

# Beginner's guide to using the DESeq2 package

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## Abstract

This vignette describes the statistical analysis of count matrices for systematic changes between conditions using the *DESeq2* package, and includes recommendations for producing count matrices from raw sequencing data. This vignette is designed for users who are perhaps new to analyzing RNA-Seq or high-throughput sequencing data in R, and so goes at a slower pace, explaining each step in detail. Another vignette, “Differential analysis of count data – the DESeq2 package” covers more of the advanced details at a faster pace.

**DESeq2 version:** 1.4.5

If you use *DESeq2* in published research, please cite:

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## 1 Introduction

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In this vignette, you will learn how to produce a read count table – such as arising from a summarized RNA-Seq experiment – analyze count tables for differentially expressed genes, visualize the results, add extra gene annotations, and cluster samples and genes using transformed counts.

## 2 Input data

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## 2.1 Preparing count matrices

As input, the *DESeq2* package expects count data as obtained, e.g., from RNA-Seq or another high-throughput sequencing experiment, in the form of a matrix of integer values. The value in the  $i$ -th row and the  $j$ -th column of the matrix tells how many reads have been mapped to gene  $i$  in sample  $j$ . Analogously, for other types of assays, the rows of the matrix might correspond e.g. to binding regions (with ChIP-Seq) or peptide sequences (with quantitative mass spectrometry).

The count values must be raw counts of sequencing reads. This is important for *DESeq2*'s statistical model to hold, as only the actual counts allow assessing the measurement precision correctly. Hence, please do not supply other quantities, such as (rounded) normalized counts, or counts of covered base pairs – this will only lead to nonsensical results.

## 2.2 Aligning reads to a reference

The computational analysis of an RNA-Seq experiment begins earlier however, with a set of FASTQ files, which contain the bases for each read and their quality scores. These reads must first be aligned to a reference genome or transcriptome. It is important to know if the sequencing experiment was single-end or paired-end, as the alignment software will require the user specify both FASTQ files for a paired-end experiment.

A number of software programs exist to align reads to the reference genome, and the development is too rapid for this document to provide a current list. We recommend reading benchmarking papers which discuss the advantages and disadvantages of each software, which include accuracy, ability to align reads over splice junctions, speed, memory footprint, and many other features.

We have experience using the TopHat2 spliced alignment software<sup>1</sup> [2] in combination with the Bowtie index available at the Illumina iGenomes page<sup>2</sup>. For full details on this software and on the iGenomes, users should follow the links to the manual and information provided in the links in the footnotes. For example, the paired-end RNA-Seq reads for the *parathyroidSE* package were aligned using TopHat2 with 8 threads, with the call:

```
tophat2 -o file_tophat_out -p 8 genome file_1.fastq file_2.fastq
samtools sort -n file_tophat_out/accepted_hits.bam _sorted
```

The second line sorts the reads by name rather than by genomic position, which is necessary for counting paired-end reads within Bioconductor. This command uses the SAMtools software<sup>3</sup> [3]. The BAM files for a number of sequencing runs can then be used to generate count matrices, as described in the following section.

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<sup>1</sup><http://tophat.cbcb.umd.edu/>

<sup>2</sup><http://tophat.cbcb.umd.edu/igenomes.html>

<sup>3</sup><http://samtools.sourceforge.net>

## 2.3 Counting reads in genes

Once the reads have been aligned, there are a number of tools which can be used to count the number of reads which can be unambiguously assigned to genomic features for each sample. These often take as input BAM or SAM alignment files and a file specifying the genomic features, e.g. GFF3 or GTF files specifying a gene model.

The following tools can be used generate count tables:

function	package	output	DESeq2 input function
<code>summarizeOverlaps</code>	<a href="#">GenomicAlignments</a> (Bioc)	<i>SummarizedExperiment</i>	<code>DESeqDataSet</code>
<code>htseq-count</code> <sup>[4]</sup>	<a href="#">HTSeq</a> (Python)	count files	<code>DESeqDataSetFromHTSeq</code>
<code>featureCounts</code> <sup>[5]</sup>	<a href="#">Rsubread</a> (Bioc)	count matrix	<code>DESeqDataSetFromMatrix</code>
<code>simpleRNASeq</code> <sup>[6]</sup>	<a href="#">easyRNASeq</a> (Bioc)	<i>SummarizedExperiment</i>	<code>DESeqDataSet</code>

In order to produce correct counts, it is important to know if the experiment was strand-specific or not. For example, `summarizeOverlaps` has the argument `ignore.strand`, which should be set to `TRUE` if the experiment was not strand-specific and `FALSE` if the experiment was strand-specific. Similarly, `htseq-count` has the argument `--stranded yes/no/reverse`, where strand-specific experiments should use `--stranded yes` and where `reverse` indicates that the positive strand reads should be counted to negative strand features.

The following example uses `summarizeOverlaps` for read counting, while produces a *SummarizedExperiment* object. This class of object contains a variety of information about an experiment, and will be described in more detail below. We will demonstrate using example BAM files from the [parathyroidSE](#) data package. First, we read in the gene model from a GTF file, using `makeTranscriptDbFromGFF`. Alternatively the `makeTranscriptDbFromBiomart` function can be used to automatically pull a gene model from Biomart. However, keeping the GTF file on hand has the advantage of bioinformatic reproducibility: the same gene model can be made again, while past versions of gene models might not always be available on Biomart. These GTF files can be downloaded from Ensembl's FTP site or other gene model repositories. The third line here produces a *GRangesList* of all the exons grouped by gene.

```
library( "GenomicFeatures" )
hse <- makeTranscriptDbFromGFF( "/path/to/your/genemodel.GTF", format="gtf" )
exonsByGene <- exonsBy( hse, by="gene" )
```

We specify the BAM files which will be used for counting.

```
fls <- list.files( "/path/to/bam/files", pattern="bam$", full=TRUE )
```

We indicate in Bioconductor that these `fls` are BAM files using the `BamFileList` function. Here we also specify details about how the BAM files should be treated, e.g., only process 100000 reads at a time.

```
library( "Rsamtools" )
bamLst <- BamFileList( fls, yieldSize=100000 )
```

We call `summarizeOverlaps` to count the reads. We use the counting mode `"Union"` which indicates that reads which overlap any portion of exactly one feature are counted. For more information

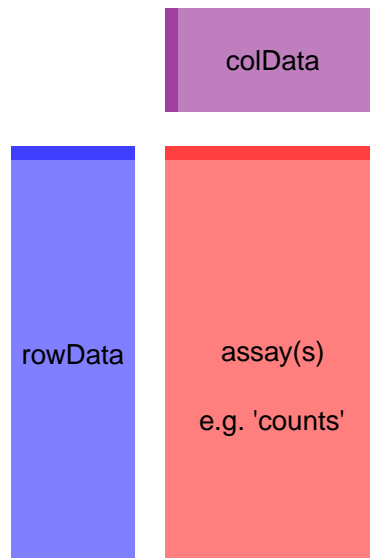


Figure 1: **Diagram of SummarizedExperiment** Here we show the component parts of a *SummarizedExperiment* object, and also its subclasses, such as the *DESeqDataSet* which is explained in the next section. The assay(s) (red block) contains the matrix (or matrices) of summarized values, the rowData (blue block) contains information about the genomic ranges, and the colData (purple block) contains information about the samples or experiments. The highlighted line in each block represents the first row (note that the first row of colData lines up with the first column of the assay).

tion on the various counting modes, see the help page for `summarizeOverlaps`. As this experiment was a paired-end, we specify `singleEnd=FALSE`. As it was not a strand-specific protocol, we specify `ignore.strand=TRUE`. `fragments=TRUE` indicates that we also want to count reads with unmapped pairs. This last argument is only for use with paired-end experiments.

```
library( "GenomicAlignments" )
se <- summarizeOverlaps( exonsByGene, bamLst,
                        mode="Union",
                        singleEnd=FALSE,
                        ignore.strand=TRUE,
                        fragments=TRUE )
```

