F	<100	28	13	15	2.5	3	3	No	1,9	X			Presence of mites	NOT TESTED
07/07/2019	Alto Sabaya	AS2	2	*Oryzomys*	M	<100	26.5	11.5	15	2.3	3.2		No	2,2	X			Presence of mites	NOT TESTED
07/07/2019	Siliamo	VI1	1	*Rattus*	M	48	23.5	9.5.	15.3	2.2	2.5		X	1,6	X				NOT TESTED
07/07/2019	Siliamo	VI2	2	*Rattus*	M	<100	33	15	18	2	3		X	3,5	X				NOT TESTED
07/07/2019	Siliamo	ATM3	3	*Oligoryzomys*	F	31	18.1	7.8	10.5	1.4	2		X	1,2	X			Presence of mites	NOT TESTED
07/07/2019	Siliamo	ATM4	4	*Oligoryzomys*	M	37	18.5	8	10.5	1.4	2.1		X	1,8	X			Presence of boron and mites	NOT TESTED
07/07/2019	Siliamo	ATM5	5	*Oryzomys*	M	<100	27	12.8	14.2	2.6	3.1		X	3,3	X				NOT TESTED
07/07/2019	Siliamo	ATM6	6	*Oligoryzomys*	F	47	21.1	8.8	12.5	1.6	2.1	4	X	1,4	X				NOT TESTED
07/07/2019	Siliamo	ATM7	7	*Oligoryzomys*	M	16	14	6.5	8	1.4	2		X	1	X			Presence of mites	NOT TESTED
07/07/2019	Siliamo	ATM8	8	*Oligoryzomys*	F	42	19.6	8.8	11	1.4	2.1		X	1,2	X				NOT TESTED
07/07/2019	Siliamo	ATM9	9	*Oligoryzomys*	F	46	18.5	8.5	10.4	1.4	2.1		X	2,5	X			Presence of boron and mites	NOT TESTED
07/07/2019	Siliamo	ATM10	10	*Oligoryzomys*	F	41	19.4	9	10.4	1.3	2.2	4	X	1,7	X			Presence of mites	NOT TESTED
07/07/2019	Siliamo	ATM11	11	*Oligoryzomys*	M	45	20	9	11	1.9	2.2		X	1,6	X				NOT TESTED
07/07/2019	Siliamo	ATM12	12	*Oligoryzomys*	F	36	18.5	7.9	10.2	1.4	2.1	3	X	1,2	X				NOT TESTED
07/07/2019	Siliamo	ATM13	13	*Oligoryzomys*	F	33	19	8	10.5	1.3	2		X	1,3	X				NOT TESTED
07/08/2019	Siliamo	AC14	14	*Oryzomys*	M	<100	26.8	13.5	13.1	1.8	3.2		X	2,1	X	X			NEGATIVE
07/08/2019	Siliamo	AC15	15	*Oryzomys*	M	<100	27.3	18.8	13.6	2	3.2		X	2,4	X			Presence of mites and ticks	NEGATIVE
07/08/2019	Siliamo	AC16	16	*Oryzomys*	F	<100	28.2	14.2	14	2	3.2	4	X	2,2	X	X	X	Presence of mites and ticks	NEGATIVE
07/08/2019	Siliamo	AC17	17	*Oligoryzomys*	F	12	13.2	5.6	7.5	0.8	1.8		X	0,5	X			Presence of mites	NEGATIVE
07/08/2019	**Siliamo**	**AC18**	**18**	***Oligoryzomys***	**F**	**30**	**18.5**	**8.3**	**10.2**	**1.1**	**2.2**		**X**	**0,8**	**X**		**X**	**Presence of mites and ticks**	**POSITIVE**
07/08/2019	Siliamo	AC19	19	*Oligoryzomys*	M	54	21.3	10	11.5	1.3	2.4		X	1,4	X			Presence of mites	NEGATIVE
07/08/2019	Siliamo	AC20	20	*Oryzomys*	F	<100	27.1	13.5	14	2.1	3.3		X	2,2	X	X		Presence of mites	NEGATIVE
07/08/2019	Siliamo	AC21	21	*Oryzomys*	F	<100	27.2	13	14.2	2.1	3.1		X	2,2	X	X		Presence of mites	NEGATIVE
07/08/2019	Siliamo	AC22	22	*Oryzomys*	M	<100	28.7	13.5	15	2.3	3.2		X	2,4	X			Presence of mites and ticks	NEGATIVE
07/08/2019	**Siliamo**	**AC23**	**23**	***Oligoryzomys***	**F**	**39**	**20.6**	**9**	**11.5**	**0.8**	**2.3**	**4**	**X**	**1,8**	**X**	**X**		**Presence of mites and ticks**	**POSITIVE**
07/08/2019	Siliamo	AC24	24	*Oryzomys*	M	<100	28.6	13.5	15.1	2.1	3.1		X	3,1	X			Presence of boron and mites	NEGATIVE
07/09/2019	Siliamo	VM25	25	*Oligoryzomys*	F	42	22	9	11	1.4	2	5	X	1,4	X			Presence of mites and ticks 10	NEGATIVE
07/09/2019	**Siliamo**	**VM26**	**26**	***Oligoryzomys***	**F**	**45**	**21.9**	**10**	**11.9**	**1**	**1.4**		**X**	**1,4**	**X**				**POSITIVE**
07/09/2019	**Siliamo**	**VM27**	**27**	***Oligoryzomys***	**F**	**32**	**17.7**	**8**	**9.2**	**1.2**	**2.1**		**X**	**1,6**	**X**	**X**		**Presence of mites and ticks 7**	**POSITIVE**
07/09/2019	Siliamo	VM28	28	*Marmosa*	F	<100	23.5	15.8	8	1.7	2.1		X	3,2	X	X	X		NEGATIVE
07/09/2019	Siliamo	VM29	29	*Oryzomys*	F	<100	28.3	13	14.5	2.3	3.1	4	X	1,6	X	X		Presence of mites	NEGATIVE
07/09/2019	**Siliamo**	**CBV30**	**30**	***Oligoryzomys***	**F**	**43**	**20.7**	**9.3**	**11.5**	**1.4**	**2.2**		**X**	**1,3**	**X**		**X**	**Scars in ear and mites**	**POSITIVE**
07/09/2019	Siliamo	CBV31	31	*Oryzomys*	F	<100	28.6	14.7	14.6	2.3	3.4		X	2,3	X	X			NEGATIVE
07/09/2019	**Siliamo**	**CBV32**	**32**	***Oligoryzomys***	**M**	**16**	**15**	**6**	**8.5**	**1.3**	**1.9**		**X**	**0,8**	**X**	**X**		**Presence of mites 15**	**POSITIVE**
07/09/2019	**Siliamo**	**CBV33**	**33**	***Oligoryzomys***	**F**	**39**	**19.1**	**9**	**10.2**	**1.4**	**2.2**		**X**	**1,1**	**X**				**POSITIVE**
07/09/2019	Siliamo	CBV34	34	*Oryzomys*	F	<100	24	12.5	12.4	2.1	2.9		X	1,7	X			Presence of mites and ticks	NEGATIVE
07/09/2019	Siliamo	CBV35	35	*Oryzomys*	F	29	15.9	8	8.3	1.6	2.5		X	1,3	X			Presence of mites 10	NEGATIVE
07/09/2019	**Siliamo**	**CBV36**	**36**	***Oryzomys***	**M**	**<100**	**28.1**	**13.6**	**14.8**	**2.1**	**3.4**		**X**	**2,2**	**X**			**Ear scars and presence of mites**	**POSITIVE**
07/09/2019	Siliamo	CBV37	37	*Oligoryzomys*	M	14	13.6	6.1	7	1	2.1		X	1	X			Presence of mites 15	NEGATIVE
07/09/2019	Siliamo	CBV38	38	*Oryzomys*	M	41	18.1	9.5	8.3	1.9	3		X	1,2	X				NEGATIVE
07/09/2019	**Siliamo**	**CBV39**	**39**	***Oligoryzomys***	**M**	**30**	**19**	**8.5**	**10.5**	**1.4**	**2.2**		**X**	**1,1**	**X**		**X**	**Presence of mites 4**	**POSITIVE**
07/09/2019	Siliamo	CM40	40	*Oligoryzomys*	M	12	14.6	6.5	7	1.3	2		X	0,4	X			Presence of mites 6	NEGATIVE
07/09/2019	Siliamo	CM41	41	*Oryzomys*	F	57	20.5	10	10.3	1.8	2.8		X	1,3	X			Presence of mites 3	NEGATIVE
07/09/2019	Siliamo	CM42	42	*Oryzomys*	M	63	22	10.8	10.5	1.9	3.1		X	1,6	X			Presence of mites 4	NEGATIVE
07/09/2019	Siliamo	CM43	43	*Oligoryzomys*	F	36	18.9	8.7	10.1	1.3	2.2		X	1	X	X		Presence of mites 6	NEGATIVE
07/09/2019	Siliamo	MA44	44	*Oligoryzomys*	M	42	19.7	9.4	10.8	1.3	2.2		X	1,6	X	X			NEGATIVE
07/09/2019	Siliamo	MA45	45	*Oryzomys*	M	54	21.7	10	11.7	1.4	2.6			2	X			Presence of mites 4 and 6 ticks	NOT TESTED
07/09/2019	Siliamo	MA46	46	*Oryzomys*	M	<100	28	13.5	15	2.1	3.2		X	2,1	X			Presence of mites 10	NOT TESTED

