

Takara Bio USA, Inc.

SMART-Seq® Mouse BCR (with UMIs) User Manual

Cat. Nos. 634351, 634352 & 634353
(010324)

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Table of Contents

I. Introduction.....	3
II. List of Components.....	5
III. Additional Materials Required (Not Provided).....	5
IV. General Considerations.....	7
A. General Recommendations.....	7
B. Sample Recommendations.....	7
V. Protocols.....	7
A. Protocol: First-Strand cDNA Synthesis.....	7
B. Protocol: BCR Amplification and Sequencing Library Generation.....	9
C. Protocol: Purification of Amplified Libraries.....	12
D. Protocol: Library Validation.....	14
VI. References.....	14
Appendix A: Guidelines for PCR Cycling.....	15
Appendix B: Sample Pooling and Indexing Recommendations.....	15
Appendix C: Guidelines for Library Sequencing.....	15
Appendix D: Guidelines for Data Analysis.....	17

Table of Figures

Figure 1. SMART-Seq Mouse BCR (with UMIs) protocol overview and timeline.....	3
Figure 2. SMART-Seq Mouse BCR (with UMIs) technology.....	4
Figure 3. Example electropherogram results for BCR heavy- and light- (kappa or lambda) chain libraries validated using the Agilent 2100 Bioanalyzer high sensitivity assay.....	14
Figure 4. SMART-Seq Mouse BCR (with UMIs) library structure.....	17

Table of Tables

Table 1. SMART-Seq Mouse BCR (with UMIs) components.....	5
Table 2. Cycling guidelines based on amount of starting material.....	9
Table 3. Illumina instrument and reagent kit recommendations.....	16
Table 4. Recommended sequencing depth for BCR libraries prepared from Mouse Spleen RNA.....	17

I. Introduction

SMART-Seq Mouse BCR (with UMIs) (Cat. Nos. 634351, 634352 & 634353) enables users to analyze B-cell receptor (BCR) repertoires from bulk total RNA samples. This kit can generate libraries from 10 ng–1 µg of total RNA from spleen, bone marrow, or peripheral blood mononuclear cells (PBMCs). This kit is also appropriate for other sample types, such as RNA extracted from mouse whole blood (RNA input ≥ 100 ng) or lymph nodes. We recommend good quality RNA [RNA integrity number (RIN) >7] be used in this workflow.

This protocol can be used to generate data for both heavy and light chains (kappa and lambda) of mouse immunoglobulin IgG, IgM, IgA, IgD, and IgE (Figure 1). Using the Unique Dual Index Kit (Cat. Nos. 634752–634756, sold separately), the protocol results in indexed libraries that are ready for sequencing on Illumina® platforms. With sequencing output from Illumina sequencers as input to our free-to-use [Cogent™ NGS Immune Profiler](#) (CogentIP) and web-based [Cogent NGS Immune Viewer](#) bioinformatics software (Appendix D), we provide an end-to-end solution from samples to publication-ready data.

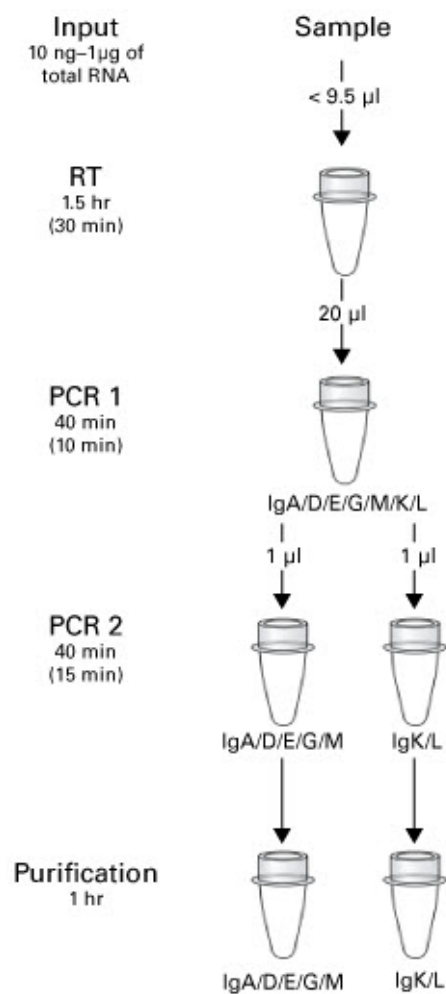


Figure 1. SMART-Seq Mouse BCR (with UMIs) protocol overview and timeline. For each sample, after reverse transcription of mRNAs, the entire volume of the RT reaction (20 µl) is used as input in the first round of PCR (PCR 1), where all seven immunoglobulin chains are amplified. 1 µl of the PCR 1 product is used as input for a second round of PCR (PCR 2) which adds the same sequencing indexes to each amplified chain for a given sample, but distinct indexes for different samples. The heavy (IgA/D/E/G/M) and light (IgK/L) chains are amplified separately. After PCR 2, the amplified libraries are purified, and we recommend analyzing and validating the results. Numbers within the parentheses are estimated hands-on times at each step.

The protocol leverages SMART® technology (Switching Mechanism at 5' End of RNA Template) and employs a 5' RACE-like approach to capture complete V(D)J variable regions of BCR transcripts (Figure 2). It also incorporates unique molecular identifiers (UMIs) to facilitate PCR error correction and clonotype quantification during data analysis.

First-strand cDNA synthesis is oligo-dT primed and catalyzed by SMARTScribe™ Reverse Transcriptase (RT), which adds nontemplated nucleotides to the 5' end of each cDNA molecule. The SMART UMI Oligo anneals to these non-templated nucleotides, serves as a template for incorporation of a PCR handle into the first-strand cDNA, and uniquely tags each cDNA molecule with a UMI. Following reverse transcription, two rounds of PCR are performed to amplify BCR cDNAs. To capture the entire V(D)J region, during PCR 1, primers anneal to sequences added by the SMART UMI Oligo at one end and the BCR constant region(s) at the other end. In PCR 2, the product from PCR 1 is used as a template and semi-nested primers are used to amplify the entire BCR variable region and a small portion of the constant region.