This example code above actually only counts a small subset of reads from the original experiment: for 3 samples and for 100 genes. Nevertheless, we can still investigate the resulting *SummarizedExperiment* by looking at the counts in the assay slot, the phenotypic data about the samples in colData slot (in this case an empty *DataFrame*), and the data about the genes in the rowData slot.

```

se

## class: SummarizedExperiment
## dim: 100 3
## exptData(0):
## assays(1): counts
## rownames(100): ENSG000000000003 ENSG000000000005 ... ENSG00000005469
## ENSG000000005471
## rowData metadata column names(0):
## colnames(3): SRR479052.bam SRR479053.bam SRR479054.bam
## colData names(0):

head( assay(se) )

##           SRR479052.bam SRR479053.bam SRR479054.bam
## ENSG000000000003      0           0           1
## ENSG000000000005      0           0           0
## ENSG000000000419      0           0           0
## ENSG000000000457      0           1           0
## ENSG000000000460      0           0           0
## ENSG000000000938      0           0           0

colSums( assay(se) )

## SRR479052.bam SRR479053.bam SRR479054.bam
##           31           21           27

colData(se)

## DataFrame with 3 rows and 0 columns

rowData(se)

## GRangesList of length 100:
## $ENSG000000000003
## GRanges with 17 ranges and 2 metadata columns:
##           seqnames           ranges strand | exon_id exon_name
##           <Rle>           <IRanges> <Rle> | <integer> <character>
## [1]           X [99883667, 99884983]     - |    664095 ENSE00001459322
## [2]           X [99885756, 99885863]     - |    664096 ENSE00000868868
## [3]           X [99887482, 99887565]     - |    664097 ENSE00000401072
## [4]           X [99887538, 99887565]     - |    664098 ENSE00001849132
## [5]           X [99888402, 99888536]     - |    664099 ENSE00003554016
## ...           ...           ...     ... |    ...      ...
## [13]          X [99890555, 99890743]     - |    664106 ENSE00003512331
## [14]          X [99891188, 99891686]     - |    664108 ENSE00001886883
## [15]          X [99891605, 99891803]     - |    664109 ENSE00001855382
## [16]          X [99891790, 99892101]     - |    664110 ENSE00001863395
## [17]          X [99894942, 99894988]     - |    664111 ENSE00001828996

```

```
##
## ...
## <99 more elements>
## ---
## seqlengths:
##           1           2 ...      LRG_98      LRG_99
##      249250621    243199373 ...    18750     13294
```

Note that the `rowData` slot is a *GRangesList*, which contains all the information about the exons for each gene, i.e., for each row of the count table.

This *SummarizedExperiment* object `se` is then all we need to start our analysis. In the following section we will show how to create the data object which is used in *DESeq2*, either using the *SummarizedExperiment*, or in general, a count table which has been loaded into R.

## 2.4 Experiment data

### 2.4.1 The *DESeqDataSet*, column metadata, and the design formula

Each Bioconductor software package often has a special class of data object, which contains special slots and requirements. The data object class in *DESeq2* is the *DESeqDataSet*, which is built on top of the *SummarizedExperiment*. One main difference is that the assay slot is instead accessed using the count accessor, and the values in this matrix must be non-negative integers.

A second difference is that the *DESeqDataSet* has an associated “design formula”. The design is specified at the beginning of the analysis, as this will inform many of the *DESeq2* functions how to treat the samples in the analysis (one exception is the size factor estimation – adjustment for differing library sizes – which does not depend on the design formula). The design formula tells which variables in the column metadata table (`colData`) specify the experimental design and how these factors should be used in the analysis.

The simplest design formula for differential expression would be `~ condition`, where `condition` is a column in `colData(dds)` which specifies which of two (or more groups) the samples belong to. For the parathyroid experiment, we will specify `~ patient + treatment`, which means that we want to test for the effect of treatment (the last factor), controlling for the effect of patient (the first factor).

You can use R's formula notation to express any experimental design that can be described within an ANOVA-like framework. Note that *DESeq2* uses the same kind of formula as in base R, e.g., for use by the `lm` function. If the question of interest is whether a fold change due to treatment is different across groups, for example across patients, “interaction terms” can be included using models such as `~ patient + treatment + patient:treatment`. More complex designs such as these are covered in the other *DESeq2* vignette.

In the following section, we will demonstrate the construction of the *DESeqDataSet* from two starting points:

1. from a *SummarizedExperiment* object created by, e.g., `summarizeOverlaps` in the above example

2. more generally, from a count table (i.e. matrix) and a column metadata table which have been loaded into R

For a full example of using the HTSeq Python package<sup>4</sup> for read counting, please see the [pasilla](#) or [parathyroid](#) data package. For an example of generating the *DESeqDataSet* from files produced by htseq-count, please see the other *DESeq2* vignette.

### 2.4.2 Starting from SummarizedExperiment

We load a prepared *SummarizedExperiment*, which was generated using `summarizeOverlaps` from publicly available data from the article by Felix Haglund et al., “Evidence of a Functional Estrogen Receptor in Parathyroid Adenomas”, J Clin Endocrin Metab, Sep 2012<sup>5</sup>. Details on the generation of this object can be found in the vignette for the [parathyroidSE](#) package.

The purpose of the experiment was to investigate the role of the estrogen receptor in parathyroid tumors. The investigators derived primary cultures of parathyroid adenoma cells from 4 patients. These primary cultures were treated with diethylstilbestrol (DES), an estrogen receptor  $\beta$  agonist, or with 4-hydroxytamoxifen (OHT). RNA was extracted at 24 hours and 48 hours from cultures under treatment and control. The blocked design of the experiment allows for statistical analysis of the treatment effects while controlling for patient-to-patient variation.

We first load the *DESeq2* package and the data package [parathyroidSE](#), which contains the example data set.

```
library( "DESeq2" )
library( "parathyroidSE" )
```

The data command loads a preconstructed data object:

```
data( "parathyroidGenesSE" )
se <- parathyroidGenesSE
colnames(se) <- se$run
```

Supposing we have constructed a *SummarizedExperiment* using one of the methods described in the previous section, we now ensure that the object contains the necessary information about the samples, i.e., a table with metadata on the count table's columns stored in the `colData` slot:

```
colData(se)[1:5,1:4]

## DataFrame with 5 rows and 4 columns
##           run experiment patient treatment
##           <character>   <factor> <factor> <factor>
## SRR479052   SRR479052   SRX140503      1   Control
## SRR479053   SRR479053   SRX140504      1   Control
## SRR479054   SRR479054   SRX140505      1     DPN
## SRR479055   SRR479055   SRX140506      1     DPN
```

<sup>4</sup>described in [4]

<sup>5</sup><http://www.ncbi.nlm.nih.gov/pubmed/23024189>



```
## SRR479056    SRR479056    SRX140507        1        OHT
```

This object does, because it was prepared so, as can be seen in the [parathyroidSE](#) vignette. However, users will most likely have to add pertinent sample/phenotypic information for the experiment at this stage. We highly recommend keeping this information in a comma-separated value (CSV) or tab-separated value (TSV) file, which can be exported from an Excel spreadsheet. The advantage of this over typing out the characters into an R script, is that while scrolling through a script, accidentally typed spaces or characters could lead to changes to the sample phenotypic information, leading to spurious results.

Suppose we have a CSV file which contains such data, we could read this file in using the base R `read.csv` function:

```
sampleInfo <- read.csv( "/path/to/file.CSV" )
```

Here we show a toy example of such a table. Note that the order is not the same as in the *SummarizedExperiment*. Here instead of run, most users will have the filename of the BAM files used for counting. We convert the `sampleInfo` object into a *DataFrame* which is the format of the `colData`.

```
head( sampleInfo )

##          run pheno
## 1 SRR479078 pheno1
## 2 SRR479077 pheno2
## 3 SRR479076 pheno1
## 4 SRR479075 pheno2
## 5 SRR479074 pheno1
## 6 SRR479073 pheno2

head( colnames(se) )

## [1] "SRR479052" "SRR479053" "SRR479054" "SRR479055" "SRR479056" "SRR479057"

sampleInfo <- DataFrame( sampleInfo )
```

We create an index which will put them in the same order: the *SummarizedExperiment* comes first because this is the sample order we want to achieve. We then check to see that we have lined them up correctly, and then we can add the new data to the existing `colData`.

```
seIdx <- match(colnames(se), sampleInfo$run)
head( cbind( colData(se)[ , 1:3 ], sampleInfo[ seIdx, ] ) )

## DataFrame with 6 rows and 5 columns
##          run experiment patient    run.1    pheno
##          <character>  <factor> <factor>  <factor> <factor>
## SRR479052    SRR479052    SRX140503        1 SRR479052    pheno1
## SRR479053    SRR479053    SRX140504        1 SRR479053    pheno2
## SRR479054    SRR479054    SRX140505        1 SRR479054    pheno1
## SRR479055    SRR479055    SRX140506        1 SRR479055    pheno2
## SRR479056    SRR479056    SRX140507        1 SRR479056    pheno1
```

The following line builds the *DESeqDataSet* from a *SummarizedExperiment* *se* and specifying a design formula, as described in the previous section. The names of variables used in the design formula must be the names of columns in the *colData* of *se*.

