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Version 1

## nCoV-2019 sequencing protocol V.1



Forked from [Ebola virus sequencing protocol](#)

DOI

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**We use this protocol and it's working**

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## Abstract





ARTIC amplicon sequencing protocol for MinION for nCoV-2019

## Troubleshooting



## cDNA preparation

- 1 Mix the following components in an 0.2mL 8-strip tube;

Component	Volume
50µM random hexamers	 1 µL
10mM dNTPs mix (10mM each)	 1 µL
Template RNA	 11 µL
<b>Total</b>	 13 µL

### Note


Viral RNA input from a clinical sample should be between Ct 18-35. If Ct is between 12-15, then dilute the sample 100-fold in water, if between 15-18 then dilute 10-fold in water. This will reduce the likelihood of PCR-inhibition.


### Note

A mastermix should be made up in the **mastermix cabinet** and aliquoted into PCR strip tubes. Tubes should be wiped down when entering and leaving the mastermix cabinet.




- 2 Gently mix by pipetting and pulse spin the tube to collect liquid at the bottom of the tube.


- 3 Incubate the reaction as follows:

 65 °C for  00:05:00

Place on ice for  00:01:00

- 4 Add the following to the annealed template RNA:

Component	Volume
SSIV Buffer	 4 µL
100mM DTT	 1 µL
RNaseOUT RNase Inhibitor	 1 µL

SSIV Reverse Transcriptase  1  $\mu\text{L}$ **Total** 20  $\mu\text{L}$ **Note**

A mastermix should be made up in the **mastermix cabinet** and added to the denatured RNA in the **extraction and sample addition cabinet**. Tubes should be wiped down when entering and leaving the mastermix cabinet.

5 Gently mix by pipetting and pulse spin the tube to collect liquid at the bottom of the tube.



6 Incubate the reaction as follows:



 42 °C  00:50:00 70 °C  00:10:00Hold at  5 °C**Primer pool preparation**

7 If required resuspend lyophilised primers at a concentration of 100 $\mu\text{M}$  each

**Note**

**nCov-2019/V1** primers for this protocol were designed using **Primal Scheme** and generate overlapping 400nt amplicons. Primer names and dilutions are listed in the table below.

8 Generate primer pool stocks by adding  5  $\mu\text{L}$  of each primer pair to a  1.5 mL Eppendorf labelled either "Pool 1 (100 $\mu\text{M}$ )" or "Pool 2 (100 $\mu\text{M}$ )". Total volume should be

 490  $\mu\text{L}$  for Pool 1 (100 $\mu\text{M}$ ) and  490  $\mu\text{L}$  for Pool 2 (100 $\mu\text{M}$ ). These are your 100 $\mu\text{M}$  stocks of each primer pool.

#### Note

Primers should be diluted and pooled in the **mastermix cabinet** which should be cleaned with decontamination wipes and UV sterilised before and after use.













- 9 Dilute this primer pool 1:10 in molecular grade water, to generate 10 $\mu\text{M}$  primer stocks. It is recommended that multiple aliquots of each primer pool are made to in case of degradation or contamination.

#### Note

Primers need to be used at a final concentration of 0.015 $\mu\text{M}$  per primer. In this case both pools have 98 primers in so the requirement is 3.6 $\mu\text{L}$  primer pools (10 $\mu\text{M}$ ) per 25 $\mu\text{L}$  reaction. For other schemes, adjust the volume added appropriately.

## Multiplex PCR

- 10 In the mastermix hood set up the multiplex PCR reactions as follows in 0.2mL 8-strip PCR tubes:

Component	Pool 1	Pool 2
5X Q5 Reaction Buffer	 5 $\mu\text{L}$	 5 $\mu\text{L}$
10 mM dNTPs	 0.5 $\mu\text{L}$	 0.5 $\mu\text{L}$
Q5 Hot Start DNA Polymerase	 0.25 $\mu\text{L}$	 0.25 $\mu\text{L}$
Primer Pool 1 or 2 (10 $\mu\text{M}$ )	 3.6 $\mu\text{L}$	 3.6 $\mu\text{L}$
Nuclease-free water	 13.15 $\mu\text{L}$	 13.15 $\mu\text{L}$
<b>Total</b>	 22.5 $\mu\text{L}$	 22.5 $\mu\text{L}$

**Note**

A PCR mastermix for each pool should be made up in the **mastermix cabinet** and aliquoted into PCR strip tubes. Tubes should be wiped down when entering and leaving the mastermix cabinet.