S3.8 Figure S1. Illustration of CHAPV initial (S1) outbreak with nosocomial infections



S3.9 Figure S2a. Testing algorithm to rule out Chapare or other New World arenaviruses in suspect viral hemorrhagic fever cases in a BSL3 laboratory prior to conducting other laboratory testing (English version).

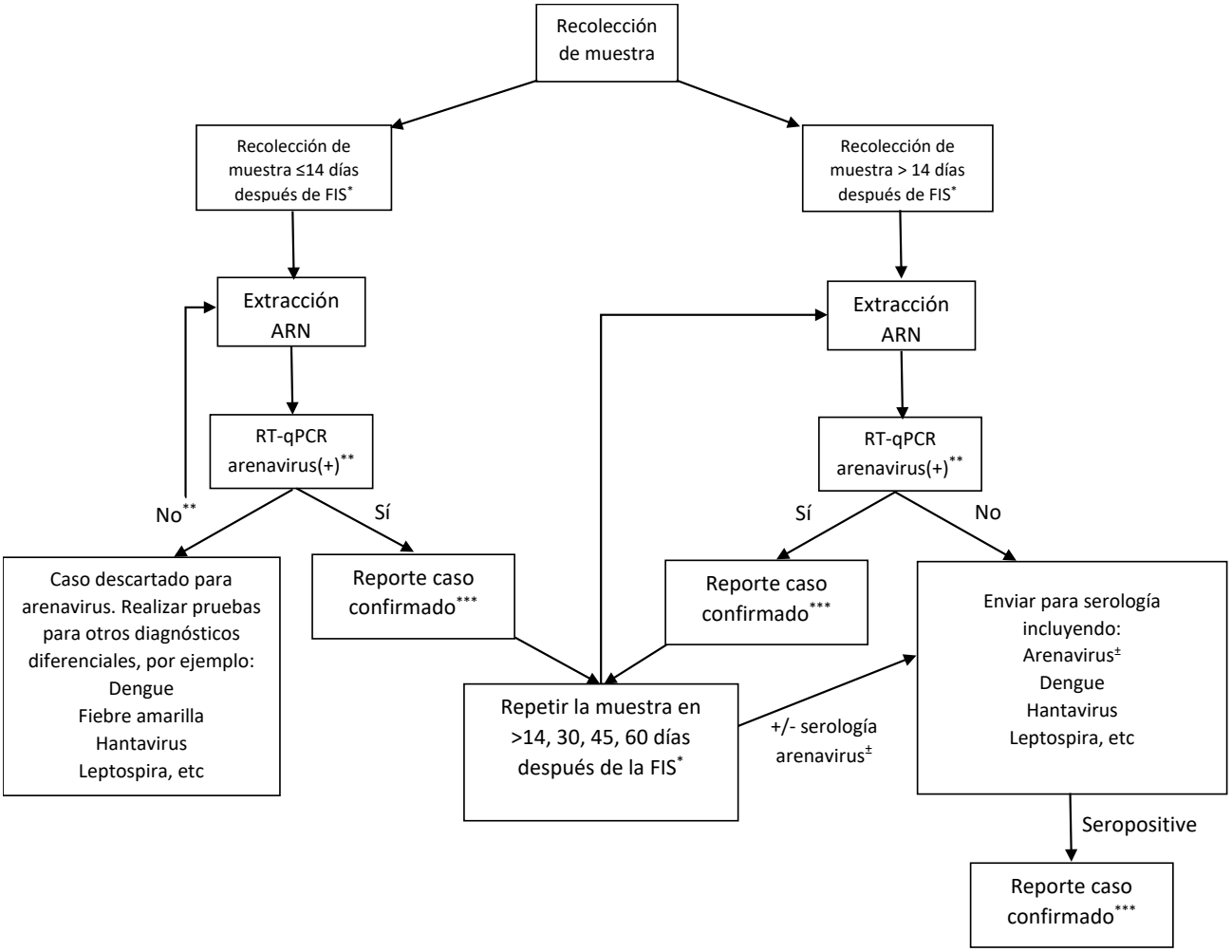


* The amount of viral RNA may be below the limit of detection if the sample was collected <3 days post-symptom onset; if negative and clinical suspicion remains, repeat sample collection >72 hours post-symptom onset.