### 2.4.3 Starting from count tables

The information in a *SummarizedExperiment* object can be accessed with accessor functions. For example, to see the actual data, i.e., here, the read counts, we use the `assay` function. (The `head` function restricts the output to the first few lines.)

```
countdata <- assay( parathyroidGenesSE )
head( countdata )

##           [,1] [,2] [,3] [,4] [,5] [,6] [,7] [,8] [,9] [,10] [,11] [,12]
## ENSG000000000003  792 1064  444  953  519  855  413  365  278 1173  463  316
## ENSG000000000005    4    1    2    3    3    1    0    1    0    0    0    0
## ENSG000000000419  294  282  164  263  179  217  277  204  189  601  257  183
## ENSG000000000457  156  184   93  145   75  122  228  171  116  422  182  122
## ENSG000000000460  396  207  210  212  221  173  611  199  426 1391  286  417
## ENSG000000000938    3    8    2    5    0    4   13   22    3   38   13   10
##           [,13] [,14] [,15] [,16] [,17] [,18] [,19] [,20] [,21] [,22]
## ENSG000000000003   987   424   305   391   586   714   957   346   433   402
## ENSG000000000005    0    0    0    0    0    0    1    0    0    0
## ENSG000000000419   588   275   263   281   406   568   764   288   259   250
## ENSG000000000457   441   211   131   115   196   266   347   133   168   148
## ENSG000000000460  1452   238   188   102   389   294   778   162    85   339
## ENSG000000000938    26   13    7    3   10   18   15    7    8    7
##           [,23] [,24] [,25] [,26] [,27]
## ENSG000000000003   277   511   366   271   492
## ENSG000000000005    0    0    0    0    0
## ENSG000000000419   147   271   227   197   363
## ENSG000000000457    83   184   136   118   195
## ENSG000000000460    75   154   314   117   233
```

```
## ENSG000000000938      5      13      8      7      8
```

In this count table, each row represents an Ensembl gene, each column a sequenced RNA library, and the values give the raw numbers of sequencing reads that were mapped to the respective gene in each library.

We also have metadata on each of the samples (the “columns” of the count table):

```
coldata <- colData( parathyroidGenesSE )
rownames( coldata ) <- coldata$run
colnames( countdata ) <- coldata$run

head( coldata[ , c( "patient", "treatment", "time" ) ] )

## DataFrame with 6 rows and 3 columns
##           patient treatment      time
##           <factor> <factor> <factor>
## SRR479052         1   Control    24h
## SRR479053         1   Control    48h
## SRR479054         1      DPN     24h
## SRR479055         1      DPN     48h
## SRR479056         1      OHT     24h
## SRR479057         1      OHT     48h
```

We now have all the ingredients to prepare our data object in a form that is suitable for analysis, namely:

- countdata: a table with the read counts
- coldata: a table with metadata on the count table's columns

To now construct the data object from the matrix of counts and the metadata table, we use:

```
ddsFullCountTable <- DESeqDataSetFromMatrix(
  countData = countdata,
  colData = coldata,
  design = ~ patient + treatment)
ddsFullCountTable

## class: DESeqDataSet
## dim: 63193 27
## exptData(0):
## assays(1): counts
## rownames(63193): ENSG000000000003 ENSG000000000005 ... LRG_98 LRG_99
## rowData metadata column names(0):
## colnames(27): SRR479052 SRR479053 ... SRR479077 SRR479078
## colData names(8): run experiment ... study sample
```

We will continue with the object generated from the *SummarizedExperiment* section.

## 2.5 Collapsing technical replicates

There are a number of samples which were sequenced in multiple runs. For example, sample SRS308873 was sequenced twice. To see, we list the respective columns of the `colData`. (The use of `as.data.frame` forces R to show us the full list, not just the beginning and the end as before.)

```
as.data.frame( colData( ddsFull ) [ ,c("sample","patient","treatment","time") ] )
```

##		sample	patient	treatment	time
##	SRR479052	SRS308865	1	Control	24h
##	SRR479053	SRS308866	1	Control	48h
##	SRR479054	SRS308867	1	DPN	24h
##	SRR479055	SRS308868	1	DPN	48h
##	SRR479056	SRS308869	1	OHT	24h
##	SRR479057	SRS308870	1	OHT	48h
##	SRR479058	SRS308871	2	Control	24h
##	SRR479059	SRS308872	2	Control	48h
##	SRR479060	SRS308873	2	DPN	24h
##	SRR479061	SRS308873	2	DPN	24h
##	SRR479062	SRS308874	2	DPN	48h
##	SRR479063	SRS308875	2	OHT	24h
##	SRR479064	SRS308875	2	OHT	24h
##	SRR479065	SRS308876	2	OHT	48h
##	SRR479066	SRS308877	3	Control	24h
##	SRR479067	SRS308878	3	Control	48h
##	SRR479068	SRS308879	3	DPN	24h
##	SRR479069	SRS308880	3	DPN	48h
##	SRR479070	SRS308881	3	OHT	24h
##	SRR479071	SRS308882	3	OHT	48h
##	SRR479072	SRS308883	4	Control	48h
##	SRR479073	SRS308884	4	DPN	24h
##	SRR479074	SRS308885	4	DPN	48h
##	SRR479075	SRS308885	4	DPN	48h
##	SRR479076	SRS308886	4	OHT	24h
##	SRR479077	SRS308887	4	OHT	48h
##	SRR479078	SRS308887	4	OHT	48h

We recommend to first add together technical replicates (i.e., libraries derived from the same samples), such that we have one column per sample. We have implemented a convenience function for this, which can take an object, either *SummarizedExperiment* or *DESeqDataSet*, and a grouping factor, in this case the sample name, and return the object with the counts summed up for each unique sample. This will also rename the columns of the object, such that they match the unique names which were used in the grouping factor. Optionally, we can provide a third argument, `run`, which can be used to paste together the names of the runs which were collapsed to create the new object. Note that `dds$variable` is equivalent to `colData(dds)$variable`.

```
ddsCollapsed <- collapseReplicates( ddsFull,
                                   groupby = ddsFull$sample,
                                   run = ddsFull$run )
head( as.data.frame( colData(ddsCollapsed)[ ,c("sample","runsCollapsed") ] ), 12 )
```

##	sample	runsCollapsed
##	SRS308865	SRR479052
##	SRS308866	SRR479053
##	SRS308867	SRR479054
##	SRS308868	SRR479055
##	SRS308869	SRR479056
##	SRS308870	SRR479057
##	SRS308871	SRR479058
##	SRS308872	SRR479059
##	SRS308873	SRR479060,SRR479061
##	SRS308874	SRR479062
##	SRS308875	SRR479063,SRR479064
##	SRS308876	SRR479065

We can confirm that the counts for the new object are equal to the summed up counts of the columns that had the same value for the grouping factor:

```
original <- rowSums( counts(ddsFull)[ , ddsFull$sample == "SRS308873" ] )
all( original == counts(ddsCollapsed)[ ,"SRS308873" ] )
## [1] TRUE
```

## 3 Running the DESeq2 pipeline

---

Here we will analyze a subset of the samples, namely those taken after 48 hours, with either control, DPN or OHT treatment, taking into account the multifactor design.

### 3.1 Preparing the data object for the analysis of interest

First we subset the relevant columns from the full dataset:

```
dds <- ddsCollapsed[ , ddsCollapsed$time == "48h" ]
```

Sometimes it is necessary to drop levels of the factors, in case that all the samples for one or more levels of a factor in the design have been removed. If time were included in the design formula, the following code could be used to take care of dropped levels in this column.

```
dds$time <- droplevels( dds$time )
```

It will be convenient to make sure that Control is the *first* level in the treatment factor, so that the default log2 fold changes are calculated as treatment over control and not the other way around. The function `relevel` achieves this:

```
dds$treatment <- relevel( dds$treatment, "Control" )
```

A quick check whether we now have the right samples:

```
as.data.frame( colData(dds) )
```

##		run	experiment	patient	treatment	time	submission	study
##	SRS308866	SRR479053	SRX140504	1	Control	48h	SRA051611	SRP012167
##	SRS308868	SRR479055	SRX140506	1	DPN	48h	SRA051611	SRP012167
##	SRS308870	SRR479057	SRX140508	1	OHT	48h	SRA051611	SRP012167
##	SRS308872	SRR479059	SRX140510	2	Control	48h	SRA051611	SRP012167
##	SRS308874	SRR479062	SRX140512	2	DPN	48h	SRA051611	SRP012167
##	SRS308876	SRR479065	SRX140514	2	OHT	48h	SRA051611	SRP012167
##	SRS308878	SRR479067	SRX140516	3	Control	48h	SRA051611	SRP012167
##	SRS308880	SRR479069	SRX140518	3	DPN	48h	SRA051611	SRP012167
##	SRS308882	SRR479071	SRX140520	3	OHT	48h	SRA051611	SRP012167
##	SRS308883	SRR479072	SRX140521	4	Control	48h	SRA051611	SRP012167
##	SRS308885	SRR479074	SRX140523	4	DPN	48h	SRA051611	SRP012167
##	SRS308887	SRR479077	SRX140525	4	OHT	48h	SRA051611	SRP012167
##		sample	run.1		runsCollapsed			
##	SRS308866	SRS308866	SRR479053		SRR479053			
##	SRS308868	SRS308868	SRR479055		SRR479055			
##	SRS308870	SRS308870	SRR479057		SRR479057			
##	SRS308872	SRS308872	SRR479059		SRR479059			
##	SRS308874	SRS308874	SRR479062		SRR479062			
##	SRS308876	SRS308876	SRR479065		SRR479065			
##	SRS308878	SRS308878	SRR479067		SRR479067			
##	SRS308880	SRS308880	SRR479069		SRR479069			
##	SRS308882	SRS308882	SRR479071		SRR479071			
##	SRS308883	SRS308883	SRR479072		SRR479072			
##	SRS308885	SRS308885	SRR479074	SRR479074,SRR479075				
##	SRS308887	SRS308887	SRR479077	SRR479077,SRR479078				

## 3.2 Running the pipeline

Finally, we are ready to run the differential expression pipeline. With the data object prepared, the *DESeq2* analysis can now be run with a single call to the function `DESeq`:

```
dds <- DESeq(dds)
```

This function will print out a message for the various steps it performs. These are described in more detail in the manual page for `DESeq`, which can be accessed by typing `?DESeq`. Briefly these are: the

estimation of size factors (which control for differences in the library size of the sequencing experiments), the estimation of dispersion for each gene, and fitting a generalized linear model.