- 11 In the **extraction and sample addition cabinet** add  2.5 µL cDNA to each tube and mix well by pipetting.

**Note**

The **extraction and sample addition cabinet** should be cleaned with decontamination wipes and UV sterilised before and after use.

- 12 Pulse centrifuge the tubes to collect the contents at the bottom of the tube.


- 13 Set-up the following program on the thermal cycler:

Step	Temperature	Time	Cycles
Heat Activation	 98 °C	 00:00:30	1
Denaturation	 98 °C	 00:00:15	25-35
Annealing	 65 °C	 00:05:00	25-35
Hold	 4 °C	Indefinite	1

**Note**

Cycle number should be 25 for Ct 18-21 up to a maximum of 35 cycles for Ct 35

## PCR clean-up

- 14 Combine the entire contents of "Pool 1" and "Pool 2" PCR reactions for each biological sample into to a single  1.5 mL Eppendorf tube.
- 15 Clean-up the amplicons using the following protocol:



## Protocol

NAME

## Amplicon clean-up using SPRI beads

CREATED BY

Josh Quick



[Preview](#)

## Note


Amplicon clean-up should be performed in the **post-PCR cabinet** which should be cleaned with decontamination wipes and UV sterilised before and after use.


- 15.1 Vortex SPRI beads thoroughly to ensure they are well resuspended, the solution should be a homogenous brown colour.

 Agencourt AMPure XP **Beckman Coulter Catalog #A63880**

- 15.2 Add an equal volume (1:1) of SPRI beads to the sample tube and mix gently by either flicking or pipetting. For example add  50 µL SPRI beads to a  50 µL reaction.

- 15.3 Pulse centrifuge to collect all liquid at the bottom of the tube.










- 15.4 Incubate for  00:05:00 at room temperature.

- 15.5 Place on magnetic rack and incubate for  00:02:00 or until the beads have pelleted and the supernatant is completely clear.

- 15.6 Carefully remove and discard the supernatant, being careful not to touch the bead pellet.





- 15.7 Add  200  $\mu$ L of room-temperature  70 % volume ethanol to the pellet.
- 15.8 Carefully remove and discard ethanol, being careful not to touch the bead pellet.
- 15.9  go to step #15.7 and repeat ethanol wash.
- 15.10 Pulse centrifuge to collect all liquid at the bottom of the tube and carefully remove as much residual ethanol as possible using a P10 pipette.
- 15.11 With the tube lid open incubate for  00:01:00 or until the pellet loses it's shine (if the pellet dries completely it will crack and become difficult to resuspend).
- 15.12 Resuspend pellet in  30  $\mu$ L Elution Buffer (EB), mix gently by either flicking or pipetting and incubate for  00:02:00 .  
 Elution Buffer (EB) **Qiagen Catalog #19086**
- 15.13 Place on magnet and transfer sample to a clean 1.5mL Eppendorf tube ensuring no beads are transferred into this tube.
- 15.14 Quantify  1  $\mu$ L product using the Quantus Fluorometer using the ONE dsDNA assay.  
 QuantiFluor(R) ONE dsDNA System, 100rxn **Promega Catalog #E4871**





### Equipment

Quantus

NAME

Fluorometer

TYPE

Promega

BRAND

E6150

SKU

<https://www.promega.co.uk/products/microplate-readers-fluorometers-luminometers/fluorometers/quantus-fluorometer>

LINK

## Quantification and normalisation

- 16 Quantify the amplicon pools using the Quantus Fluorometer using the ONE dsDNA assay.

### Protocol

NAME

**DNA quantification using the Quantus fluorometer**

CREATED BY








Josh Quick

Preview

### Note


If the concentration is greater than 25 ng/ $\mu$ L dilute the sample by a factor of 10 by adding 270 $\mu$ L 10mM Tris and quantify again using the Quantus fluorometer.