** Report and conduct follow up investigation.

***If available.

S3.10 Figure S2b. Algoritmo para descartar Chapare u otros arenavirus del Nuevo Mundo en casos sospechosos de fiebre hemorrágica viral en un laboratorio BSL3 antes de realizar otras pruebas de laboratorio (versión en español).



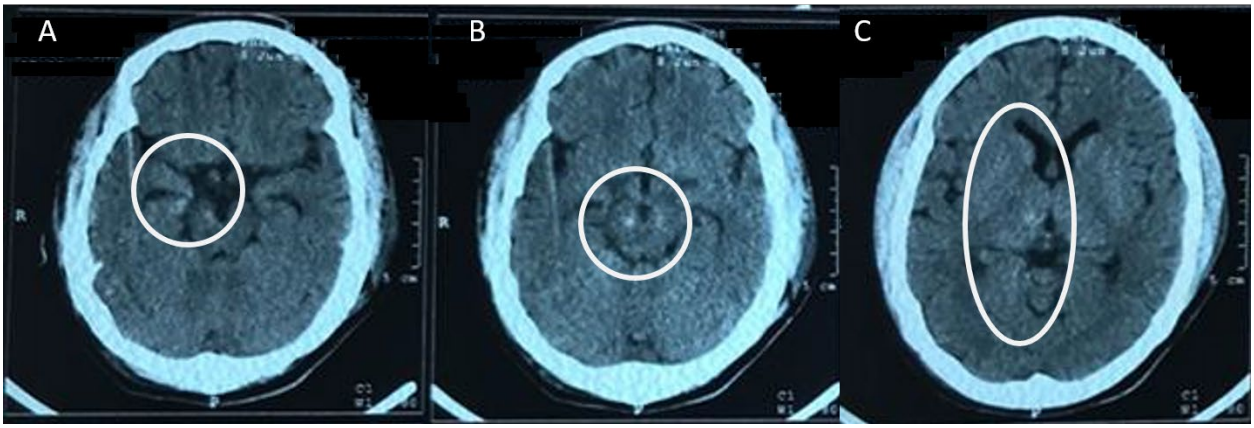
* FIS = La fecha de inicio de los síntomas.

** La cantidad de ARN viral es baja <3 días después de la FIS. Si el resultado fue negativo y persiste la sospecha clínica, repita la muestra >72 horas después de FIS.

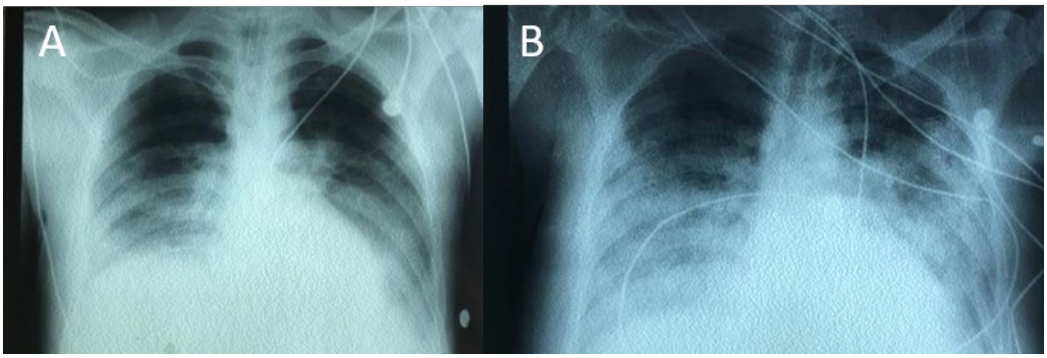
*** Acción de seguimiento para un caso de arenavirus.

± Si está disponible.

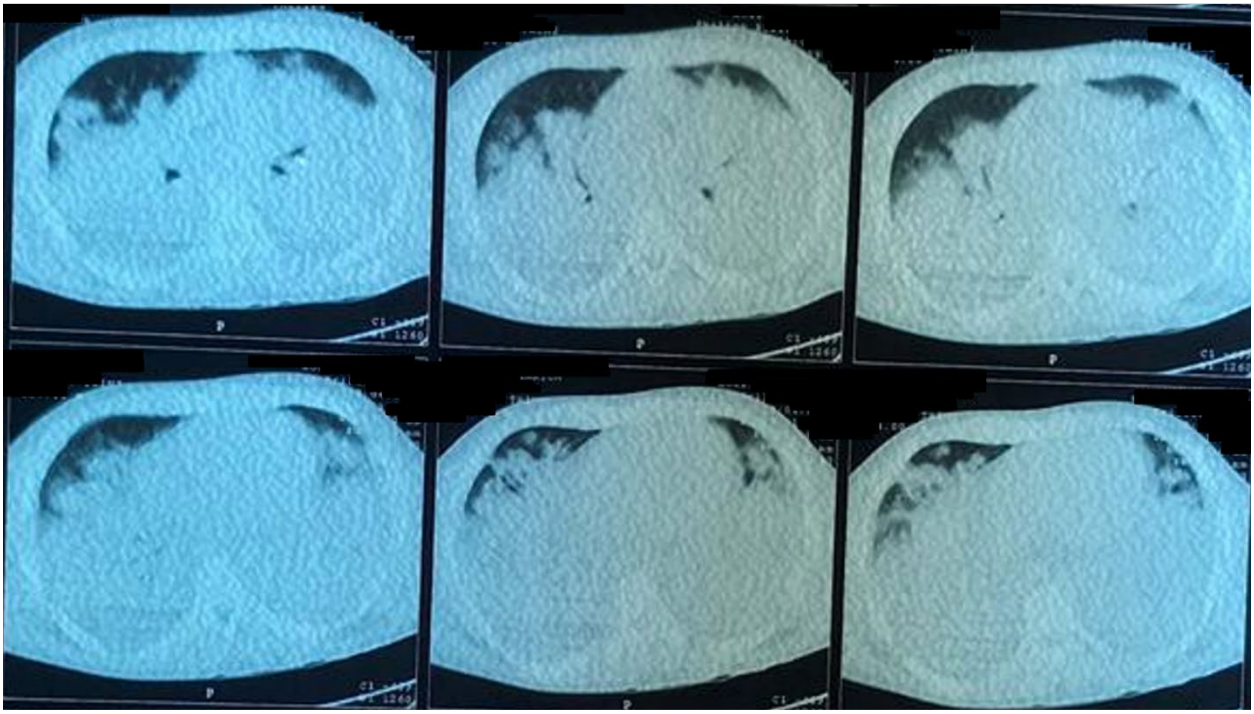
S3.11 Figure S3. Computed tomography (CT) of brain of patient S1-3 showing right-sided intracerebral hemorrhage correlating with onset of left-sided hemiparesis in Panel A) involvement of right hippocampus; B) involvement of right cerebral peduncle; C) involvement of right medial thalamus (June 8; 9 days post-symptom onset).