A *DESeqDataSet* is returned which contains all the fitted information within it, and the following section describes how to extract out results tables of interest from this object.

### 3.3 Inspecting the results table

Calling `results` without any arguments will extract the estimated log2 fold changes and *p* values for the last variable in the design formula. If there are more than 2 levels for this variable – as is the case in this analysis – `results` will extract the results table for a comparison of the last level over the first level. The following section describes how to extract other comparisons.

```
res <- results( dds )
res
```

```
## log2 fold change (MAP): treatment OHT vs Control
## Wald test p-value: treatment OHT vs Control
## DataFrame with 4000 rows and 6 columns
##
```

	baseMean	log2FoldChange	lfcSE	stat	pvalue
##	<numeric>	<numeric>	<numeric>	<numeric>	<numeric>
## ENSG000000000003	616.390	-0.04917	0.0868	-0.5667	0.57094
## ENSG000000000005	0.553	-0.08167	0.1658	-0.4927	0.62225
## ENSG000000000419	305.400	0.10779	0.0945	1.1405	0.25410
## ENSG000000000457	184.341	0.00189	0.1196	0.0158	0.98742
## ENSG000000000460	208.305	0.44504	0.1371	3.2456	0.00117
## ...	...	...	...	...	...
## ENSG00000112851	1071.1	-0.04915	0.0628	-0.7827	0.434
## ENSG00000112852	11.8	0.24846	0.3184	0.7803	0.435
## ENSG00000112855	257.8	0.03485	0.1019	0.3421	0.732
## ENSG00000112874	311.4	0.00817	0.1058	0.0772	0.938
## ENSG00000112877	44.4	-0.01252	0.2085	-0.0600	0.952
##	padj				
##	<numeric>				
## ENSG000000000003	0.9984				
## ENSG000000000005	NA				
## ENSG000000000419	0.9894				
## ENSG000000000457	0.9984				
## ENSG000000000460	0.0995				
## ...	...				
## ENSG00000112851	0.998				
## ENSG00000112852	NA				
## ENSG00000112855	0.998				
## ENSG00000112874	0.998				
## ENSG00000112877	NA				

As `res` is a `DataFrame` object, it carries metadata with information on the meaning of the columns:

```
mcols(res, use.names=TRUE)

## DataFrame with 6 rows and 2 columns
##               type               description
##      <character>      <character>
## baseMean      intermediate      the base mean over all rows
## log2FoldChange results log2 fold change (MAP): treatment OHT vs Control
## lfcSE          results          standard error: treatment OHT vs Control
## stat           results          Wald statistic: treatment OHT vs Control
## pvalue         results          Wald test p-value: treatment OHT vs Control
## padj           results          BH adjusted p-values
```

The first column, `baseMean`, is just the average of the normalized count values, dividing by size factors, taken over all samples. The remaining four columns refer to a specific *contrast*, namely the comparison of the levels *DPN* versus *Control* of the factor variable *treatment*. See the help page for `results` (by typing `?results`) for information on how to obtain other contrasts.

The column `log2FoldChange` is the effect size estimate. It tells us how much the gene's expression seems to have changed due to treatment with DPN in comparison to control. This value is reported on a logarithmic scale to base 2: for example, a log2 fold change of 1.5 means that the gene's expression is increased by a multiplicative factor of  $2^{1.5} \approx 2.82$ .

Of course, this estimate has an uncertainty associated with it, which is available in the column `lfcSE`, the standard error estimate for the log2 fold change estimate. We can also express the uncertainty of a particular effect size estimate as the result of a statistical test. The purpose of a test for differential expression is to test whether the data provides sufficient evidence to conclude that this value is really different from zero. *DESeq2* performs for each gene a *hypothesis test* to see whether evidence is sufficient to decide against the *null hypothesis* that there is no effect of the treatment on the gene and that the observed difference between treatment and control was merely caused by experimental variability (i. e., the type of variability that you can just as well expect between different samples in the same treatment group). As usual in statistics, the result of this test is reported as a *p value*, and it is found in the column `pvalue`. (Remember that a *p value* indicates the probability that a fold change as strong as the observed one, or even stronger, would be seen under the situation described by the null hypothesis.)

We note that a subset of the *p values* in `res` are `NA` ("not available"). This is *DESeq2*'s way of reporting that all counts for this gene were zero, and hence no test was applied. In addition, *p values* can be assigned `NA` if the gene was excluded from analysis because it contained an extreme count outlier. For more information, see the outlier detection section of the advanced vignette.

### 3.4 Other comparisons

In general, the results for a comparison of any two levels of a variable can be extracted using the `contrast` argument to `results`. The user should specify three values: the name of the variable, the



name of the level in the numerator, and the name of the level in the denominator. Here we extract results for the log2 of the fold change of DPN / Control.

```
res <- results( dds, contrast = c("treatment", "DPN", "Control") )
res
```

```
## log2 fold change (MAP): treatment DPN vs Control
## Wald test p-value: treatment DPN vs Control
## DataFrame with 4000 rows and 6 columns
##
```

	baseMean	log2FoldChange	lfcSE	stat	pvalue
##	<numeric>	<numeric>	<numeric>	<numeric>	<numeric>
## ENSG000000000003	616.390	-0.01579	0.0856	-0.1844	0.8537
## ENSG000000000005	0.553	-0.00836	0.1703	-0.0491	0.9609
## ENSG000000000419	305.400	-0.01647	0.0935	-0.1762	0.8601
## ENSG000000000457	184.341	-0.09290	0.1180	-0.7874	0.4310
## ENSG000000000460	208.305	0.33561	0.1360	2.4669	0.0136
## ...	...	...	...	...	...
## ENSG00000112851	1071.1	-0.0420	0.062	-0.677	0.498
## ENSG00000112852	11.8	0.2125	0.317	0.671	0.502
## ENSG00000112855	257.8	-0.0832	0.101	-0.826	0.409
## ENSG00000112874	311.4	0.1196	0.104	1.153	0.249
## ENSG00000112877	44.4	0.0542	0.204	0.266	0.790
##	padj				
##	<numeric>				
## ENSG000000000003	0.971				
## ENSG000000000005	NA				
## ENSG000000000419	0.973				
## ENSG000000000457	0.879				
## ENSG000000000460	0.250				
## ...	...				
## ENSG00000112851	0.892				
## ENSG00000112852	NA				
## ENSG00000112855	0.872				
## ENSG00000112874	0.777				
## ENSG00000112877	NA				

If results for an interaction term are desired, the `name` argument of `results` should be used. Please see the more advanced vignette for more details.

### 3.5 Multiple testing

Novices in high-throughput biology often assume that thresholding these  $p$  values at a low value, say 0.01, as is often done in other settings, would be appropriate – but it is not. We briefly explain why:

There are 121 genes with a  $p$  value below 0.01 among the 4000 genes, for which the test succeeded in reporting a  $p$  value:

```
sum( res$pvalue < 0.01, na.rm=TRUE )
## [1] 121
table( is.na(res$pvalue) )
##
## FALSE
## 4000
```

Now, assume for a moment that the null hypothesis is true for all genes, i.e., no gene is affected by the treatment with DPN. Then, by the definition of *p value*, we expect up to 1% of the genes to have a *p* value below 0.01. This amounts to 40 genes. If we just considered the list of genes with a *p* value below 0.01 as differentially expressed, this list should therefore be expected to contain up to  $40/121 = 33\%$  false positives!

DESeq2 uses the so-called Benjamini-Hochberg (BH) adjustment; in brief, this method calculates for each gene an *adjusted p value* which answers the following question: if one called significant all genes with a *p* value less than or equal to this gene's *p* value threshold, what would be the fraction of false positives (the *false discovery rate*, FDR) among them (in the sense of the calculation outlined above)? These values, called the BH-adjusted *p* values, are given in the column *padj* of the results object.