- 16.1 Remove Lambda DNA 400 ng/ $\mu$ L standard from the freezer and leave on ice to thaw. Remove ONE dsDNA dye solution from the fridge and allow to come to room temperature.
-  QuantiFluor(R) ONE dsDNA System, 500rxn **Promega Catalog #E4870**
- 16.2 Set up two  0.5 mL tubes for the calibration and label them 'Blank' and 'Standard'
- 16.3 Add  200  $\mu$ L ONE dsDNA Dye solution to each tube.
- 16.4 Mix the Lambda DNA standard 400 ng/ $\mu$ L standard by pipetting then add  1  $\mu$ L to one of the standard tube.
- 16.5 Mix each sample vigorously by vortexing for  00:00:05 and pulse centrifuge to collect the liquid.
- 16.6 Allow both tubes to incubate at room temperature for  00:02:00 before proceeding.
- 16.7 Selection 'Calibrate' then 'ONE DNA' then place the blank sample in the reader then select 'Read Blank'. Now place the standard in the reader and select 'Read Std'.
- 16.8 Set up the required number of  0.5 mL tubes for the number of DNA samples to be quantified.

**Note**


Use only thin-wall, clear, 0.5mL PCR tubes such as Axygen #PCR-05-C

- 16.9 Label the tubes on the lids, avoid marking the sides of the tube as this could interfere with the sample reading.
- 16.10 Add  199  $\mu$ L ONE dsDNA dye solution to each tube.

16.11 Add  1  $\mu\text{L}$  of each user sample to the appropriate tube.

**Note**

Use a P2 pipette for highest accuracy.


16.12 Mix each sample vigorously by vortexing for  00:00:05 and pulse centrifuge to collect the liquid.

16.13 Allow all tubes to incubate at room temperature for  00:02:00 before proceeding.

16.14 On the Home screen of the Quantus Fluorometer, select 'Protocol', then select 'ONE DNA' as the assay type.

**Note**

If you have already performed a calibration for the selected assay you can continue, there is no need to perform repeat calibrations when using ONE DNA pre diluted dye solution. If you want to use the previous calibration, skip to step 11. Otherwise, continue with step 9.

16.15 On the home screen navigate to 'Sample Volume' and set it to  1  $\mu\text{L}$  then 'Units' and set it to ng/ $\mu\text{L}$ .

16.16 Load the first sample into the reader and close the lid. The sample concentration is automatically read when you close the lid.



16.17 Repeat step 16 until all samples have been read.

16.18 The value displayed on the screen is the dsDNA concentration in ng/ $\mu\text{L}$ , carefully record all results in a spreadsheet or laboratory notebook.

17 Label a  1.5 mL Eppendorf tube for each sample.

**Note**

This is a 'one-pot ligation' protocol for native barcoded ligation libraries. We have seen no reduction in performance compared to standard libraries, and is made faster by using the Ultra II® ligation module which is compatible with the Ultra II® end repair/dA-tailing module removing a clean-up step.

- 18 Normalise the input by diluting each sample to [M] 1 ng/μL . Use  5 μL input for the One-pot native barcoding reaction to give a total of  5 ng per sample.

**Note**

Input to the one-pot native barcoding reaction will vary depending on the amplicon length but we have determined 5ng is the correct input for efficient barcoding of this amplicon length. Process at least 7 samples plus one negative control per native barcoded library in order to have sufficient material at the end.

## Native barcoding

- 19 Barcode the amplicon pools using native barcodes.

**Protocol**

NAME

**One-pot native barcoding of amplicons**

CREATED BY

Josh Quick

**Preview**

- 19.1 Set up the following reaction for each sample:

**Component****Volume**

DNA amplicons

5 µL

Nuclease-free water

7.5 µL

Ultra II End Prep Reaction Buffer

1.75 µL

Ultra II End Prep Enzyme Mix

0.75 µL

**Total**

15 µL

19.2 Incubate at room temperature for 00:10:00

Incubate at 65 °C for 00:05:00

Incubate on ice for 00:01:00

19.3 Add the following directly to the previous reactions:

**Component****Volume**

NBXX barcode

2.5 µL

Ultra II Ligation Master Mix

17.5 µL

Ligation Enhancer

0.5 µL

**Total**

35.5 µL

**Note**

Use one native barcode from the EXP-NBD104 (1-12) or EXP-NBD114 (13-24) per sample. Use from 6 to 24 barcodes in a library, any fewer and there will be insufficient total material to achieve good yields.

19.4 Incubate at room temperature for 00:15:00

Incubate at 70 °C for 00:10:00

Incubate on ice for 00:01:00

**Note**

The 70°C incubation is to inactivate the DNA ligase to prevent barcode cross-ligation when reactions are pooled in the next step.