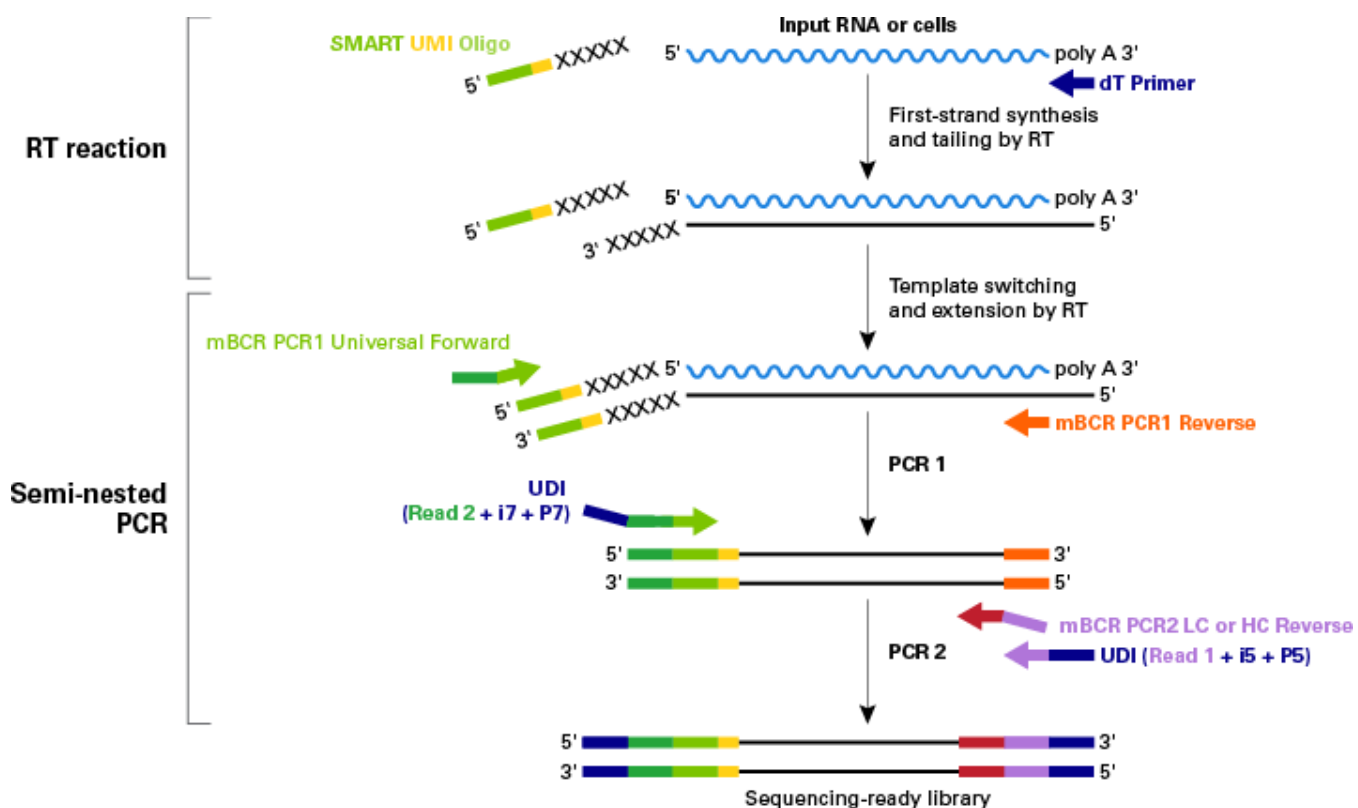


Figure 2. SMART-Seq Mouse BCR (with UMIs) technology. dT-primed, first-strand cDNA synthesis is followed by two rounds of successive PCR for amplification of cDNA sequences. After post-PCR purification, size selection, and quality analysis, the library is ready for sequencing.

II. List of Components

- The components of SMART-Seq Mouse BCR (with UMIs) have been designed to work together and are optimized for this protocol. The substitution of reagents in the kit and/or a modification of the protocol may lead to unexpected results.
- The reaction number for each kit specifies the number of samples that can be put into RT reactions. There is enough material included in the kit to generate heavy-chain (IgA, IgD, IgE, IgG, IgM), and light-chain (IgK, and IgL) sequencing libraries from each RT reaction.

NOTE: UDI Kits are not included and sold separately (Cat. Nos. 634752–634756). The user may select the appropriate size UDI kit depending on the number of libraries to be pooled; the UDI kits allow for preparing and pooling up to 384 Illumina-compatible libraries.

Table 1. SMART-Seq Mouse BCR (with UMIs) components.

SMART-Seq Mouse BCR (with UMIs)	Cap color	634352 (24 rxns)	634353 (96 rxns)	634351 (384 rxns)
Box 1 (Store at –70°C)				
Control RNA* (1 µg/µl)	–	5 µl	5 µl	4 x 5 µl
SMART UMI Oligo	Pink	24 µl	96 µl	4 x 96 µl
Box 2 (Store at –20°C)				
dT Primer	Light blue	48 µl	192 µl	4 x 192 µl
First-Strand Buffer (5X)	Purple	96 µl	384 µl	4 x 384 µl
SMARTScribe Reverse Transcriptase (100 U/µl)	Purple	48 µl	192 µl	4 x 192 µl
Nuclease-Free Water†	–	5 ml	20 ml	4 x 20 ml
BCR Enhancer†	White	24 µl	96 µl	4 x 96 µl
RNase Inhibitor (40 U/µl)	White	24 µl	96 µl	4 x 96 µl
Elution Buffer‡ (10 mM Tris HCl, pH 8.5)	–	1 ml	4 x 1 ml	16 x 1 ml
mBCR PCR1 Universal Forward	Blue	24 µl	96 µl	4 x 96 µl
mBCR PCR1 Reverse	Red	24 µl	96 µl	4 x 96 µl
mBCR PCR2 HC Reverse	Yellow	24 µl	96 µl	4 x 96 µl
mBCR PCR2 LC Reverse	Orange	24 µl	96 µl	4 x 96 µl
PrimeSTAR® GXL Premix (2X)	White	2 x 1 ml	8 x 1 ml	32 x 1 ml

*Control RNA is Mouse Spleen Total RNA.