Hence, if we consider a fraction of 10% false positives acceptable, we can consider all genes with an *adjusted p* value below  $10\%=0.1$  as significant. How many such genes are there?

```
sum( res$padj < 0.1, na.rm=TRUE )
## [1] 73
```

We subset the results table to these genes and then sort it by the log2 fold change estimate to get the significant genes with the strongest down-regulation

```
resSig <- res[ which(res$padj < 0.1), ]
head( resSig[ order( resSig$log2FoldChange ), ] )

## log2 fold change (MAP): treatment DPN vs Control
## Wald test p-value: treatment DPN vs Control
## DataFrame with 6 rows and 6 columns
##           baseMean log2FoldChange      lfcSE      stat      pvalue
##           <numeric>      <numeric> <numeric> <numeric> <numeric>
## ENSG000000091137      1150        -0.642    0.100     -6.39  1.65e-10
## ENSG000000041982      1384        -0.639    0.173     -3.69  2.24e-04
## ENSG000000105655      5217        -0.546    0.182     -3.00  2.70e-03
## ENSG000000035928      2180        -0.543    0.153     -3.55  3.87e-04
## ENSG000000111799       822        -0.527    0.108     -4.89  1.03e-06
## ENSG000000107518       198        -0.481    0.143     -3.37  7.56e-04
##                padj
##                <numeric>
## ENSG000000091137  1.32e-07
```

```
## ENSG000000041982 1.99e-02
## ENSG000000105655 9.25e-02
## ENSG000000035928 3.09e-02
## ENSG000000111799 2.76e-04
## ENSG000000107518 4.32e-02
```

and with the strongest upregulation

```
tail( resSig[ order( resSig$log2FoldChange ), ] )

## log2 fold change (MAP): treatment DPN vs Control
## Wald test p-value: treatment DPN vs Control
## DataFrame with 6 rows and 6 columns
##           baseMean log2FoldChange      lfcSE      stat      pvalue
##           <numeric>      <numeric> <numeric> <numeric> <numeric>
## ENSG000000044574      4691          0.482    0.0642      7.51 5.87e-14
## ENSG000000103942       238          0.486    0.1404      3.46 5.36e-04
## ENSG000000070669       284          0.520    0.1521      3.42 6.31e-04
## ENSG000000103257       169          0.781    0.1544      5.06 4.22e-07
## ENSG000000101255       256          0.837    0.1590      5.27 1.40e-07
## ENSG000000092621       561          0.873    0.1203      7.26 3.99e-13
##           padj
##           <numeric>
## ENSG000000044574 1.41e-10
## ENSG000000103942 4.02e-02
## ENSG000000070669 4.21e-02
## ENSG000000103257 1.26e-04
## ENSG000000101255 4.79e-05
## ENSG000000092621 4.79e-10
```

### 3.6 Diagnostic plots

A so-called MA plot provides a useful overview for an experiment with a two-group comparison:

```
plotMA( res, ylim = c(-1, 1) )
```

The plot (Fig. 2) represents each gene with a dot. The  $x$  axis is the average expression over all samples, the  $y$  axis the log2 fold change between treatment and control. Genes with an adjusted  $p$  value below a threshold (here 0.1, the default) are shown in red.

This plot demonstrates that only genes with a large average normalized count contain sufficient information to yield a significant call.

Also note *DESeq2*'s shrinkage estimation of log fold changes (LFCs): When count values are too low to allow an accurate estimate of the LFC, the value is “shrunk” towards zero to avoid that these values, which otherwise would frequently be unrealistically large, dominate the top-ranked log fold changes.

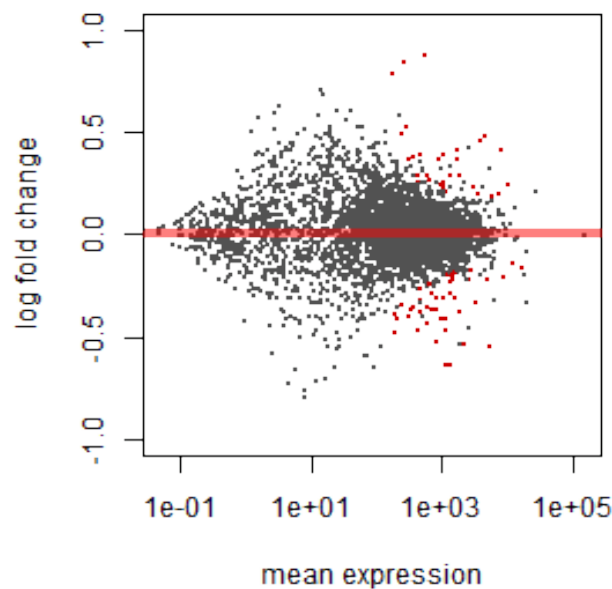


Figure 2: **MA-plot** The MA-plot shows the log<sub>2</sub> fold changes from the treatment over the mean of normalized counts, i.e. the average of counts normalized by size factor. The *DESeq2* package incorporates a prior on log<sub>2</sub> fold changes, resulting in moderated estimates from genes with low counts and highly variable counts, as can be seen by the narrowing of spread of points on the left side of the plot.

Whether a gene is called significant depends not only on its LFC but also on its within-group variability, which *DESeq2* quantifies as the *dispersion*. For strongly expressed genes, the dispersion can be understood as a squared coefficient of variation: a dispersion value of 0.01 means that the gene's expression tends to differ by typically  $\sqrt{0.01} = 10\%$  between samples of the same treatment group. For weak genes, the Poisson noise is an additional source of noise, which is added to the dispersion.

The function `plotDispEsts` visualizes *DESeq2*'s dispersion estimates:

```
plotDispEsts( dds, ylim = c(1e-6, 1e1) )
```

The black points are the dispersion estimates for each gene as obtained by considering the information from each gene separately. Unless one has many samples, these values fluctuate strongly around their true values. Therefore, we fit the red trend line, which shows the dispersions' dependence on the mean, and then shrink each gene's estimate towards the red line to obtain the final estimates (blue points) that are then used in the hypothesis test. The blue circles above the main "cloud" of points are genes which have high gene-wise dispersion estimates which are labelled as dispersion outliers. These estimates are therefore not shrunk toward the fitted trend line.

Another useful diagnostic plot is the histogram of the *p* values (Fig. 4).

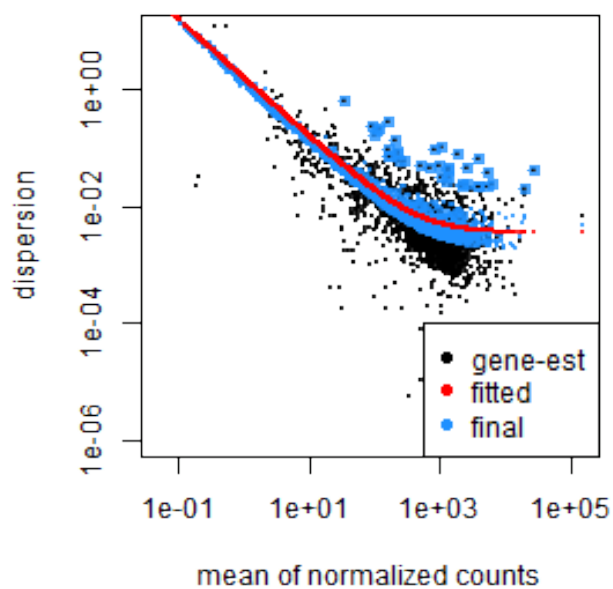


Figure 3: **Plot of dispersion estimates** See text for details

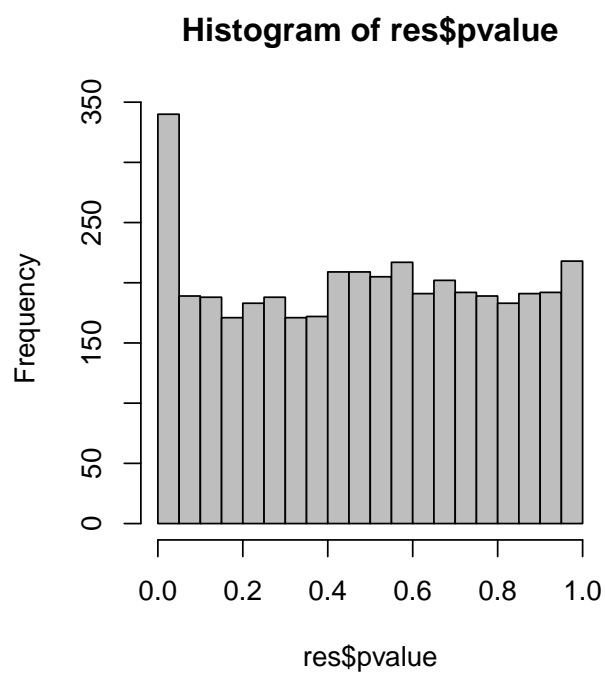


Figure 4: **Histogram** of the  $p$  values returned by the test for differential expression