19.5 Pool all barcoded fragments together into a new 1.5 ml Eppendorf tube.

#### Protocol

NAME

#### Amplicon clean-up using SPRI beads

CREATED BY

Josh Quick

Preview

19.6 Quantify the barcoded amplicon pools using the Quantus Fluorometer using the ONE dsDNA assay.

#### Protocol

NAME

#### DNA quantification using the Quantus fluorometer

CREATED BY

Josh Quick

Preview

19.7 Set up the following AMII adapter ligation reaction:

#### Component

#### Volume

Barcoded amplicon pools

30  $\mu$ L

NEBNext Quick Ligation Reaction Buffer (5X)

10  $\mu$ L

AMII adapter mix

5  $\mu$ L

Quick T4 DNA Ligase


5  $\mu$ L


**Total**

50  $\mu$ L

**Note**

The input of barcoded amplicon pools will depend on the number of barcoded pools and should be between 40 ng (8 barcodes) and 160 ng (24 barcodes).


19.8 Incubate at room temperature for  00:15:00


19.9 Add  50  $\mu\text{L}$  (1:1) of SPRI beads to the sample tube and mix gently by either flicking or pipetting.

**Note**

Vortex SPRI beads thoroughly before use to ensure they are well resuspended, the solution should be a homogenous brown colour.

19.10 Pulse centrifuge to collect all liquid at the bottom of the tube.

19.11 Incubate for  00:05:00 at room temperature.

19.12 Place on magnetic rack and incubate for  00:02:00 or until the beads have pelleted and the supernatant is completely clear.

19.13 Carefully remove and discard the supernatant, being careful not to touch the bead pellet.

19.14 Add  200  $\mu\text{L}$  SFB and resuspend beads completely by pipette mixing.

**Note**

SFB will remove excess adapter without damaging the adapter-protein complexes. Do not use 70% ethanol as in early clean-ups.

19.15 Pulse centrifuge to collect all liquid at the bottom of the tube.




19.16 Remove supernatant and discard.

19.17 Repeat steps 14-16 to perform a second SFB wash.

19.18 Pulse centrifuge and remove any residual SFB.

#### Note

You do not need to allow to air dry with SFB washes.

19.19 Add  15  $\mu$ L EB and resuspend beads by pipette mixing.

19.20 Incubate at room temperature for  00:02:00 .

19.21 Place on magnetic rack.

19.22 Transfer final library to a new 1.5mL Eppendorf tube.

20 Quantify the final library using the Quantus Fluorometer using the ONE dsDNA assay.

#### Protocol

NAME

**DNA quantification using the Quantus fluorometer**

CREATED BY

Josh Quick


Preview




**Note**


Final library can now be stored in 10 mM Tris pH 8 at 4°C for up to a week if needed otherwise proceed directly to MinION sequencing.


- 20.1 Remove Lambda DNA 400 ng/μL standard from the freezer and leave on ice to thaw. Remove ONE dsDNA dye solution from the fridge and allow to come to room temperature.

 QuantiFluor(R) ONE dsDNA System, 500rxn **Promega Catalog #E4870**

- 20.2 Set up two  0.5 mL tubes for the calibration and label them 'Blank' and 'Standard'


- 20.3 Add  200 μL ONE dsDNA Dye solution to each tube.

- 20.4 Mix the Lambda DNA standard 400 ng/μL standard by pipetting then add  1 μL to one of the standard tube.

- 20.5 Mix each sample vigorously by vortexing for  00:00:05 and pulse centrifuge to collect the liquid.

- 20.6 Allow both tubes to incubate at room temperature for  00:02:00 before proceeding.


- 20.7 Selection 'Calibrate' then 'ONE DNA' then place the blank sample in the reader then select 'Read Blank'. Now place the standard in the reader and select 'Read Std'.

- 20.8 Set up the required number of  0.5 mL tubes for the number of DNA samples to be quantified.

**Note**

Use only thin-wall, clear, 0.5mL PCR tubes such as Axygen #PCR-05-C


20.9 Label the tubes on the lids, avoid marking the sides of the tube as this could interfere with the sample reading.

20.10 Add  199  $\mu\text{L}$  ONE dsDNA dye solution to each tube.

20.11 Add  1  $\mu\text{L}$  of each user sample to the appropriate tube.

#### Note

Use a P2 pipette for highest accuracy.