S3.12 Figure S4. Frontal chest radiograph of patient S1-3 showing Panel A) Bilateral inferior alveolar opacities (June 8; 9 days post-symptom onset); Panel B) Increasing lung alveolar opacities, compromising medium and lower lung fields (June 13; 14 days post-symptom onset).



S3.13 Figure S5. Computed tomography (CT) of chest of patient S1-3 revealing diffuse bilateral consolidations with gravitational gradient (sparing anterior lung zone), suggesting diffuse alveolar damage and suspected diffuse alveolar hemorrhage.



S3.14 Figure S6a. Community audience brochure for prevention of rodent-borne illnesses (English version).

What Can I Do if Someone in My Family Begins to Feel Sick?

- Visit a local clinic or hospital if you or someone in your family may have been exposed to a rodent (or their waste) and is showing signs or symptoms of a severe febrile illness spread by rodents.
- Seek care early to improve chances of survival if you become seriously ill.
- Encourage everyone in your household to practice good hand hygiene.
- Avoid having sex with someone that has signs or symptoms of a severe febrile illness spread by rodents.

Contact a healthcare provider in your community to learn more about preventing illnesses spread by rodents.

A Pan American Health Organization – U.S. Centers for Diseases Control and Prevention Collaboration

HOW TO PROTECT YOUR FAMILY

from a Severe Febrile Illness Spread by Rodents.

Learn How to Identify Illness and Reduce Exposure to Rodents In and Around Your Home.

A Pan American Health Organization – U.S. Centers for Diseases Control and Prevention Collaboration

Sick rats and mice can spread illnesses to humans. These illnesses can be severe, and sometimes fatal.

Anyone in direct contact with a rodent or their waste is at risk of becoming sick with an illness spread by rodents. Even healthy people can become sick if exposed to an infected rodent.

Rodent infestation in and around your home puts you and your family at higher risk of getting sick.

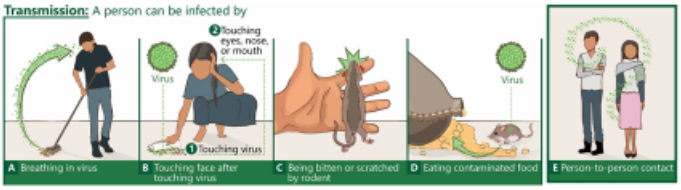
What Signs and Symptoms Should I Look For?

Fatigue · Fever · Headache · Muscle or joint aches · Nausea or stomach pain · Vomit · Diarrhea · Chills · Shortness of breath · Retro-orbital pain · Dizziness · Unexplained bleeding

How Do These Illnesses Spread to Humans?

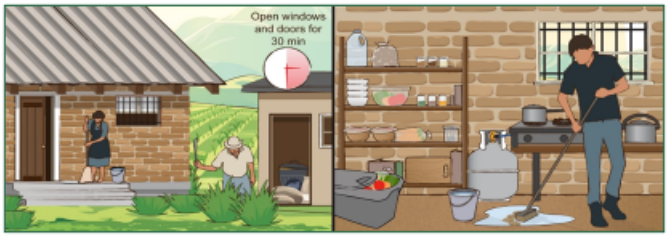
Illnesses spread by rodents are commonly spread to people through infected rodent urine, droppings, and saliva, and less frequently by a bite from an infected rodent. Some are then spread from person to person.

Transmission: A person can be infected by



How Do I Prevent Getting an Illness Spread by Rodents?

- Avoid rodents.
- Keep areas inside and around your home clean and clear.
- Open doors and windows for 30 minutes and leave the area before cleaning.
- Safely sweep regularly. Wet ground and floor areas with water before sweeping to avoid kicking up dust in the air.
- Keep trash and food scraps off the ground. Use a container with a tight lid for garbage.
- Store food in containers inside the home and in fields. Store grains in a container with a lid.
- Store household items off the ground.



S3.15 Figure S6b. Folleto para la audiencia comunitaria para la prevención de enfermedades transmitidas por roedores (versión en español).

¿Qué puedo hacer si alguien en mi familia comienza a sentirse enfermo?

- Vaya a un centro médico o un hospital si usted o alguien en su familia estuvieron expuestos a un roedor (o a los excrementos de un roedor) y muestran signos y síntomas de enfermedad grave.
- Busque atención médica pronto si se enferma gravemente.
- Anime a todos en su hogar a lavarse las manos con frecuencia.
- Evite tener relaciones sexuales con alguien que muestre signos o síntomas de enfermedad grave.

Comuníquese con un proveedor de atención médica de su comunidad para obtener más información sobre cómo prevenir las enfermedades que los roedores propagan.

Una colaboración entre la Organización Panamericana de la Salud y los Centros para el Control y la Prevención de Enfermedades de los EE. UU.

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CÓMO PROTEGER A SU FAMILIA

de las enfermedades graves que los roedores propagan

Infórmese sobre cómo identificar si alguien está enfermo y cómo reducir la exposición a roedores adentro y alrededor de su casa

Una colaboración entre la Organización Panamericana de la Salud y los Centros para el Control y la Prevención de Enfermedades de los EE. UU.

Las ratas y los ratones enfermos les pueden transmitir enfermedades a las personas. Estas enfermedades pueden ser graves y, a veces, mortales.

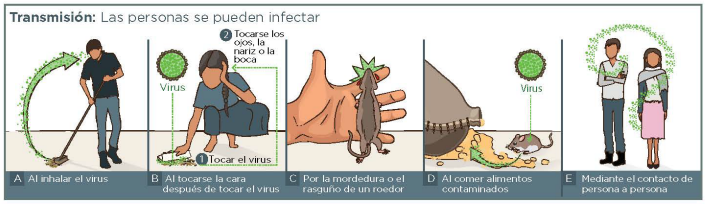
Todas las personas que entren en contacto directo con un roedor o sus excrementos, corren el riesgo de enfermarse con una las enfermedades que propagan. Incluso las personas sanas se pueden enfermar si se exponen a un roedor infectado.

Las infestaciones de roedores adentro y alrededor de la casa los pone a usted y a su familia en mayor riesgo de enfermarse.