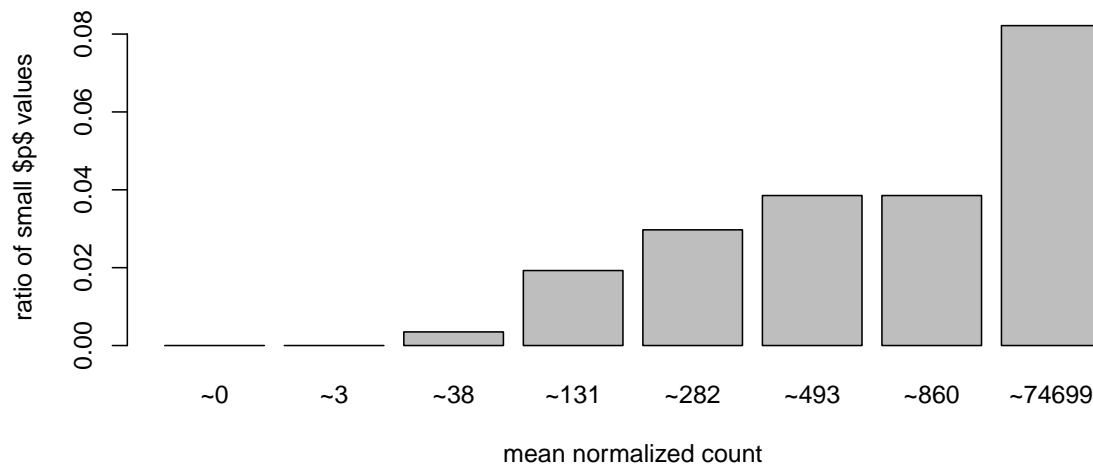


Figure 5: **Ratio of small  $p$  values** for groups of genes binned by mean normalized count

```
hist( res$pvalue, breaks=20, col="grey" )
```

## 4 Independent filtering

The MA plot (Figure 2) highlights an important property of RNA-Seq data. For weakly expressed genes, we have no chance of seeing differential expression, because the low read counts suffer from so high Poisson noise that any biological effect is drowned in the uncertainties from the read counting. We can also show this by examining the ratio of small  $p$  values (say, less than, 0.01) for genes binned by mean normalized count:

```
# create bins using the quantile function
qs <- c( 0, quantile( res$baseMean[res$baseMean > 0], 0:7/7 ) )
# "cut" the genes into the bins
bins <- cut( res$baseMean, qs )
# rename the levels of the bins using the middle point
levels(bins) <- paste0("~", round(.5*qs[-1] + .5*qs[-length(qs)]))
# calculate the ratio of $p$ values less than .01 for each bin
ratios <- tapply( res$pvalue, bins, function(p) mean( p < .01, na.rm=TRUE ) )
# plot these ratios
barplot(ratios, xlab="mean normalized count", ylab="ratio of small $p$ values")
```

At first sight, there may seem to be little benefit in filtering out these genes. After all, the test found them to be non-significant anyway. However, these genes have an influence on the multiple testing adjustment, whose performance improves if such genes are removed. By removing the weakly-expressed

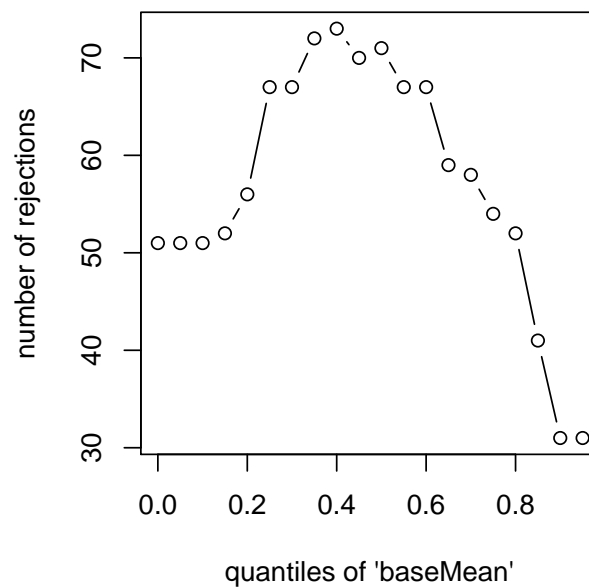


Figure 6: **Independent filtering.** *DESeq2* automatically determines a threshold, filtering on mean normalized count, which maximizes the number of genes which will have an adjusted  $p$  value less than a critical value.

genes from the input to the FDR procedure, we can find more genes to be significant among those which we keep, and so improved the power of our test. This approach is known as *independent filtering*.

The *DESeq2* software automatically performs independent filtering which maximizes the number of genes which will have adjusted  $p$  value less than a critical value (by default,  $\alpha$  is set to 0.1). This automatic independent filtering is performed by, and can be controlled by, the `results` function. We can observe how the number of rejections changes for various cutoffs based on mean normalized count. The following optimal threshold and table of possible values is stored as an *attribute* of the results object.

```
attr(res, "filterThreshold")
## 40%
## 165

plot(attr(res, "filterNumRej"), type="b",
      xlab="quantiles of 'baseMean'",
      ylab="number of rejections")
```

The term *independent* highlights an important caveat. Such filtering is permissible only if the filter criterion is independent of the actual test statistic [7]. Otherwise, the filtering would invalidate the test and consequently the assumptions of the BH procedure. This is why we filtered on the average over *all*

samples: this filter is blind to the assignment of samples to the treatment and control group and hence independent.

## 4.1 Adding gene names

Our result table only uses Ensembl gene IDs, but gene names may be more informative. Bioconductor's *biomaRt* package can help with mapping various ID schemes to each other.

First, we split up the rownames of the results object, which contain ENSEMBL gene ids, separated by the plus sign, +. The following code then takes the first id for each gene by invoking the open square bracket function "[" and the argument, 1.

```
res$ensembl <- sapply( strsplit( rownames(res), split="\\+" ), "[", 1 )
```

The following chunk of code uses the ENSEMBL mart, querying with the ENSEMBL gene id and requesting the Entrez gene id and HGNC gene symbol.

```
library( "biomaRt" )
ensembl = useMart( "ensembl", dataset = "hsapiens_gene_ensembl" )
genemap <- getBM( attributes = c("ensembl_gene_id", "entrezgene", "hgnc_symbol"),
                  filters = "ensembl_gene_id",
                  values = res$ensembl,
                  mart = ensembl )
idx <- match( res$ensembl, genemap$ensembl_gene_id )
res$entrez <- genemap$entrezgene[ idx ]
res$hgnc_symbol <- genemap$hgnc_symbol[ idx ]
```

Now the results have the desired external gene ids:

```
head(res,4)

## log2 fold change (MAP): treatment DPN vs Control
## Wald test p-value: treatment DPN vs Control
## DataFrame with 4 rows and 9 columns
##           baseMean log2FoldChange      lfcSE      stat      pvalue
##           <numeric>      <numeric> <numeric> <numeric> <numeric>
## ENSG000000000003    616.390      -0.01579    0.0856   -0.1844     0.854
## ENSG000000000005     0.553      -0.00836    0.1703   -0.0491     0.961
## ENSG000000000419    305.400      -0.01647    0.0935   -0.1762     0.860
## ENSG000000000457    184.341      -0.09290    0.1180   -0.7874     0.431
##           padj          ensembl      entrez hgnc_symbol
##           <numeric>      <character> <integer> <character>
## ENSG000000000003     0.971 ENSG000000000003      7105      TSPAN6
## ENSG000000000005      NA ENSG000000000005     64102      TNMD
## ENSG000000000419     0.973 ENSG000000000419     8813      DPM1
## ENSG000000000457     0.879 ENSG000000000457    57147      SCYL3
```



## 4.2 Exporting results

Finally, we note that you can easily save the results table in a CSV file, which you can then load with a spreadsheet program such as Excel:

```
res[1:2,]

## log2 fold change (MAP): treatment DPN vs Control
## Wald test p-value: treatment DPN vs Control
## DataFrame with 2 rows and 9 columns
##
```

	baseMean	log2FoldChange	lfcSE	stat	pvalue
##	<numeric>	<numeric>	<numeric>	<numeric>	<numeric>
## ENSG000000000003	616.390	-0.01579	0.0856	-0.1844	0.854
## ENSG000000000005	0.553	-0.00836	0.1703	-0.0491	0.961

```
##
```

	padj	ensembl	entrez	hgnc_symbol
##	<numeric>	<character>	<integer>	<character>
## ENSG000000000003	0.971	ENSG000000000003	7105	TSPAN6
## ENSG000000000005	NA	ENSG000000000005	64102	TNMD

```
write.csv( as.data.frame(res), file="results.csv" )
```

## 5 Working with rlog-transformed data

### 5.1 The rlog transform

Many common statistical methods for exploratory analysis of multidimensional data, especially methods for clustering and ordination (e.g., principal-component analysis and the like), work best for (at least approximately) homoskedastic data; this means that the variance of an observable quantity (i.e., here, the expression strength of a gene) does not depend on the mean. In RNA-Seq data, however, variance grows with the mean. For example, if one performs PCA directly on a matrix of normalized read counts, the result typically depends only on the few most strongly expressed genes because they show the largest absolute differences between samples. A simple and often used strategy to avoid this is to take the logarithm of the normalized count values plus a small pseudocount; however, now the genes with low counts tend to dominate the results because, due to the strong Poisson noise inherent to small count values, they show the strongest relative differences between samples.