†Store Nuclease-Free Water and BCR Enhancer at –20°C. Once thawed, they can be stored at 4°C.

‡Store Elution Buffer at –20°C. Once thawed, the buffer can be stored at room temperature.

III. Additional Materials Required (Not Provided)

The following reagents and materials are required but not supplied. The named products have been validated to work with this protocol. Please do not make any substitutions as you may not obtain the expected results.

- Unique Dual Index Kit
 - 96 indexes: Takara Bio, Cat. Nos. 634752, 634753, 634754, and/or 634755
 - 24 indexes: Takara Bio, Cat. No. 634756
- Single-channel pipettes: 10 µl, 20 µl, and 200 µl
- Eight-channel pipettes (recommended): 20 µl and 200 µl
- Filter pipette tips: 2 µl, 20 µl, and 200 µl
- Minicentrifuge for 1.5 ml tubes
- Minicentrifuge for 0.2 ml tubes or strips

For PCR Amplification & Validation:

- Thermal cyclers
 - One dedicated for first-strand cDNA synthesis (Protocol V.A)
 - One dedicated for library amplification by PCR (Protocol V.B)

NOTE: The thermal cycler should always be used with the heated-lid option turned on. If prompted to input a specific temperature, use 105°C.

Most thermal cyclers with heated lids will automatically adjust the lid temperature just above the highest block temperature within a cycling program. However, if your thermal cycler does not make this automatic adjustment, you may want to follow the manufacturer's instructions to choose a lower lid temperature for the reverse transcription step.

- For validation (Section V.D)
 - Agilent 2100 Bioanalyzer: Agilent High Sensitivity DNA Kit (Agilent Technologies, Cat. No. 5067-4626) or DNA 1000 Kit (Agilent Technologies, Cat. No. 5067-1504)
 - Qubit dsDNA HS Kit (Thermo Fisher Scientific, Cat. No. Q32851 or Q32854)
- Nuclease-free thin-wall PCR tubes, 96 well plates, or strips (0.2 ml PCR 8-tube strip; USA Scientific, Item No.1402-4700 or similar)
- Nuclease-free low-adhesion 1.5 ml tubes (USA Scientific, Item No. 1415-2600), LoBind tubes (Eppendorf, Cat. No. 022431021), or similar

For Bead Purifications (Protocol V.C):

- NucleoMag NGS Clean-up and Size Select (Takara Bio; 5 ml size: Cat. No. 744970.5; 50ml size: Cat. No. 744970.50; 500 ml size: Cat. No. 744970.500) If the NucleoMag product is not available, the AMPure XP PCR purification kit (Beckman Coulter; 5 ml size: Cat. No. A63880; 60 ml size: Cat. No. A63881) is an appropriate substitute and can be used in the same bead:sample ratio as NucleoMag NGS Clean-Up and Size Select.

NOTES:

- The kit has been specifically validated with the beads listed above. Substitutions may lead to unexpected results.
 - Beads need to come to room temperature before the container is opened. We strongly recommend aliquoting the beads into 1.5 ml tubes upon receipt and then refrigerating the aliquots. Individual tubes can be removed for each experiment, allowing them to come to room temperature more quickly (~30 minutes). Aliquoting is also instrumental in decreasing the chances of bead contamination.
- 100% ethanol (molecular biology grade)
 - Magnetic separation device for small volumes—used to purify amplified libraries.
 - For 8-tube strips: SMARTer-Seq™ Magnetic Separator - PCR Strip (Takara Bio, Cat. No. 635011)
 - For 96-well plates: Magnetic Stand-96 (Thermo Fisher, Cat. No. AM10027)
 - Optional, depending on the choice of magnetic separation device (96-well format):
 - 96-well V-bottom Plate (500 µl; VWR, Cat. No. 47743-996)
 - MicroAmp Clean Adhesive Seal (Thermo Fisher, Cat. No. 4306311)
 - Low-speed benchtop centrifuge for a 96-well plate

IV. General Considerations

A. General Recommendations

IMPORTANT! For Cat. No. 634353 (96 rxns) and Cat. No. 634351 (4 x 96 rxns), we recommend performing a **minimum** of 12 reactions per protocol run to ensure sufficient reagents to utilize 96 (or 384) reactions per kit.

- We recommend using two physically separated workstations to minimize contamination:
 - **A PCR Clean Workstation** for all pre-PCR experiments that require clean room conditions, e.g., first-strand cDNA synthesis (Protocol V.A). The PCR Clean Workstation should be in a clean room with positive air flow.
 - **A second workstation located in the general laboratory** where you will perform PCR (Protocol V.B) and measure library concentration (Protocol V.D).
- All lab supplies related to cDNA synthesis should be stored in a closed, DNA-free cabinet. Reagents for cDNA synthesis should be stored in a freezer/refrigerator that has not previously been used to store PCR amplicons.
- Add enzymes to reaction mixtures last and thoroughly incorporate them by gently pipetting the reaction mixture up and down.
- Do not increase (or decrease) the amount or concentration of any reaction component. The amounts and concentrations have been carefully optimized for the included reagents and protocol.
- If you are using this protocol for the first time, we strongly recommend that you perform negative and positive control reactions to verify that kit components are working properly.