¿A qué signos y síntomas debo estar atento?

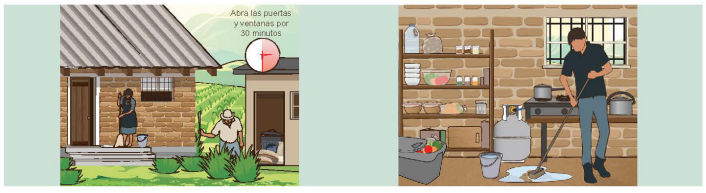
¿Cómo se transmiten estas enfermedades a las personas?

Las enfermedades que los roedores propagan se transmiten comúnmente a las personas a través de la orina, las heces, la saliva y, con menor frecuencia, la mordedura de un roedor infectado. Algunas se transmiten después de una persona a otra.

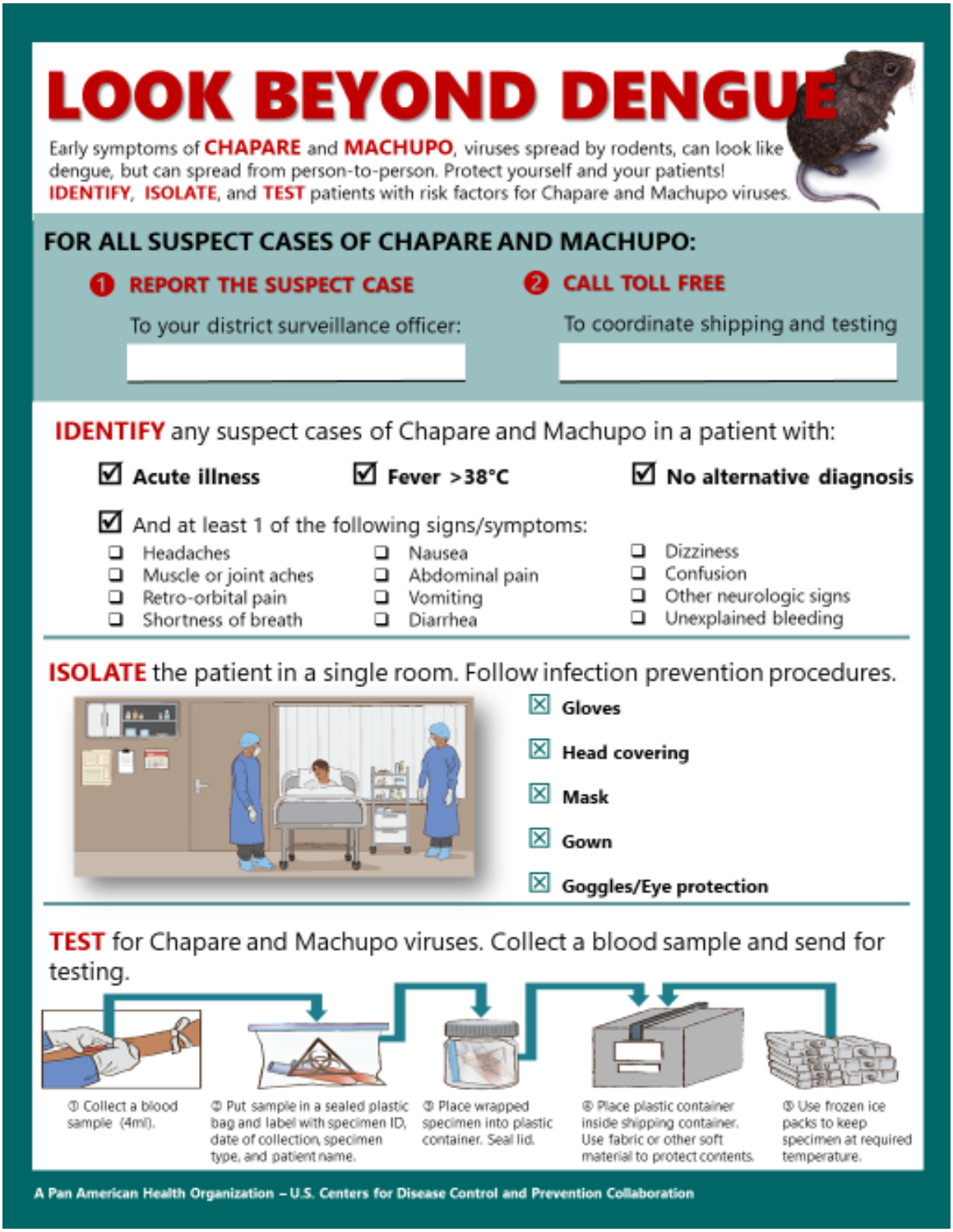


¿Cómo puedo prevenir contagiarme una de las enfermedades que los roedores propagan?

- Evite a los roedores.
- Mantenga las áreas de adentro y alrededor de su casa limpias y despejadas.
- Antes de limpiar un área, abra las puertas y ventanas y aléjese del área por 30 minutos.
- Barra con regularidad y hágalo de manera segura. Moje el suelo y los pisos con agua antes de barrerlos para no levantar polvo.
- No permita que queden restos de comida y basura sobre el suelo. Use un recipiente con tapa que cierre bien para la basura.
- Tanto adentro de la casa como afuera en el campo. Almacene los granos en recipientes con tapa.
- No almacene artículos del hogar sobre el suelo.



S3.16 Figure S7a. Healthcare facility poster to raise awareness to "look beyond dengue" and identify, isolate, and test for Chapare and Machupo viruses in all suspect cases (English version).



S3.17 Figure S7b. Póster para que los médicos creen conciencia para "mirar más allá del dengue" e identificar, aislar y realizar pruebas para los virus Chapare y Machupo en todos los casos sospechosos (versión en español).

MIRE MÁS ALLÁ DEL DENGUE

Los síntomas tempranos de infección por los virus de **CHAPARE** y **MACHUPO** que transmiten los roedores pueden ser similares a los del dengue, pero estos virus pueden transmitirse de persona a persona. ¡Protéjase y proteja a sus pacientes! **IDENTIFIQUE**, **AÍSLE** y **HÁGALES PRUEBAS** a los pacientes con factores de riesgo de contraer los virus de Chapare y Machupo.