As a solution, *DESeq2* offers the *regularized-logarithm transformation*, or *rlog* for short. For genes with high counts, the rlog transformation differs not much from an ordinary log<sub>2</sub> transformation. For genes with lower counts, however, the values are shrunk towards the genes' averages across all samples. Using an empirical Bayesian prior in the form of a *ridge penalty*, this is done such that the rlog-transformed data are approximately homoskedastic.

Note that the rlog transformation is provided for applications other than differential testing. For differential testing we recommend the *DESeq* function applied to raw counts, as described earlier in this

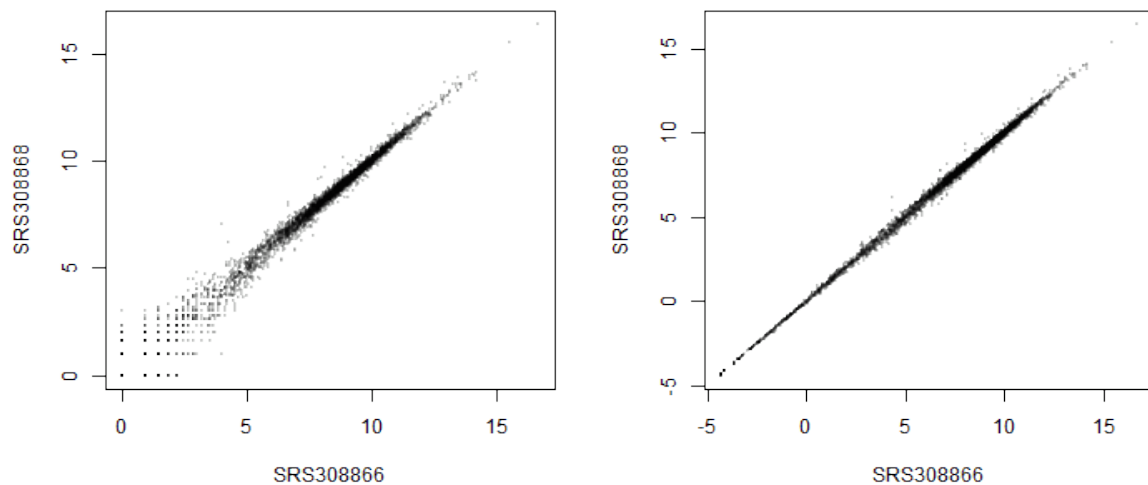


Figure 7: **Scatter plot of sample 2 vs sample 1.** Left: using an ordinary  $\log_2$  transformation. Right: Using the rlog transformation.

vignette, which also takes into account the dependence of the variance of counts on the mean value during the dispersion estimation step.

The function `rlogTransform` returns a *SummarizedExperiment* object which contains the rlog-transformed values in its *assay* slot:

```
rld <- rlog( dds )
head( assay(rld) )
```

##	SRS308866	SRS308868	SRS308870	SRS308872	SRS308874	SRS308876
## ENSG000000000003	9.800	9.778	9.918	9.095	9.163	9.062
## ENSG000000000005	-0.922	-0.812	-0.904	-0.893	-0.975	-0.975
## ENSG000000000419	8.055	8.080	8.117	8.238	8.297	8.379
## ENSG000000000457	7.414	7.255	7.312	7.869	7.739	7.907
## ENSG000000000460	7.573	7.704	7.727	8.076	8.287	8.079
## ENSG000000000938	3.225	3.108	3.114	3.995	3.571	3.573
##	SRS308878	SRS308880	SRS308882	SRS308883	SRS308885	SRS308887
## ENSG000000000003	8.883	8.752	8.809	9.049	9.073	8.857
## ENSG000000000005	-0.975	-0.977	-0.975	-0.975	-0.977	-0.977
## ENSG000000000419	8.323	8.313	8.420	8.272	8.157	8.323
## ENSG000000000457	7.163	7.296	7.383	7.617	7.490	7.501
## ENSG000000000460	7.057	7.433	7.630	6.904	7.347	7.654
## ENSG000000000938	3.025	3.356	3.287	3.331	3.435	3.288

To show the effect of the transformation, we plot the first sample against the second, first simply using the `log2` function (after adding 1, to avoid taking the log of zero), and then using the rlog-transformed values.

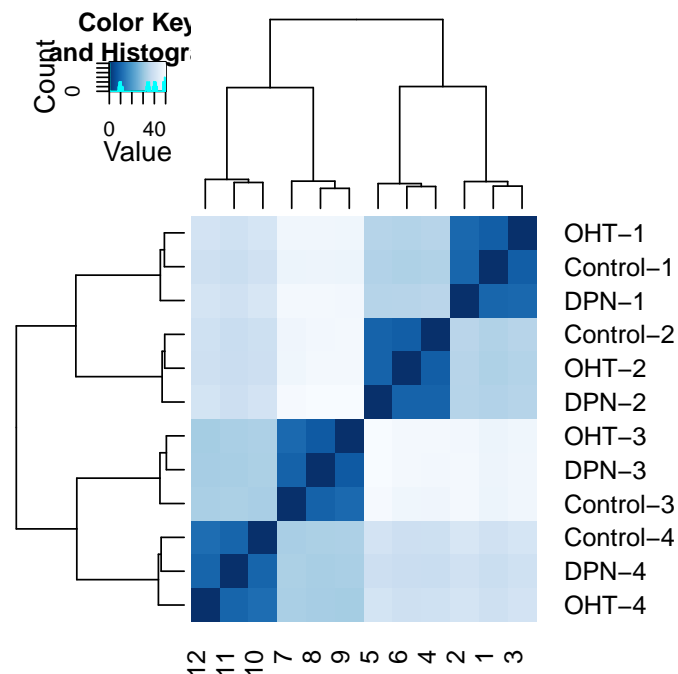


Figure 8: **Heatmap of Euclidean sample distances after rlog transformation.**

```
par( mfrow = c( 1, 2 ) )
plot( log2( 1+counts(dds, normalized=TRUE)[, 1:2] ), col="#00000020", pch=20, cex=0.3 )
plot( assay(rld)[, 1:2], col="#00000020", pch=20, cex=0.3 )
```

Note that, in order to make it easier to see where several points are plotted on top of each other, we set the plotting color to a semi-transparent black (encoded as #00000020) and changed the points to solid disks (pch=20) with reduced size (cex=0.3)<sup>6</sup>.

In Figure 7, we can see how genes with low counts seem to be excessively variable on the ordinary logarithmic scale, while the rlog transform compresses differences for genes for which the data cannot provide good information anyway.

## 5.2 Sample distances

A useful first step in an RNA-Seq analysis is often to assess overall similarity between samples: Which samples are similar to each other, which are different? Does this fit to the expectation from the experiment's design?

We use the R function `dist` to calculate the Euclidean distance between samples. To avoid that the distance measure is dominated by a few highly variable genes, and have a roughly equal contribution

<sup>6</sup>The function `heatscatter` from the package [LSD](#) offers a colorful alternative.

from all genes, we use it on the rlog-transformed data:

```
sampleDists <- dist( t( assay(rld) ) )
sampleDists
```

##	SRS308866	SRS308868	SRS308870	SRS308872	SRS308874	SRS308876	SRS308878
## SRS308868	10.55						
## SRS308870	8.97	10.98					
## SRS308872	34.38	35.82	35.34				
## SRS308874	34.64	35.29	35.21	10.02			
## SRS308876	33.99	35.48	34.90	9.04	9.96		
## SRS308878	47.60	49.41	48.57	48.35	49.78	48.53	
## SRS308880	47.84	49.23	48.64	49.04	50.14	49.32	9.69
## SRS308882	47.71	49.12	48.48	49.25	50.32	49.36	11.15
## SRS308883	40.27	42.28	41.69	39.70	41.14	39.63	33.32
## SRS308885	38.97	40.28	40.10	38.63	39.50	38.75	33.71
## SRS308887	40.02	41.56	41.06	40.14	41.27	39.97	33.51
##	SRS308880	SRS308882	SRS308883	SRS308885			
## SRS308868							
## SRS308870							
## SRS308872							
## SRS308874							
## SRS308876							
## SRS308878							
## SRS308880							
## SRS308882	8.21						
## SRS308883	33.56	33.80					
## SRS308885	32.98	33.42	10.35				
## SRS308887	32.79	32.58	11.91	10.44			

Note the use of the function `t` to transpose the data matrix. We need this because `dist` calculates distances between data *rows* and our samples constitute the columns.