B. Sample Recommendations

- This protocol has been optimized for 10 ng of total RNA extracted from spleen.
- RNA should be of high integrity (RIN>7) to enable oligo(dT)-priming.
- The success of your experiment depends on the quality of your input RNA. Prior to cDNA synthesis, ensure your RNA is in nuclease-free water, intact, and free of contaminants. Input RNA should be free from poly(A) carrier RNA that will interfere with oligo(dT)-primed cDNA synthesis.
- When choosing a purification kit, ensure that it is appropriate for your sample type, input mass, and includes DNase treatment. We recommend NucleoSpin RNA XS kits (Takara Bio, Cat. No. 740902.10, 740902.50, or 740902.250).
- After RNA extraction, if your sample size is not limiting, we recommend evaluating total RNA quality using the Agilent RNA 6000 Pico Kit (Agilent Technologies, Cat. No. 5067-1513) or an equivalent platform. Refer to the manufacturer for instructions.

V. Protocols

A. Protocol: First-Strand cDNA Synthesis

First-strand cDNA synthesis (from RNA) is primed by dT Primer and uses SMART UMI Oligo for template switching at the 5' end of the transcript.

For this protocol, you will need the following components:

First-Strand Buffer (5X), BCR Enhancer, SMART UMI Oligo, dT Primer, SMARTScribe Reverse Transcriptase, and RNase Inhibitor.

1. Thaw the First-Strand Buffer (5X) at room temperature. Do not store on ice.
2. Thaw BCR Enhancer, SMART UMI Oligo, and dT Primer on ice. Gently vortex each reagent to mix and centrifuge briefly. Store on ice until needed.
3. Preheat the thermal cycler to 72°C.
4. On ice, prepare samples and controls in nuclease-free, thin-wall PCR tubes, plates, or strips by adding the reagents in the order shown below.

Components	Sample	Negative control	Positive control
Sample	1–9.5 µl	–	–
Diluted Control RNA*	–	–	1–9.5 µl
Nuclease-Free Water	Up to 8.5 µl	9.5 µl	Up to 8.5 µl
BCR Enhancer	1 µl	1 µl	1 µl
dT Primer	2 µl	2 µl	2 µl
Total Volume	12.5 µl	12.5 µl	12.5 µl

*The Control RNA is supplied at a concentration of 1 µg/µl. It should be thawed on ice and diluted serially in Nuclease-Free Water to obtain the appropriate concentration (10 ng–1 µg, your choice). We have tested this protocol extensively using the PCR cycling conditions below ([Table 2](#)) (16 cycles for PCR 1 and 20 cycles for PCR 2) with 10 ng of input Control RNA

Mix by gently vortexing and then centrifuge briefly.

5. Incubate the tubes or plates at 72°C in the preheated, heated-lid thermal cycler for 3 min.

NOTE: Prepare the RT Premix (Step 7) while your tubes are incubating. The SMARTScribe Reverse Transcriptase will be added just before use (Step 10). Steps 11 & 12 below are critical for first-strand cDNA synthesis and should not be delayed after completing Step 10.

6. Remove the RNase Inhibitor from the freezer, centrifuge briefly to mix, and store on ice.
7. In a 1.5 ml tube, prepare enough RT Premix for all the reactions (scale up as needed), plus 10% of the total reaction mix volume, by combining the following reagents in the order shown at room temperature:

RT Premix:

4 µl	First-Strand Buffer (5X)*
1 µl	SMART UMI Oligo
0.5 µl	RNase Inhibitor (40 U/µl)

5.5 µl Total volume per reaction

*The First-Strand Buffer may form precipitates; vortex before using to ensure all components are completely in solution.

8. Immediately after the 3-min incubation at 72°C (Step 5), place the samples on ice or at 4°C for 2 min (but no more than 10 min).
9. Preheat the thermal cycler to 42°C.

- Remove the SMARTScribe Reverse Transcriptase from the freezer, centrifuge briefly. Calculate the total volume to add for all reactions based on 2 µl per reaction, plus 10% of the total volume, then add to the RT Premix (from Step 7) to make the RT Master Mix.

RT Master Mix:

5.5 µl	RT Premix
2 µl	SMARTScribe Reverse Transcriptase
7.5 µl	Total volume per reaction

Mix well by gently vortexing and then spin the tube briefly in a minicentrifuge to collect the contents at the bottom of the tube.

- Add 7.5 µl of the RT Master Mix to each reaction tube or well. Mix the contents of each tube or well by pipetting gently and centrifuge briefly.
- Place the tubes/plate in the preheated thermal cycler and run the following program:

42°C	90 min
70°C	10 min
4°C	forever

SAFE STOPPING POINT: The tubes can be stored at 4°C overnight.

B. Protocol: BCR Amplification and Sequencing Library Generation

Semi-nested PCR amplifies the entire V(D)J region and a portion of the constant regions of BCR cDNA and incorporates adapters and barcodes for Illumina sequencing platforms. Table 2 provides PCR-cycling recommendations, but optimal parameters may vary for different sample types, input amounts, and thermal cyclers. We recommend trying a range of cycle numbers to determine the minimum number necessary to obtain the desired yield. For more information, see Appendix A.

Table 2. Cycling guidelines based on amount of starting material.

RNA source	Input amount	Number of PCR 1 cycles	Number of PCR 2 cycles*
PBMC	10 ng	16	20
	100 ng	16	18
	1 µg	16	16
Spleen	10 ng	16	20
	100 ng	16	18
	1 µg	16	16
Control RNA	10 ng	16	20
	100 ng	16	18
	1 µg	16	16

*If the number of cycles generates an insufficient library for sequencing, repeat PCR 2 with more cycles. See Appendix A for more details.

1. PCR 1

The first PCR selectively amplifies full-length BCR V(D)J regions from first-strand cDNA generated with the previous protocol (Section V.A). The mBCR PCR1 Universal Forward primer anneals to the SMART UMI oligo sequence (incorporated during first-strand cDNA synthesis) and adds the Illumina Read 2 sequence. The mBCR PCR1 Reverse primer—a mixture of reverse

primers for IgA/D/E/G/M heavy chains and IgK/L light chains—anneals to sequences in the constant regions of mBCR heavy-chain and light-chain cDNA.

For this protocol, you will need the following components:

PrimeSTAR GXL Premix (2X), mBCR PCR1 Universal Forward, mBCR PCR1 Reverse, and Nuclease-Free Water.