EN TODOS LOS CASOS PRESUNTOS DE FIEBRE DE CHAPARE Y MACHUPO:

1 NOTIFIQUE EL CASO PRESUNTO al funcionario de vigilancia de su distrito: _____

2 LLAME AL NÚMERO GRATUITO para coordinar el envío y la prueba _____

IDENTIFIQUE todo caso presunto de fiebre de Chapare y Machupo de pacientes:

☐ **con enfermedad aguda** ☐ **con fiebre >38 °C** ☐ **sin diagnóstico alternativo**

☐ **Y al menos uno de los siguientes signos o síntomas:**

- ❑ Dolores de cabeza
- ❑ Dolores musculares o articulares
- ❑ Dolor retroorbital
- ❑ Dificultad para respirar
- ❑ Náuseas
- ❑ Dolor abdominal
- ❑ Vómitos
- ❑ Diarrea
- ❑ Mareos
- ❑ Confusión
- ❑ Otros signos neurológicos
- ❑ Sangrado sin causa aparente

AÍSLE al paciente en una habitación individual. Siga los procedimientos de prevención de infecciones.

- ☐ **Guantes**
- ☐ **Protección para la cabeza**
- ☐ **Mascarilla**
- ☐ **Bata**
- ☐ **Gafas u otra protección para los ojos**

HAGA PRUEBAS de detección de los virus de Chapare y Machupo. Tome una muestra de sangre y envíela a analizar.

1. Tome una muestra de sangre (4 ml).
2. Coloque la muestra en una bolsa plástica, séllela y etiquétela con el tipo y núm. de ID del espécimen, la fecha de obtención y el nombre del paciente.
3. Coloque el espécimen envuelto adentro de un envase de plástico. Cierre la tapa.
4. Coloque el envase de plástico dentro del paquete de envío. Use tela u otro material suave para proteger el contenido.
5. Use paquetes de hielo para mantener el espécimen a la temperatura requerida.

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Colaboración entre la Organización Panamericana de la Salud y los Centros para el Control y la Prevención de Enfermedades de los EE. UU.

S4 Supplemental recommendations for survivors (English and Spanish versions)

S4.1 Limiting Spread of Chapare Virus: Interim Considerations for Management of Recent Chapare Hemorrhagic Fever Patients and Survivors

Chapare virus can be transmitted from one person to another through contact with an infected person's body fluids, such as blood, urine, respiratory secretions, saliva, and semen. The risk of transmission may be increased during aerosol-generating procedures such as chest compressions or intubation. When managing a patient who is recovering from Chapare hemorrhagic fever, there are important considerations for monitoring viral shedding and protecting healthcare workers and caretakers from being inadvertently infected through contact with survivors while they may still be infectious. Chapare virus may remain in body fluids for days, weeks, or months after a person recovers from infection and their symptoms have resolved. Data on how long a survivor can spread the virus is limited. The length of time may vary between survivors and different types of body fluids.

The following interim recommendations are based on the current understanding of Chapare hemorrhagic fever, as well as other viral hemorrhagic fevers, and may change over time as more information becomes available. It is important for body fluids of all survivors to be regularly monitored, and until negative, precautions should be used to limit the risk of human-to-human transmission, even after symptoms have resolved.

S4.1.1 Survivor Monitoring and Precautions

Clinical samples (for example blood, saliva, urine, and semen when applicable) from survivors should be collected at least once per week after clinical signs have resolved. Samples should be tested using real-time reverse-transcriptase polymerase chain reaction (rRT-PCR).

If a patient has clinically recovered, the hospital team may consider discharging the patient from the hospital even if samples from the patient remain positive by rRT-PCR. A discharge plan should be discussed with the survivor and their family, which would include discussion of follow-up appointments, monitoring, and appropriate precautions. The following precautions should be continued until the survivor has cleared their infection. The precautions described below should be continued until Chapare virus RNA is no longer detectable by rRT-PCR in two consecutive samples collected at least 48 hours apart.

S4.1.2 If Chapare virus RNA is still detectable in any body fluid samples from a survivor:

- A local hospital should be identified in their home village or municipality in case there is need for urgent medical attention or follow up sample collection.
- If the survivor needs medical attention before they have cleared their infection, they should inform the hospital prior to arrival that they are a survivor of Chapare hemorrhagic fever and may still be infectious.

S4.1.2.1 If Chapare virus RNA is still detectable in urine:

- If possible, the survivor should use a separate bathroom from the rest of the people living in the home.
- Both men and women should sit down to urinate in order to minimize splashing. The lid of the

toilet should be closed before flushing the toilet, to minimize the chances that the contaminated water is splashed outside of the bowl.

- Wash hands immediately with soap and water after going to the bathroom.

S4.1.2.2 Chapare virus RNA is still detectable in semen:

- The survivor should practice abstinence or use male condoms during any sexual activity (vaginal, anal, oral, or manual)
- The survivor should be the only one to remove the condom and dispose of it in the toilet or in a container that contains disinfectant (for example, bleach or Clorox). After touching used condoms or semen, wash hands and genitals with soap and water.

S4.1.3 Precautions for Survivor Cohabitants and Caretakers

- Clean and disinfect surfaces and objects touched by the survivor (like door knobs, toilets, toilet or sink handles, light switches, counter tops, tables, kitchenware or silverware, children's toys, etc)
    - Use disinfectant products such as bleach or Clorox, especially in bathrooms.
- Wear disposable gloves when cleaning toilets and other surfaces that may have been contaminated with the survivor's body fluids (such as blood, urine, saliva, respiratory secretions, or semen)
- Wear disposable gloves when handling clothing, bedding, towels, or other linens that may have been soiled with the survivor's body fluids. Wash separately using hot water and detergent.
- Take off gloves immediately after handling any materials that may have been contaminated and throw them in the trash. Do not reuse contaminated gloves. Immediately wash hands after removing used gloves.
- If surfaces are visibly dirty, use disposable gloves to clean the surface with paper towels and discard them in the trash, then disinfect the surfaces. After disposing of the gloves in the trash, immediately wash hands.

S4.1.4 Special Considerations for Pediatric Survivors

- In general, recommendations for management of pediatric survivors of Chapare virus infection are the same as for adults.
- However, children may be more likely to contaminate household surfaces and objects through saliva and urine than adults. Family members and caretakers should avoid contact with potentially contaminated items like clothing, bedding, food and cookware, and toys.
- The hospital team may consider waiting to discharge a pediatric case until they test negative by rRT-PCR in two consecutive saliva and urine samples collected at least 48 hours apart.

S4.1.5 Special Considerations for Pregnant Survivors

In addition to the general recommendations for management of survivors of Chapare hemorrhagic fever, there are special considerations for women who become infected during pregnancy and recover from infection. The uterus is an immune-privileged site (the immune system may not clear the virus in the uterus in the same way that it can clear the virus from the rest of the body), so there is a potential risk of spreading Chapare virus through contact with body fluids of the patient during labor (vaginal secretions, amniotic fluid, placenta, and the fetus), even if the blood of the patient has previously tested negative for Chapare virus. This

has not been documented with Chapare virus, but it has been documented in other viral hemorrhagic fevers like Lassa fever and Ebola virus disease. Because of this risk, there are important considerations and recommended precautions before, during, and after birth to prevent the spread of Chapare virus.