20.12 Mix each sample vigorously by vortexing for  00:00:05 and pulse centrifuge to collect the liquid.

20.13 Allow all tubes to incubate at room temperature for  00:02:00 before proceeding.

20.14 On the Home screen of the Quantus Fluorometer, select 'Protocol', then select 'ONE DNA' as the assay type.

#### Note

If you have already performed a calibration for the selected assay you can continue, there is no need to perform repeat calibrations when using ONE DNA pre diluted dye solution. If you want to use the previous calibration, skip to step 11. Otherwise, continue with step 9.

20.15 On the home screen navigate to 'Sample Volume' and set it to  1  $\mu\text{L}$  then 'Units' and set it to ng/ $\mu\text{L}$ .


20.16 Load the first sample into the reader and close the lid. The sample concentration is automatically read when you close the lid.

20.17 Repeat step 16 until all samples have been read.



- 20.18 The value displayed on the screen is the dsDNA concentration in ng/ $\mu$ L, carefully record all results in a spreadsheet or laboratory notebook.

## MinION sequencing

- 21 Prime the flowcell and load  20 ng sequencing library onto the flowcell.

### Protocol

NAME

### Priming and loading a MinION flowcell

CREATED BY

Josh Quick

Preview

### Note

From experience we know 20 ng is optimum loading input for short amplicons.

- 21.1 Thaw the following reagents at room temperature before placing on ice:

Sequencing buffer (SQB)

Loading beads (LB)

Flush buffer (FLB)


Flush tether (FLT)

- 21.2 Add  30  $\mu$ L FLT to the FLB tube and mix well by vortexing.

- 21.3 If required place a new MinION flowcell onto the MinION by flipping open the lip and pushing one end of the flowcell under the clip and pushing down gently.


- 21.4 Rotate the inlet port cover clockwise by 90° so that the priming port is visible.




- 21.5 Take a P1000 pipette and tip and set the volume to  800  $\mu\text{L}$  . Place the tip in the inlet port and holding perpendicularly to the plane of the flowcell remove any air from the inlet port by turning the volume dial anti-clockwise.


**Note**

Be careful not to remove so much volume that air is introduced onto the rectangular array via the outlet.





- 21.6 Load  800  $\mu\text{L}$  of FLB (plus FLT) into the flow cell via the inlet port, dispense slowly and smoothly trying to avoid the introduction of any air bubbles.

- 21.7 Wait for  00:05:00 .

- 21.8 Gently lift the SpotON cover to open the SpotON port.

- 21.9 Load another  200  $\mu\text{L}$  of FLB (plus FLT) into the flow cell via the inlet port, this will initiate a siphon at the SpotON port to allow you to load the library dilution.

- 21.10 In a new tube prepare the library dilution for sequencing:


Component	Volume
SQB	 37.5 $\mu\text{L}$
LB	 25.5 $\mu\text{L}$
Final library	 12 $\mu\text{L}$
<b>Total</b>	 75 $\mu\text{L}$

**Note**

Mix LB immediately before use as they settle quickly.

Dilute library in EB if required.



- 21.11 Mix the prepared library gently by pipetting up and down just prior to loading.
- 21.12 Add the  75 µL library dilution to the flow cell via the SpotON sample port in a dropwise fashion. Ensure each drop siphons into the port before adding the next.
- 21.13 Gently replace the SpotON sample port cover, making sure the bung enters the SpotON port, close the inlet port and close the MinION lid.
- 22 Start the sequencing run using MinKNOW.

#### Protocol

NAME

**Starting a MinION sequencing run using MinKNOW**

CREATED BY

Josh Quick

Preview

- 22.1 If required plug the MinION into the computer and wait for the MinION and flowcell to be detected.
- 22.2 Choose flow cell 'FLO-MIN106' from the drop-down menu.
- 22.3 Then select the flowcell so a tick appears.
- 22.4 Click the 'New Experiment' button in the bottom left of the screen.
- 22.5 On the New experiment popup screen, select the running parameters for your experiment from the individual tabs:



**Experiment:** Name the run in the experiment field, leave the sample field blank.

**Kit: Selection:** Select LSK109 as there is no option for native barcoding (NBD104).

**Run Options:** Set the run length to 6 hours (you can stop the run once sufficient data has been collected as determined using RAMPART).

**Basecalling:** Leave basecalling turned but select 'fast basecalling'.

**Output:** The number of files that MinKNOW will write to a single folder. By default this is set to 4000 but can be reduced to make RAMPART update more frequently.

Click 'Start run'.

22.6 Monitor the progress of the run using the MinKNOW interface.