1. Thaw all the reagents needed for PCR on ice. Gently vortex each reagent tube (except PrimeSTAR GXL Premix) briefly to mix and spin down. Store on ice.
2. Prepare PCR1 Master Mix by combining the following components in the order shown in the table below for all reactions (scale up as needed), plus 10% of the total reaction mix volume.

PCR1 Master Mix:

3 µl	Nuclease-Free Water
1 µl	mBCR PCR1 Universal Forward
1 µl	mBCR PCR1 Reverse
25 µl	PrimeSTAR GXL Premix (2X)
<hr/>	
30 µl	Total volume per reaction

Gently vortex to mix then briefly centrifuge.

3. Add 30 µl of the PCR1 Master Mix to each tube or well containing 20 µl of the first-strand cDNA produced in Section V.A. Mix well and briefly spin to collect the contents at the bottom of the tubes/wells.

PCR1 reaction mixture:

20 µl	First-strand cDNA product (Section V.A)
30 µl	PCR1 Master Mix
<hr/>	
50 µl	Total volume per reaction

4. Place the tubes/plate in a preheated thermal cycler with a heated lid and run the following program:

PCR 1

95°C	1 min	
16 cycles:		
98°C	10 sec	}
60°C	15 sec	
68°C	45 sec	
4°C	forever	

SAFE STOPPING POINT: The tubes or plate may be stored at 4°C overnight.

2. PCR 2

The second PCR reaction further amplifies sequences corresponding to the full-length BCR variable regions and adds Illumina sequencing adapters using a semi-nested approach. The Unique Dual Index primers are added during this step and anneal to a sequence added by mBCR PCR1 Universal Forward (from the previous step) and add Illumina P7-i7 index sequences. The nested mBCR PCR2 HC Reverse and mBCR PCR2 LC Reverse primers anneal to sequences in

BCR constant regions that are internal to the sequences bound by the mBCR PCR1 Reverse primer and add both the Illumina Read 1 and P5-i5 index sequences.

Libraries can be pooled at this point of the protocol. For sample pooling and indexing recommendations, refer to Appendix B.

For this protocol, you will need the following components:

PrimeSTAR GXL Premix (2X), mBCR PCR2 HC Reverse, mBCR PCR2 LC Reverse, selected Unique Dual Index Kit, and Nuclease-Free Water.

NOTE: The heavy chains (IgG/M/A/D/E) and the light chains (IgK/L) are amplified separately in this PCR step. You will need to prepare two reactions if you want to amplify both heavy and light chains from a sample.

1. Thaw all the reagents needed for PCR (except PrimeSTAR GXL Premix) on ice. Gently vortex each reagent tube to mix and spin down briefly. Store on ice.
2. On ice, prepare a PCR2 Master Mix by combining the following components in the order shown in the table (scale up as needed), plus 10% of the total reaction mix volume. Remove the PrimeSTAR GXL Premix (2x) from the freezer, gently mix the tube without vortexing, and add to the master mix just before use.

PCR2 Master Mix:

21 µl	Nuclease-Free Water
1 µl	mBCR PCR2 HC Reverse* -or- mBCR PCR2 LC Reverse*
25 µl	PrimeSTAR GXL Premix (2X)

47 µl Total volume per reaction

*Each PCR primer is used in a separate PCR Master Mix. Use mBCR PCR2 HC Reverse when amplifying heavy chains; use mBCR PCR2 LC Reverse when preparing to amplify light chains.

Gently vortex to mix, then briefly centrifuge.

NOTE: Alternatively, you can prepare the PCR2 Master Mix with only the Nuclease-Free Water and PrimeStar GXL Premix (2X), then plan to add 1 µl of each primer individually instead of including in the PCR2 Master Mix. This is particularly recommended if the number of samples is low.

3. For each reaction, add 47 µl of PCR2 Master Mix to a clean, nuclease-free 96-well plate, 0.2 ml tube, or 8-tube strip.
4. Add 1 µl of appropriate PCR1 product to each well/tube containing PCR2 Master Mix.
5. Add 2 µl of the appropriate UDI from the Unique Dual Index Kit (12.5 µM) to each well/tube.

PCR2 reaction mixture:

47 µl	PCR2 Master Mix (Step 3)
1 µl	PCR1 product (Step 4)
2 µl	UDI

50 µl Total volume per reaction

Mix well and briefly spin to collect the contents at the bottom of the wells/tubes.

6. Place the tubes/plate in a preheated thermal cycler and run the following program:

PCR 2

95°C	1 min	
<i>N</i> cycles*:		
98°C	10 sec]
60°C	15 sec	
68°C	45 sec	
4°C	forever	

*Consult [Table 2](#) (above) for PCR cycle number (*N*) guidelines.

SAFE STOPPING POINT: The tubes may be stored at 4°C overnight.

C. Protocol: Purification of Amplified Libraries

PCR-amplified libraries are purified by immobilization on NucleoMag NGS Clean-up and Size Select beads. Purify the libraries again by immobilization on NucleoMag NGS Clean-up and Size Select beads. The beads are then washed with 80% ethanol and the libraries eluted with Elution Buffer.

NOTES:

- Before each use, bring a 1.5 ml aliquot of NucleoMag NGS Clean-up and Size Select beads to room temperature for at least 30 min and mix well by vortexing. Use room-temperature Elution Buffer for this protocol.
- Bead:sample ratio is 0.7:1
- Prepare fresh 80% ethanol for each experiment. You will need 400 µl per sample.
- Use a magnetic separation device for 0.2 ml tubes, strip tubes, or a 96-well plate.

1. Add 25 µl of the NucleoMag beads to each sample. Mix thoroughly by gently pipetting the entire volume up and down at least 10 times.

NOTE: The beads are viscous; pipette the entire volume and push it out slowly. **Do not vortex. Vortexing will generate bubbles, making subsequent handling of the beads difficult.**

2. Incubate at room temperature for 8 min to let the DNA bind to the beads.
3. Briefly spin the samples to collect the liquid from the side of the tube or sample well. Place the samples on the magnetic separation device for ~5 min or longer until the liquid appears completely clear, and there are no beads left in the supernatant; any bead carryover will decrease the efficiency of size selection.