S4.1.5.1 Signs of miscarriage or labor

Pregnant survivors should seek medical attention immediately at the first sign of labor or possible miscarriage (such as cramping/abdominal pain or vaginal bleeding). If possible, specimens (such as swabs of blood or vaginal fluid) should be immediately collected at the earliest sign of labor and tested for Chapare virus to provide information to the healthcare team on the risk of transmission of the virus. Anyone who has contact with the patient and baby should use proper biosafety precautions to avoid contact with infected body fluids.

S4.1.5.2 Labor precautions

Biosecurity (airborne) precautions should be carefully planned to reduce the risk of transmission to healthcare workers involved in the labor process, whether delivery is by cesarean section or by natural birth. The labor should only take place in a hospital with adequate biosecurity capabilities to prevent transmission of the virus to healthcare workers. Precautions such as admitting the patient to the hospital prior to estimated delivery date should be planned to prevent a home delivery.

S4.1.5.3 Testing and care of the baby after birth

It is possible that the baby of a pregnant survivor may become infected during pregnancy or may be exposed to the virus during labor and become infected and develop Chapare hemorrhagic fever after birth. A blood specimen from the baby should be collected as soon as possible after birth and tested for Chapare virus by rRT- PCR. An additional sample should be collected and tested at approximately 72 hours after birth. Only after testing negative by rRT-PCR should the baby be considered free of Chapare virus infection. Until then, the baby should be considered potentially infectious and all healthcare providers or caretakers who have contact with the baby should use proper biosafety precautions to avoid contact with potentially infected body fluids. If the baby appears to be ill in any way within 3 weeks following birth, a sample should be immediately collected and tested for Chapare virus.

Certain procedures, such as bulb suctioning or suctioning for meconium, and general cleaning of the baby following birth, may put hospital staff at increased risk and biosafety (contact and droplet) precautions should be used to prevent exposure to potentially infected materials. Additionally, in the case of meconium aspiration or other causes of respiratory distress, intubation of the baby may become necessary. If intubation of the baby becomes necessary, airborne precautions should be used as this is a potentially aerosol-generating procedure.

S4.1.5.4 Breastfeeding

Because the mammary glands (breasts) are immune-privileged sites, there is a risk of transmission of Chapare virus through breast milk, even if the mother has tested negative for Chapare virus in other body fluids. Prior to breastfeeding, a sample of colostrum and breast milk should be collected and tested for Chapare virus by rRT- PCR. Breastfeeding should only occur after testing negative by rRT-PCR in two consecutive breastmilk samples collected at least 48 hours apart.

S4.2 Limitación de la propagación del virus Chapare: Consideraciones provisionales para el manejo de pacientes y sobrevivientes de fiebre hemorrágica por virus Chapare

El virus Chapare se puede transmitir de una persona a otra a través del contacto con los fluidos corporales de una persona infectada, como sangre, orina, secreciones respiratorias, saliva y semen. El riesgo de transmisión aumenta durante los procedimientos que generan aerosoles, como las compresiones torácicas o la intubación. Al atender a un paciente que se está recuperando de la fiebre hemorrágica por virus Chapare, hay consideraciones importantes para monitorear la persistencia viral; así como para proteger a los trabajadores de la salud y los cuidadores de ser infectados inadvertidamente a través del contacto con sobrevivientes mientras estos mismos podrían aún ser infecciosos. El virus Chapare puede persistir en los fluidos corporales durante días, semanas o meses después de que una persona se ha recuperado de la infección y sus síntomas se han resuelto. Los datos sobre cuánto tiempo un sobreviviente puede propagar el virus son limitados. El período de tiempo puede variar entre los sobrevivientes y los diferentes tipos de fluidos corporales.

Las siguientes recomendaciones provisionales se basan en los datos disponibles a la fecha respecto a la fiebre hemorrágica por virus Chapare, así como otras fiebres hemorrágicas virales, y podrían cambiar con el tiempo a medida que se disponga de más información. Es importante que los fluidos corporales de todos los sobrevivientes sean monitoreados regularmente, y hasta que estos mismos sean negativos, se debe tomar precauciones para limitar el riesgo de transmisión de persona a persona, incluso después de la recuperación.

S4.2.1 Monitoreo y precauciones para sobrevivientes

Las muestras clínicas (por ejemplo, sangre, saliva, orina y semen cuando corresponda) de los sobrevivientes deben recolectarse al menos una vez por semana después de la recuperación. Las muestras deben ser analizadas usando la reacción en cadena de la polimerasa con transcriptasa inversa en tiempo real (RT-qPCR)

Si un paciente se ha recuperado clínicamente, el equipo médico hospitalario puede considerar dar de alta al paciente, aún si las muestras del paciente siguen siendo positivas por RT-qPCR. Se debe discutir un plan de alta con el sobreviviente y su familia, que debería incluir recomendaciones de citas de seguimiento, monitoreo, precauciones apropiada, consejería y soporte psicosocial. Se recomienda tomar las precauciones presentadas a continuación, hasta que el sobreviviente haya eliminado la infección. Por lo tanto, estas precauciones se deben seguir hasta que el ARN del virus Chapare ya no sea detectable por RT-qPCR en dos muestras consecutivas recolectadas en un intervalo de al menos 48 horas.

S4.2.2 Si el ARN del virus Chapare es detectable en muestras de fluidos corporales de un sobreviviente:

- Se debe identificar un hospital local en su aldea o municipio de origen en caso de que sea necesario recibir atención médica urgente o repetir la recolección de muestras.
- Si el sobreviviente necesita atención médica antes de que haya totalmente curado su infección, debe informarse al hospital antes de su llegada que es un sobreviviente de la fiebre hemorrágica de virus Chapare y que aún podría ser infeccioso.

S4.2.2.1 Si el ARN del virus Chapare es detectable en la orina:

- Si es posible, el sobreviviente debe usar un baño separado de las otras personas que habitan en la misma vivienda.
- Tanto hombres como mujeres deben sentarse a orinar para minimizar las salpicaduras. La tapa del

inodoro debe cerrarse antes de tirar el agua, para minimizar las posibilidades de que el agua contaminada salpique fuera de la taza.
- Mantener a mano y colocar desinfectante después de usar el inodoro.
- Lávese las manos inmediatamente con agua y jabón después de usar al baño.