We visualize the distances in a heatmap, using the function `heatmap.2` from the [gplots](#) package.

```
sampleDistMatrix <- as.matrix( sampleDists )
rownames(sampleDistMatrix) <- paste( rld$treatment,
  rld$patient, sep="-" )
colnames(sampleDistMatrix) <- NULL
library( "gplots" )
library( "RColorBrewer" )
colours = colorRampPalette( rev(brewer.pal(9, "Blues")) )(255)
heatmap.2( sampleDistMatrix, trace="none", col=colours)
```

Note that we have changed the row names of the distance matrix to contain treatment type and patient number instead of sample ID, so that we have all this information in view when looking at the heatmap (Fig. 8).

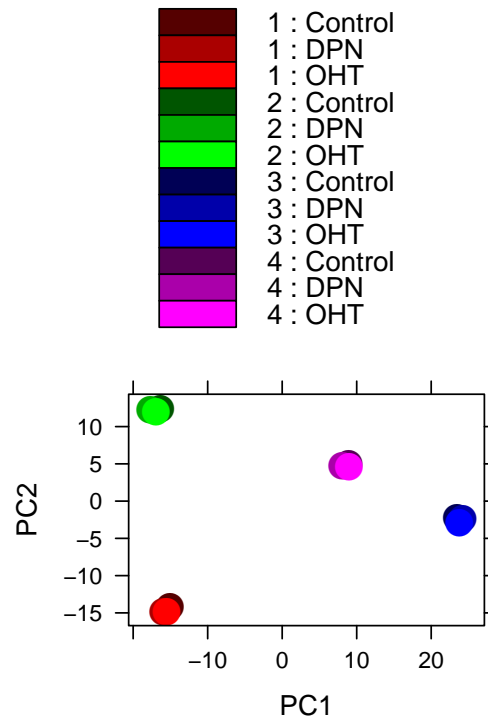


Figure 9: **Principal components analysis (PCA)** of samples after rlog transformation.

Another way to visualize sample-to-sample distances is a principal-components analysis (PCA). In this ordination method, the data points (i.e., here, the samples) are projected onto the 2D plane such that they spread out optimally (Fig. 9). If we only had two treatments, the `plotPCA` will automatically use a paired color palette for each combination of the levels of patient and treatment. As we have three treatments, we supply a vector which specifies three shades of red, green, blue and purple for each patient.

```
ramp <- 1:3/3
cols <- c(rgb(ramp, 0, 0),
          rgb(0, ramp, 0),
          rgb(0, 0, ramp),
          rgb(ramp, 0, ramp))
print( plotPCA( rld, intgroup = c( "patient", "treatment"), col=cols ) )
```

Here, we have used the function `plotPCA` which comes with *DESeq2*. The two terms specified as `intgroup` are column names from our sample data; they tell the function to use them to choose colours.

From both visualizations, we see that the differences between patients is much larger than the difference between treatment and control samples of the same patient. This shows why it was important to account for this paired design (“paired”, because each treated sample is paired with one control sample from the *same* patient). We did so by using the design formula `~ patient + treatment` when setting up the data object in the beginning. Had we used an un-paired analysis, by specifying only `~ treatment`, we

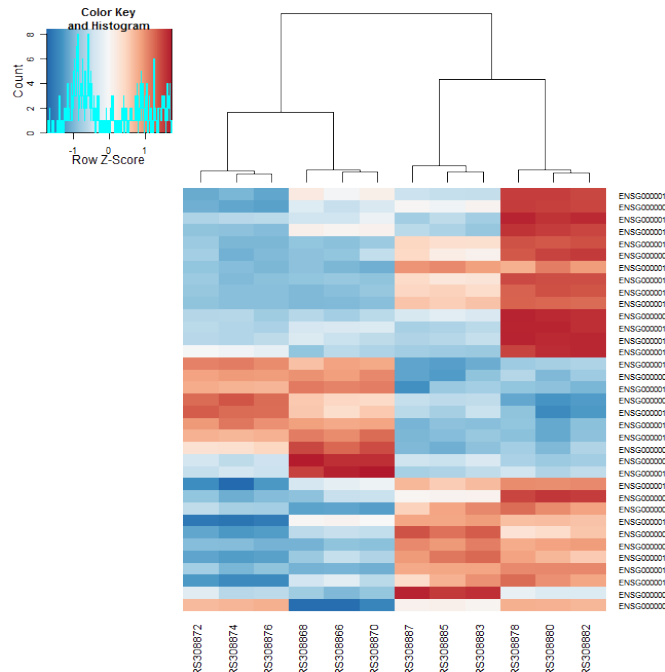


Figure 10: **Heatmap with gene clustering.**

would not have found many hits, because then, the patient-to-patient differences would have drowned out any treatment effects.

Here, we have performed this sample distance analysis towards the end of our analysis. In practice, however, this is a step suitable to give a first overview on the data. Hence, one will typically carry out this analysis as one of the first steps in an analysis. To this end, you may also find the function `arrayQualityMetrics`, from the package of the same name, useful.

## 5.3 Gene clustering

In the heatmap of Fig. 8, the dendrogram at the side shows us a hierarchical clustering of the samples. Such a clustering can also be performed for the genes.

Since the clustering is only relevant for genes that actually carry signal, one usually carries it out only for a subset of most highly variable genes. Here, for demonstration, let us select the 35 genes with the highest variance across samples:

```
library( "genefilter" )
topVarGenes <- head( order( rowVars( assay(rld) ), decreasing=TRUE ), 35 )
```

The heatmap becomes more interesting if we do not look at absolute expression strength but rather at the amount by which each gene deviates in a specific sample from the gene's average across all samples. Hence, we center and scale each genes' values across samples, and plot a heatmap.

```
heatmap.2( assay(rld)[ topVarGenes, ], scale="row",
           trace="none", dendrogram="column",
           col = colorRampPalette( rev(brewer.pal(9, "RdBu")) )(255))
```

We can now see (Fig. 10) blocks of genes which covary across patients. Often, such a heatmap is insightful, even though here, seeing these variations across patients is of limited value because we are rather interested in the effects between the treatments from each patient.

## References

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## 6 Session Info

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As last part of this document, we call the function `sessionInfo`, which reports the version numbers of R and all the packages used in this session. It is good practice to always keep such a record as it will

help to trace down what has happened in case that an R script ceases to work because a package has been changed in a newer version. The session information should also always be included in any emails to the Bioconductor mailing list.

- R version 3.1.0 RC (2014-04-02 r65358), i386-w64-mingw32
- Locale: LC\_COLLATE=C, LC\_CTYPE=English\_United States.1252, LC\_MONETARY=English\_United States.1252, LC\_NUMERIC=C, LC\_TIME=English\_United States.1252
- Base packages: base, datasets, grDevices, graphics, methods, parallel, stats, utils
- Other packages: AnnotationDbi 1.26.0, BSgenome 1.32.0, Biobase 2.24.0, BiocGenerics 0.10.0, Biostrings 2.32.0, DESeq2 1.4.5, GenomInfoDb 1.0.2, GenomicAlignments 1.0.1, GenomicFeatures 1.16.0, GenomicRanges 1.16.3, IRanges 1.22.6, RColorBrewer 1.0-5, Rcpp 0.11.1, RcppArmadillo 0.4.300.0, Rsamtools 1.16.0, XVector 0.4.0, biomaRt 2.20.0, genefilter 1.46.1, gplots 2.13.0, knitr 1.5, parathyroidSE 1.2.0, pasilla 0.4.0, vsn 3.32.0
- Loaded via a namespace (and not attached): BBmisc 1.6, BatchJobs 1.2, BiocInstaller 1.14.2, BiocParallel 0.6.0, BiocStyle 1.2.0, DBI 0.2-7, DESeq 1.16.0, KernSmooth 2.23-12, RCurl 1.95-4.1, RSQLite 0.11.4, XML 3.98-1.1, affy 1.42.2, affyio 1.32.0, annotate 1.42.0, bitops 1.0-6, brew 1.0-6, caTools 1.17, codetools 0.2-8, digest 0.6.4, evaluate 0.5.5, fail 1.2, foreach 1.4.2, formatR 0.10, gdata 2.13.3, geneplotter 1.42.0, grid 3.1.0, gtools 3.4.0, highr 0.3, iterators 1.0.7, lattice 0.20-29, limma 3.20.1, locfit 1.5-9.1, plyr 1.8.1, preprocessCore 1.26.1, rtracklayer 1.24.0, sendmailR 1.1-2, splines 3.1.0, stats4 3.1.0, stringr 0.6.2, survival 2.37-7, tools 3.1.0, xtable 1.7-3, zlibbioc 1.10.0