NOTE: There is no disadvantage to separating the samples for longer than 5 min, if needed.

4. Keep the reaction plate or tubes on the magnetic separation device. Use a pipette to transfer the supernatant (which contains your library) to a clean PCR plate or tubes. After transferring, remove the plate or tubes containing the beads from the magnetic separation device and discard them.

5. Add 10 µl of NucleoMag beads to each tube containing supernatant.

NOTE: Before use, ensure that the beads are fully resuspended. If the beads appear to have settled at the bottom of the tube, vortex briefly to ensure that they are completely mixed before adding them to the tubes.

6. Mix thoroughly by gently pipetting the entire volume up and down at least 10 times.

7. Incubate at room temperature for 8 min to let the DNA bind to the beads.

8. Place the tubes on the magnetic separation device for ~10 min or until the solution is completely clear. The libraries are now bound to the beads.
9. Keep the plate or tubes on the magnetic separation device. Remove the supernatant with a pipette and discard it.
10. Add 200 µl of freshly made 80% ethanol to each sample, without disturbing the beads, to wash away contaminants. Wait for 30 sec and use a pipette to carefully remove the supernatant containing contaminants. The library will remain bound to the beads during the washing process.
11. Repeat the ethanol wash (Step 10) once more.
12. Briefly spin the plate or tubes (~2,000g) to collect the remaining liquid at the bottom of each well or tube.
13. Place the plate or tubes on the magnetic separation device for 30 sec, then remove all remaining liquid with a pipette.
14. Let the sample plate or tubes rest open on the magnetic separation device at room temperature for ~2–2.5 min until the pellet appears dry and is no longer shiny. You may see a tiny crack in the pellet.

NOTE: Check the pellet frequently during this time and continue to Step 15 when it is dry enough. Please refer to our visual guide for pellet dry-down on our website if you'd like additional assistance with this determination.

<https://www.takarabio.com/learning-centers/next-generation-sequencing/faqs-and-tips/rna-seq-tips>

If the beads are overdried, there will be cracks in the pellet. If this occurs, the DNA may not elute well from the beads and recovery may be reduced.

15. Once the bead pellet has dried, remove the plate or tubes from the magnetic separation device and add 17 µl of Elution Buffer to cover each pellet. Mix thoroughly by pipetting up and down to ensure complete bead dispersion.

NOTE: Be sure that the beads are completely resuspended. The beads can sometimes stick to the sides of the well or tube.

16. Incubate at room temperature for at least 5 min to rehydrate.
17. Briefly spin the samples to collect the liquid from the side of the sample well or tube. Place the samples back on the magnetic separation device for 2 min or longer until the solution is completely clear.

NOTE: There may be a small population of beads that do not pellet against the magnet during incubation. Pipette these nonpelleted beads (without disrupting the existing pellet) up and down to resuspend them with the supernatant, and then pipette them towards the magnet where the rest of the beads have already pelleted. Continue the incubation until there are no beads left in the supernatant.

18. Transfer clear supernatant containing purified BCR library from each well or tube to a nuclease-free, low-adhesion tube. Label each tube with sample information and store at –20°C.

SAFE STOPPING POINT: The tubes may be stored at –20°C indefinitely.

D. Protocol: Library Validation

To assess the success of library preparation, purification, and size selection, we recommend

- analyzing and validating final libraries using a Qubit 2.0 Fluorometer and Qubit dsDNA HS Assay Kit (Thermo Fisher Scientific, Cat. No. Q32851 or Q32854) and
- evaluating the libraries' size distributions with an Agilent 2100 Bioanalyzer and the DNA High Sensitivity Kit (Agilent Technologies, Cat. No. 5067-4626) or Agilent DNA 1000 Kit (Agilent Technologies, Cat. No. 5067-1504).

Please refer to the corresponding user manuals for detailed instructions.

1. **Qubit:** Use 1 μ l of undiluted library for quantification using a Qubit 2.0 Fluorometer (see manufacturer's instructions for more details).
2. **Bioanalyzer:** To validate libraries using the DNA High Sensitivity Kit, dilute libraries to a concentration of 0.2 ng/ μ l. If using the Agilent DNA 1000 Kit, dilute libraries to 1 ng/ μ l.

Compare the results for your samples with Figure 3 to verify whether each sample is suitable for further processing. High quality libraries should yield no product for negative control reactions and a broad peak spanning 500–1,200 bp, with a maximum between ~600–900 bp, for positive controls and samples containing BCR RNA. The position and shape of electropherogram peaks will vary depending on which chain sequences are included in the library, the nature of the RNA sample, and the analysis method.

Following validation, libraries are ready for sequencing on Illumina platforms. See Appendix C for sequencing guidelines.