S4.2.2.2 Si el ARN del virus Chapare es detectable en el semen:

- El sobreviviente debe practicar la abstinencia o usar condones masculinos durante la actividad sexual (vaginal, anal, oral o manual)
- El sobreviviente debe ser la única persona que se quite el condón y lo deseche en un recipiente que contenga desinfectante (por ejemplo, lejía o Clorox). Después de tocar condones o semen usados, lávese las manos y los genitales con agua y jabón.

S4.2.3 Precauciones para los convivientes y cuidadores de los sobrevivientes

- Limpiar y desinfectar superficies y objetos tocados por el sobreviviente (como pomos de puertas, inodoros, manijas de inodoros o lavabos, interruptores de luz, superficies, mesas, utensilios de cocina o cubiertos, juguetes para niños, etc.)
    - Use productos desinfectantes como Clorox, especialmente en baños.
- Use guantes desechables cuando limpie los inodoros y otras superficies que puedan haber sido contaminadas con los fluidos corporales del sobreviviente (como sangre, orina, saliva, secreciones respiratorias o semen).
- Use guantes desechables cuando toque la ropa, la ropa de cama, las toallas u otras sábanas que puedan haber sido contaminadas con los fluidos corporales del sobreviviente. Lavar por separado con agua caliente y detergente.
- Quítese los guantes inmediatamente después de manipular cualquier material que pueda haber sido contaminado y tírelos a la basura. No reutilice los guantes contaminados. Lávese las manos inmediatamente después de quitarse los guantes usados.
- Si las superficies están visiblemente sucias, use guantes desechables para limpiar la superficie con toallas de papel y deséchelas en la basura, luego desinfecte las superficies. Después de tirar los guantes a la basura, lávese las manos inmediatamente.

S4.2.4 Consideraciones especiales para sobrevivientes pediátricos

- En general, las recomendaciones para el manejo de los niños sobrevivientes de la infección por el virus del Chapare son las mismas que para los adultos.
- Los niños tienen más probabilidades de contaminar las superficies y los objetos domésticos con de la saliva y la orina que los adultos.
- Los familiares y cuidadores deben evitar el contacto con artículos potencialmente contaminados como ropa, ropa de cama, alimentos y utensilios de cocina y juguetes.

S4.2.5 Consideraciones especiales para las sobrevivientes embarazadas

Además de las recomendaciones generales para el manejo de los sobrevivientes de la fiebre hemorrágica por virus Chapare, existen consideraciones especiales para las mujeres que se infectan durante el embarazo y se recuperan de la infección. El útero es un sitio de privilegio inmunológico (es posible que el sistema inmunológico no elimine al virus en el útero de la misma manera que puede eliminar al virus del resto del cuerpo), por lo que existe un riesgo potencial de propagar el virus Chapare a través del contacto con los fluidos corporales de la paciente durante el trabajo de parto (secreciones vaginales, fluido amniótico, placenta

y feto), incluso si la sangre de la paciente previamente dio negativo para el virus del Chapare. Esto no se ha documentado con el virus Chapare, pero se ha documentado en otras fiebres hemorrágicas virales como la fiebre de Lassa y enfermedad por el virus del Ébola. Debido a este riesgo, existen consideraciones importantes y precauciones recomendadas antes, durante y después del nacimiento para prevenir la propagación del virus Chapare.

S4.2.5.1 Signos de aborto espontáneo o trabajo de parto

Las sobrevivientes embarazadas deben buscar atención médica inmediata a la primera señal de trabajo de parto o posible aborto espontáneo (como calambres / dolor abdominal o sangrado vaginal). Si es posible, las muestras (como hisopos de sangre o fluido vaginal) deben recolectarse inmediatamente a la primera señal de trabajo de parto y analizarse para detectar el virus Chapare y brindar información al equipo de atención médica sobre el riesgo de transmisión del virus. Cualquiera que tenga contacto con la paciente y el bebé debe tomar las precauciones de bioseguridad adecuadas para evitar el contacto con fluidos corporales potencialmente infectados.

S4.2.5.2 Precauciones durante la labor de parto

Las precauciones de bioseguridad (incluyendo infección por aerosoles) deben planificarse cuidadosamente para reducir el riesgo de transmisión a los trabajadores de salud involucrados en el proceso de parto, ya sea que el parto sea por cesárea o por parto natural. El trabajo de parto solo debe realizarse en un hospital con la capacidad de bioseguridad adecuada para evitar la transmisión del virus a los trabajadores de salud. Se debe planificar medidas de precaución tales como admitir a la paciente en el hospital antes de la fecha estimada de parto para evitar un parto a domicilio.

S4.2.5.3 Pruebas y cuidados del bebé después del nacimiento.

Es posible que el bebé de una sobreviviente embarazada se infecte durante el embarazo o se exponga al virus durante el trabajo de parto y se infecte y desarrolle fiebre hemorrágica del Chapare después del nacimiento. Se debe recolectar una muestra de sangre del bebé tan pronto como sea posible después del nacimiento y analizarla para detectar el virus Chapare mediante RT-qPCR. Se debe recolectar y analizar una muestra adicional aproximadamente 72 horas después del nacimiento. Solo después de dar negativo en la prueba RT-qPCR se debe considerar que el bebé está libre de infección por el virus Chapare. Hasta entonces, el bebé debe considerarse potencialmente infeccioso y todos los proveedores de atención médica o cuidadores que tengan contacto con el bebé deben tomar las precauciones de bioseguridad adecuadas para evitar el contacto con fluidos corporales potencialmente infectados. Si el bebé parece estar enfermo de alguna manera dentro de las 3 semanas posteriores al nacimiento, se debe recolectar una muestra de inmediato y analizarla para detectar el virus Chapare.

Ciertos procedimientos, como la succión con bulbo o la succión de meconio y la limpieza general del bebé después del nacimiento, pueden poner al personal hospitalario en mayor riesgo y se recomienda tomar precauciones de bioseguridad (contacto y gotitas) para evitar la exposición a materiales potencialmente infectados. Además, en el caso de aspiración de meconio u otras causas de dificultad respiratoria, puede ser necesaria la intubación del bebé. Si es necesaria la intubación del bebé, se debe tomar precauciones de transmisión por aire, ya que se trata de un procedimiento potencialmente generador de aerosoles.

S4.2.5.4 Lactancia materna

Debido a que las glándulas mamarias (senos) son sitios de privilegio inmune, existe el riesgo de transmisión del virus Chapare a través de la leche materna, incluso si la madre ha dado negativo en la prueba del virus Chapare en otros fluidos corporales. Antes de amamantar, se debe recolectar una muestra de calostro y leche materna y analizar el virus Chapare mediante RT-qPCR. La lactancia materna solo debe ocurrir después de que

la prueba de RT-qPCR sea negativa en dos muestras consecutivas de leche materna recolectadas con al menos 48 horas de diferencia.