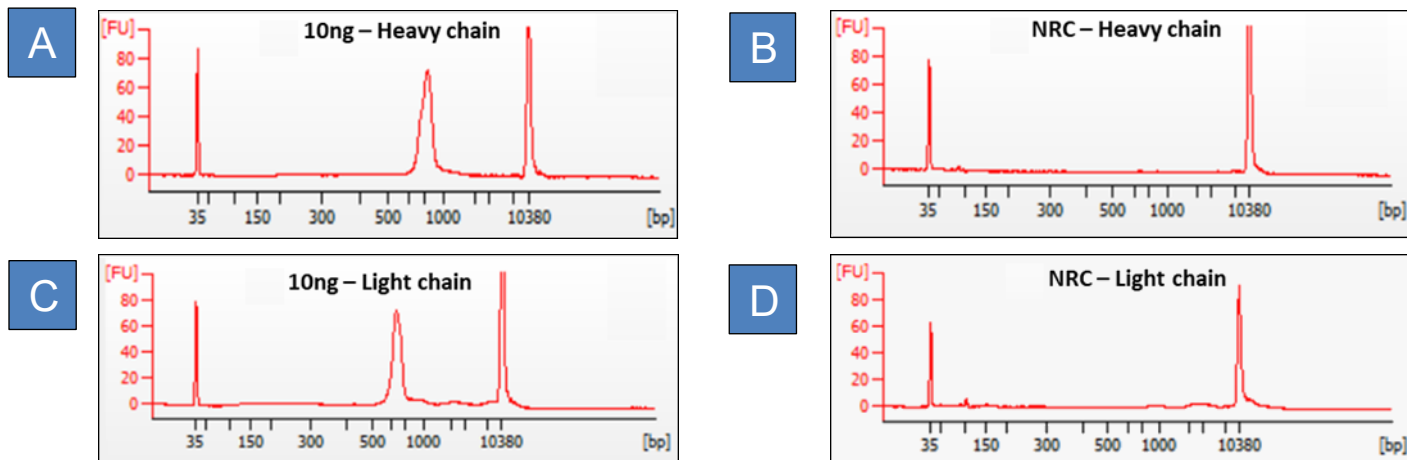


Figure 3. Example electropherogram results for BCR heavy- and light- (kappa or lambda) chain libraries validated using the Agilent 2100 Bioanalyzer high sensitivity assay. Libraries containing BCR heavy- and light-chain sequences were generated using SMART-Seq Mouse BCR (with UMIs) and 10 ng of RNA obtained from mouse spleen RNA. Libraries were produced using 20 μ l of first-strand cDNA as a template in a single PCR reaction for 16 cycles. 1 μ l of the product from PCR 1 was used as a template in a second, nested PCR reaction for 20 cycles. Following purification and size selection, libraries were analyzed on an Agilent 2100 Bioanalyzer. (**Panels A, C**) Broad peaks seen between ~500–1,200 bp and maximal peaks in the range of ~600–900 bp (typical results for a library generated from PBMC RNA), analyzed on the Bioanalyzer. (**Panels B, D**) No RNA Control (NRC) samples show no library produced and a flat Bioanalyzer profile within the range of 500–1,200 bp. NRC libraries often show dimer peaks in the 50–100 bp region.

VI. References

Chenchik, A., Zhu, Y., Diatchenko, L., Li, R., Hill, J. & Siebert, P. Generation and use of high-quality cDNA from small amounts of total RNA by SMART PCR. In *RT-PCR Methods for Gene Cloning and Analysis*. Eds. Siebert, P. & Larrick, J. (BioTechniques Books, MA), pp. 305–319 (1998).

Appendix A: Guidelines for PCR Cycling

The gene expression of each BCR chain can vary significantly from one B-cell population to another; it is therefore challenging to predict how many cycles of PCR will amplify sufficient material for downstream sequencing without overamplification such that there are significant off-target PCR products. The guidelines provided in [Table 2](#) are a good starting point for determining the number of cycles to perform in PCR 2 and have been determined by testing several varieties of sample types and input amounts.

It is possible that the suggested number of cycles will not generate enough material for downstream sequencing (see Appendix C). In these cases, we recommend increasing the number of cycles. The optimal range of library yield is between 3 ng/μl and 30 ng/μl (as determined by quantification with a Qubit dsDNA HS Assay Kit). If the library yield is lower than 3 ng/μl or over 30 ng/μl, we recommend increasing or reducing, respectively, the number of PCR 2 cycles by 1 to reach the optimal yield range.

Appendix B: Sample Pooling and Indexing Recommendations

Unique combinations of Illumina indexes are required to ensure enough diversity and allow for discrimination between samples when sequencing a pool of two or more libraries on a single flow cell. If you anticipate that the number of libraries will exceed the maximum index combinations provided in the UDI kits (384), the same library indexes could be used for each PCR 2 amplification of different chains originating from the same sample so that they are sequenced together as a single library. For example, the same index can be used for both heavy chain library and light chain library if they are from the same sample. Consult the Illumina literature (e.g., TruSeq® DNA Sample Preparation Guide) for appropriate pooling guidelines.

SMART-Seq Mouse BCR (with UMIs) requires use of the Unique Dual Index Kits (Cat. Nos, 634752–634756, sold separately). The indexes are 8-nt long and employ "IDT for Illumina TruSeq UD Indexes" i5 and i7 dual index sequences. In all versions of the UDI kits, the primers are provided in a 96-well plate format; the indexes in Unique Dual Index Kit (1–24) (Cat. No. 634756) are a subset of Unique Dual Index Kit (1–96) (Cat. No. 634752). Please consult the following resources for component information, best practices, pooling strategies, an index plate map, and index sequences.

- Unique Dual Index Kits Protocol-At-A-Glance ([download](#))
- Indexes and plate maps (Excel files)
 - Unique Dual Index Kit (1-96) Indexes and Plate Map ([download](#))
 - Unique Dual Index Kit (97-192) Indexes and Plate Map ([download](#))
 - Unique Dual Index Kit (193-288) Indexes and Plate Map ([download](#))
 - Unique Dual Index Kit (289-384) Indexes and Plate Map ([download](#))
 - Unique Dual Index Kit (1-24) Indexes and Plate Map ([download](#))

Appendix C: Guidelines for Library Sequencing

Following library validation by Qubit and Bioanalyzer, prepare the desired library pools for the sequencing run. Prior to pooling, libraries must be carefully quantified. We recommend quantification by qPCR using the Library Quantification Kit (Takara Bio, Cat. No. 638324). Alternatively, by combining the quantification obtained with the Qubit with the average library size determined by the Bioanalyzer, the concentration in ng/μl can be converted to nM. The following web tool is convenient for the conversion:

http://www.molbiol.ru/eng/scripts/01_07.html

Most Illumina sequencing library preparation protocols require libraries with a final concentration of 4 nM, including the MiSeq instrument that we recommend for this kit.

Prepare a pool of 4 nM as follows:

1. Dilute each library to 4 nM in Nuclease-Free Water. To avoid pipetting error, use at least 2 µl of each original library for dilution.
2. Pool the diluted libraries by combining an equal amount of each library in a low-bind 1.5 ml tube. Mix by vortexing at low speed or by pipetting up and down. To avoid pipetting error, use at least 2 µl of each diluted library.
3. Use a 5 µl aliquot of the 4 nM concentration pooled libraries. Follow the library denaturation protocol according to the latest edition of your Illumina sequencing instrument's User Guide.

You should also plan to include a 20% PhiX control spike-in (PhiX Control v3, Illumina, Cat. No. FC-110-3001). The addition of the PhiX control is essential to increase the nucleotide diversity and achieve high-quality data generation.

NOTE: Follow Illumina guidelines on how to denature, dilute, and combine a PhiX control library with your own pool of libraries. Make sure to use a fresh and reliable stock of the PhiX control library.

To obtain the full length of V(D)J information from the BCR clones, sequencing should be performed on an Illumina MiSeq® sequencer using the 600-cycle MiSeq Reagent Kit v3 (Illumina, Cat. No. MS-102-3003) with paired-end, 2 x 300 base pair reads. If using qPCR for quantification, we recommend diluting the pooled denatured libraries to a final concentration of 7.5 pM and using 20% PhiX control spike-in to achieve optimal cluster density. When relying on Qubit quantification, you may need to use a higher final concentration.

Alternatively, if only CDR3 region information is needed, use paired-end, 2 x 150 base reads and the following Illumina instrument and kit combinations (Table 3).

Table 3. Illumina instrument and reagent kit recommendations.

Sequence	Kit	Cat. No.
MiniSeq™	MiniSeq High Output reagent kit 300-cycle	Cat. No. FC-420-1003
	Mid Output reagent kit 300-cycle	Cat. No. FC-420-1004
MiSeq	MiSeq reagent kit v2 300-cycle	Cat. No. MS-102-2002
	MiSeq reagent Micro kit v2 300-cycle	Cat. No. MS-103-1002
	MiSeq reagent Nano kit v2 300-cycle	Cat. No. MS-103-1001
	MiSeq reagent kit v3 600-cycle	Cat. No. MS-102-3003
NextSeq® 550	NextSeq High Output kit v2.5 300-cycle	Cat. No. 20024908
	NextSeq Mid Output kit v2.5 300-cycle	Cat. No. 20024905
NextSeq 1000/2000	NextSeq 1000/2000 P1 Reagents 300 Cycles	Cat. No. 20050264
	NextSeq 1000/2000 P1 Reagents 600 Cycles	Cat. No. 20075294
	NextSeq 1000/2000 P2 Reagents (300 Cycles) v3	Cat. No. 20046813
	NextSeq 1000/2000 P2 Reagents 600 Cycles	Cat. No. 20075295
NovaSeq™	NovaSeq SP reagent kit 300-cycle	Cat. No. 20027465
	NovaSeq SP reagent kit 500-cycle	Cat. No. 20029137
	NovaSeq S4 reagent kit 300-cycle	Cat. No. 20012866

The complexity of the mouse BCR repertoire varies between animals. We generally recommend a minimum of 2×10^6 reads from an input of 10 ng PBMC RNA for heavy-chain and light-chain libraries. For libraries generated from >10 ng PBMC RNA, higher sequencing depth is recommended, as shown in Table 4 (using spleen as an example).

NOTE: The optimal conditions may vary for different sample types, sample masses, and sample complexities. To determine the optimal sequencing depth, we recommend trying a higher sequencing depth then downsampling to determine the minimum number of reads per library.

Table 4. Recommended sequencing depth for BCR libraries prepared from mouse spleen RNA.

Input	Heavy chain	Light chain
Spleen RNA (10 ng)	2×10^6 reads	2×10^6 reads
Spleen RNA (100 ng)	$\sim 6 \times 10^6$ reads	$\sim 6 \times 10^6$ reads
Spleen RNA (1 μ g)	$\sim 2.5 \times 10^7$ reads	$\sim 2.5 \times 10^7$ reads

Appendix D: Guidelines for Data Analysis

Upon completion of a sequencing run, data can be analyzed with our Cogent NGS Immune Profiler Software (CogentIP). To obtain CogentIP, please visit takarabio.com/ngs-immune-profiler. You can also generate tabulated outputs and publication-ready plots of CDR3 length distribution, V/D usage distribution, and clonotype diversity using our Cogent NGS Immune Viewer at takarabio.com/ngs-immune-viewer.

CogentIP can also be used to remove duplicated sequences and correct errors from the PCR amplification process through analysis of the 12-nucleotide (nt) UMI contained within the BCR library (Figure 4). Analysis of UMIs provides higher accuracy for clonotype diversity and abundance measurements.

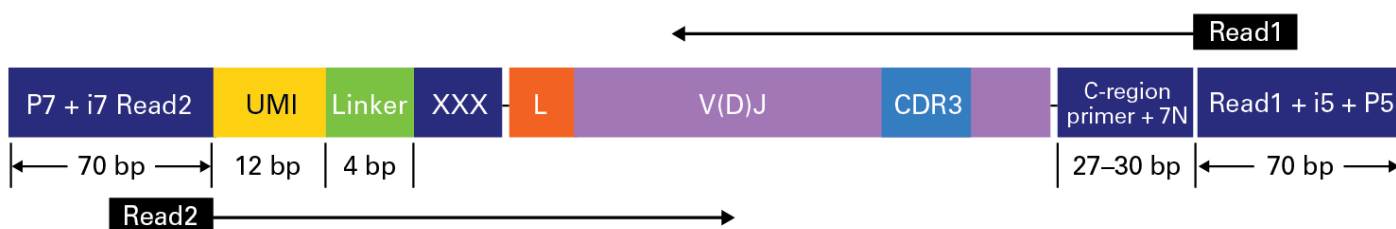


Figure 4. SMART-Seq Mouse BCR (with UMIs) library structure. The first 19 nucleotides from Read2 can be trimmed off if UMI analysis is not performed. "XXX" represents nontemplated nucleotides originating from the SMART UMI Oligo. "L" is the leader sequence.

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